DISSERTATION

ADVANCES IN TORTRICID SYSTEMATICS AND IDENTIFICATION (LEPIDOPTERA: TORTRICIDAE)

Submitted by

Todd Michael Gilligan

Department of Bioagricultural Sciences and Pest Management

In partial fulfillment of the requirements

For the Degree of Doctor of Philosophy

Colorado State University

Fort Collins, Colorado

Fall 2012

Doctoral Committee:

Advisor: Paul Opler
Co-Advisor: Boris Kondratieff

Terrence Walters
John Brown
Mark Simmons

ABSTRACT

ADVANCES IN TORTRICID SYSTEMATICS AND IDENTIFICATION (LEPIDOPTERA: TORTRICIDAE)

The Tortricidae is a large family in the microlepidoptera, consisting of more than 10,300 species in 1,070 genera. Many tortricids are considered to be economically important, and the family contains the highest percentage of pest species in the Lepidoptera. This dissertation provides significant advances in tortricid systematics and identification through the implementation of modern technologies such as matrix-based identification keys and molecular phylogenetics. Chapter 2 focuses on Tortricidae threatening U.S. agriculture. The resulting resource, Tortricids of Agricultural Importance (TortAI – http://www.tortai.org), is designed to aid in the identification of tortricid adults encountered during domestic surveys and tortricid larvae encountered during quarantine inspections at U.S. ports of entry. Chapters 3–5 provide details on the three tortricid pests currently of greatest importance to U.S. agriculture: *Epiphyas postvittana* (Walker), *Lobesia botrana* ([Denis and Schiffermüller]), and *Thaumatotibia leucotreta* (Meyrick). Chapter 6 details the discovery of a new tortricid species with the potential to threaten avocado production in California. Chapter 7 describes the benefits and disadvantages to using DNA barcoding for identification purposes and outlines a novel DNA sequence search tool developed for use in TortAI. Phylogenetic relationships surrounding *Eucosma*, the largest genus within the Tortricidae, are examined in chapters 8–9. A molecular phylogeny and revised classification for *Eucosma* and related genera provide the groundwork for future study of these highly diverse taxa.

ACKNOWLEDGMENTS

This dissertation and all of its associated publications would not have been possible without the assistance of numerous individuals, all of whom I will attempt to list here. The contributions and efforts of everyone are greatly appreciated.

I would like to thank my CSU committee for all of their assistance during my time at CSU. Paul Opler and Boris Kondratieff were great advisors and provided constant support, advice, and expertise. Terrence Walters provided funding, supervised cooperative agreements, and the majority of this dissertation would not have been possible without his support. John Brown was a wealth of knowledge for everything related to the Tortricidae and provided access to the excellent collection at the Smithsonian. Mark Simmons provided lab and office space, assistance with all aspects of the phylogenetic analyses presented here, and valuable advice.

Special thanks to my home committee, Ella, Tara, and Vixen, for their constant support, patience, and barking. In addition to moving to Colorado and working so that I could play with bugs for the past five years, Ella also helped with web design, databasing, and writing code for the DNA sequence search tool.

Thanks to my parents, Mike and Thora, for continued support and encouragement. I would especially like to thank my father for assistance with field work, specimen preparation, and collecting equipment.

Marc Epstein, CDFA Sacramento, travelled the world with me collecting tortricids during extended trips to Europe, Australia, and Africa. I would like to thank him for all of his help, despite getting us kidnapped by nuns, eaten by alive by leeches, and almost killed by elephants.

Norman Barr, USDA/CPHST Mission Lab, helped with all sequencing projects. I want to thank him for hosting me several times at his lab and for all of his valuable assistance.

I would like to thank everyone, past and present, in the Simmons' lab, including Jennifer Cappa, Luke Tembrock, Jenna McAleer, Salem Eleague, Christine Bacon, Kendra Yakobson, and Jack Munz, for putting up with me in the office, working or helping in the lab, and generally making my time at CSU enjoyable.

Thanks to Donald Wright for helping with all aspects of Eucosmini research, providing specimens, and coauthoring numerous publications.

Many individuals in BSPM provided valuable assistance though teaching support and help with logistics, paperwork, and account management. Special thanks to Janet Dill, Maggie Hirko, Paula White, Cynthia Brown, Andrew Norton, and Tom Holtzer for all of their help.

The following individuals and institutions assisted with, provided logistics for, or provided permits for field work in various countries: Leif Aarvik, Natural History Museum, University of Oslo, Norway; David Agassiz, Weston-Super-Mare, England; Joaquin Baixeras Almela, Institut Cavanilles de Biodiversitat i Biologia Evolutiva, Universitat de Valencia, Spain; Gianfranco Anfora, Claudio Ioriatti, and Valerio Mazzoni, IASMA, S. Michele all'Adige, Italy; Bruno Bagnoli, Consiglio per la Ricerca e la Sperimentazione in Agricoltura, Firenze, Italy; Frans Groenen, Luyksgestel, Netherlands; Bobbie Hitchcock, CSIRO Ecosystem Sciences, Australian National University, Canberra; Marianne Horak, Australian National Insect Collection, CSIRO, Canberra; Margaret Kinnaird, Mpala Research Centre, Kenya; Esther Kioko, Grace Ngugi, and J. Mugambi Ruthiiri, National Museums of Kenya, Nairobi; Andrea Lucchi, University of Pisa, Italy; Max Moulds, David Rentz, and Paul Zborowski, Kuranda, Queensland, Australia; Gionata Pulignani, Mazzei, Castello di Fonterutoli, Castellina in Chianti, Italy; Queensland Parks and Wildlife, Cairns, Queensland; Angus Simpson, Gilgil, Kenya; Marja van der Straten, Plant Protection Service, Wageningen, Netherlands; Daniel Strickman and Rene Sforza, USDA/ARS/European Biological Control Laboratory, Montferrier-sur-Lez, France.

The following persons and institutions provided loans of specimens or donated specimens for morphological study and/or DNA sequencing: John Brown, USDA/ARS/SEL, Smithsonian, Washington D.C.; Bong-Kyu Byun, Department of Biological Sciences, Hannam University, Korea; Soowon Cho, Department of Plant Medicine, Chungbuk National University, Korea; Jamie Davies, Catherine Young, and Graeme Anderson, Department of Primary Industries, Parks, Water and Environment, Tasmania, Australia; Jason Dombroskie, Department of Entomology, Cornell University, Ithaca; Ted Edwards, Bobbie Hitchcock, and Marianne Horak, Australian National Insect Collection, CSIRO, Canberra; Sabine Gaal-Haszler, Naturhistorisches Museum Wien, Vienna; Roberto Gonzalez, Universidad de Chile, Santiago, Chile; Frans Groenen, Luyksgestel, Netherlands; Mark Hoddle and Doug Yanega, Department of Entomology, University of California, Riverside; Esther Kioko and J. Mugambi Ruthiiri, National Museums of Kenya, Nairobi; Eric LaGasa, Washington State Department of Agriculture, Olympia; Steven Passoa, USDA/APHIS/PPQ, Columbus, Ohio; Jerry Powell, University of California, Essig Museum of Entomology, Berkeley; Obie Sage, California Department of Food and Agriculture, Sacramento; Marja van der Straten, Plant Protection Service, Wageningen, Netherlands; Luis Miguel Torres-Vila, Servicio de Sanidad Vegetal, Spain; Kevin Tuck, The Natural History Museum, London; Richard Worth, Oregon Department of Agriculture, Salem; Doug Yanega, University of California, Riverside.

The following individuals and institutions assisted with or provided facilities for DNA sequencing, or provided sequence data used in the DNA sequence search tool: Norman Barr and Lisa Ledezma, USDA/APHIS/PPQ/CPHST, Mission Lab, Edinburg; BOLD Data Systems, Biodiversity Institute of Ontario; Jonathan Brusch, USDA/APHIS/PPQ, Boston; Paul Hebert, University of Guelph, Ontario; Bobbie Hitchcock, CSIRO Ecosystem Sciences, Australian National University, Canberra; Peter Kerr, Scott Kinnee, and Obie Sage, California Department of Food and Agriculture, Sacramento; Richard Newcomb and Leah Tooman, The New Zealand Institute for Plant & Food Research Limited, Auckland, New Zealand; Rodolphe Rougerie, Universite de Rouen, France; Mark Simmons, Colorado State University, Fort Collins; Kendra Yakobson and Jack Munz, formally Colorado State University, Fort Collins.

The following persons provided special assistance or access to equipment: Kevin Hoffman and Obie Sage, California Department of Food and Agriculture, Sacramento; Steven Passoa, USDA/APHIS/PPQ, Columbus; Amanda Redford and Christina Southwick, ITP - USDA/APHIS/PPQ/CPHST, Fort Collins.

Funding was provided by USDA/APHIS/PPQ/CPHST through the following two cooperative agreements with Colorado State University and California Department of Food and Agriculture (principle investigators: T. M. Gilligan and P. A. Opler): “Diagnostic tools for tortricid moths of immediate and future concern to United States agriculture” ($154,400; January, 2009–January, 2012) and “Diagnostic tools for the identification of western U.S. Tortricidae easily confused with the light brown apple moth” ($42,680; March, 2008–March, 2009). Cooperative agreements were supervised by Terrence Walters, Identification Technology Program, USDA/APHIS/PPQ/CPHST, Fort Collins.

TABLE OF CONTENTS

APPENDICES

LIST OF TABLES

CHAPTER 2

CHAPTER 3

CHAPTER 4

CHAPTER 5

CHAPTER 6

CHAPTER 7

CHAPTER 8

CHAPTER 9

LIST OF FIGURES

CHAPTER 2

CHAPTER 3

CHAPTER 4

CHAPTER 5

CHAPTER 6

CHAPTER 7

CHAPTER 8

CHAPTER 9

Chapter 1

General Introduction

Tortricidae

The Tortricoidea is one of the most diverse superfamilies in the microlepidoptera, second only to the Gelechioidea in number of described species (Horak 1998). It contains a single family, Tortricidae, which is composed of more than 10,300 species in approximately 1,070 genera (Gilligan et al. 2012). Tortricid adults are characterized by a combination of the following character states: head rough-scaled above; scaling of lower frons short, appressed and upwardly directed; proboscis well developed and unscaled; labial palpi three-segmented and generally held horizontally or porrect, with apical segment short and blunt; maxillary palpi reduced; ocelli and chaetosema present; and ovipositor lobes flat (Horak 2006). The family is divided into three subfamilies: Tortricinae, Olethreutinae, and Chlidanotinae (Horak 1998). The entire family has been recently catalogued (Brown 2005), and an updated online taxonomic database is available (Gilligan et al. 2012).

Economic importance

Agriculture in the United States is constantly threatened by invasive species. Crop losses in the U.S. because of introduced insect pests are estimated to exceed $14 billion USD annually (Pimentel et al. 2005), and many of these pests are Lepidoptera. Worldwide, Tortricidae contains close to 700 economically important species, a number that is third only to the Noctuoidea and Pyraloidea (Zhang 1994). Identification of tortricid pests can be difficult, and the availability of diagnostic resources prior to an invasion is essential for responding in a timely and appropriate manner to new introductions. This dissertation significantly contributes towards the protection of U.S. agriculture from invasive Lepidoptera pests by providing adult and larval identification resources, descriptions of new pest species, and novel molecular diagnostic tools.

Chapter 2 focuses on Tortricidae threatening U.S. agriculture. The resulting resource, Tortricids of Agricultural Importance (TortAI – http://www.tortai.org), is designed to aid in the identification of tortricid adults encountered during domestic surveys and tortricid larvae encountered during quarantine inspections at U.S. ports of entry. Identification is facilitated through interactive identification keys that include both adult and larval characters along with detailed fact sheets for each tribe, genus, and species, and numerous photographs of adults, larvae, and important morphological characters. This is the first study to use interactive keys for the identification of tortricid pests.

Chapters 3–5 provide details on the three tortricid pests currently of greatest importance to U.S. agriculture: *Epiphyas postvittana* (Walker), *Lobesia botrana* ([Denis and Schiffermüller]), and *Thaumatotibia leucotreta* (Meyrick). *Epiphyas postvittana*, the light brown apple moth, was discovered in California in 2006 (Brown 2010). It rapidly spread throughout the state, resulting in quarantine restrictions on agricultural products that made identification of both adults and larvae a top priority. Chapter 3 details the adult and larval morphology of this pest, and discusses characters that can be used to distinguish it from similar species. *Lobesia botrana*, the European grapevine moth, was discovered in Napa Valley, California in 2009. It is one of the most destructive pests of grape in the Palearctic, and its arrival in California seriously threatens that state's wine industry. Chapter 4 provides an account of the discovery of *L. botrana*, details diagnostic character states, and discusses closely related species present in the eastern U.S. *Thaumatotibia leucotreta*, the false codling moth, is one of the most destructive pests of avocado, citrus, and cotton in Africa. A single male was discovered in California in 2008. Chapter 5 provides a detailed description of this species that can be used by identifiers during domestic surveys for this important pest.

The list of species threatening U.S. agriculture includes undescribed species as well as well-documented pests. Chapter 6 details the discovery of a new tortricid species with the potential to threaten avocado production in California. *Cryptaspasma perseana* Gilligan and Brown is described as new. Adults and larvae are described in detail and its life history is reviewed. A worldwide list of avocado-feeding tortricids is provided along with a key to identifying Lepidoptera larvae threatening avocados in California.

DNA barcoding, or the use of a standardized segment of DNA for species identification and discrimination, is a widely accepted procedure that has been used to identify economically important insects. Chapter 7 describes the benefits and disadvantages to using DNA barcoding for identification purposes, and assesses the utility of the two largest public DNA barcode databases, NCBI-GenBank and BOLD. The criteria for implementing a successful DNA barcoding system for quarantine and regulatory functions are discussed. Details of a DNA barcode identification tool developed to identify tortricid pests and implemented in TortAI (Chapter 2) are provided.

Systematics

Only recently has molecular data been used to infer phylogenetic relationships within the Tortricidae and between the Tortricoidea and other superfamilies. Tortricids are included in the non-obtectomeran Apoditrysia, and have been hypothesized to be related to the Cossoidea or the Cossoidea + Sesioidea by various authors (Common 1990; Horak and Brown 1991, Horak 1998). Recent molecular studies using molecular data to determine relationships within the Ditrysia failed to define a sister group for the Tortricoidea, although the superfamily was consistently recovered as monophyletic (Regier et al. 2009, 2012). Regier et al. (2012) were the first to publish a comprehensive molecular phylogeny of the Tortricidae. They included all three subfamilies and 19 out of 22 tribes in their analysis, and used the results to examine life history evolution within the family. Dombroskie (2011) constructed a molecular phylogeny for the

Archipini using both nuclear and mitochondrial data. He used the results to examine the evolution of secondary sexual characters within the tribe. No published molecular studies have examined relationships below the tribal level within the Olethreutinae.

In chapters 8–9, I examine phylogenetic relationships within *Eucosma*, the largest genus within the Tortricidae, and related genera. Chapter 8 investigates the type species of *Eucosma*, *E. circulana*, for which the type specimen has been lost or destroyed. Candidate taxa are evaluated to match the original description, and a neotype is proposed for *E. circulana*. Chapter 9 presents the first study to infer phylogenetic relationships within the Eucosmini using a combination of molecular and morphological data. The monophyly of *Eucosma*, *Epiblema*, *Pelochrista*, and *Phaneta* is tested in a simultaneous parsimony analysis consisting of 2692 bp of DNA data, 27 morphological characters, and five gap characters. Morphological characters are identified to separate and better define the four genera. The pine-feeding *Eucosma* species are resolved as a monophyletic lineage and placed in a new genus. A new world catalogue of *Eucosma-Pelochrista-Phaneta* is produced that aligns the generic definitions of these genera in the Nearctic and Palearctic. The world catalogue contains 504 species and 320 new combinations.

Summary

This dissertation provides significant advances in tortricid systematics and identification through the implementation of modern technologies such as matrix-based identification keys and molecular phylogenetics. It incorporates studies of worldwide pest species that threaten U.S. agriculture as well as non-pest species that comprise the largest genus in the family. The identification resources presented here are already in use by identifiers at the local, state, and federal levels. A molecular phylogeny and revised classification for genera in the Eucosmini provides the groundwork for future study of taxa in this highly diverse tribe.

References

Brown, J. W. 2005. World catalogue of insects. Volume 5: Tortricidae (Lepidoptera). Apollo Books, Stenstrup, Denmark. 741 pp.

Brown, J. W., M. E. Epstein, T. M. Gilligan, S. Passoa and J. A. Powell. 2010. Biology, identification, and history of the light brown apple moth, *Epiphyas postvittana* (Walker) (Lepidoptera: Tortricidae: Archipini) in California: an example of the importance of local faunal surveys to document the establishment of exotic insects. American Entomologist. 56: 34–43.

Common, I. F. B. 1990. Moths of Australia. Melbourne University Publishing. 535 pp.

Dombroskie, J. J. 2011. Aspects of archipine evolution (Lepidoptera: Tortricidae). University of Alberta, Department of Biological Sciences. Ph.D. dissertation. 488 pp.

Gilligan, T. M., J. Baixeras, J. W. Brown and K. R. Tuck. 2012. T@RTS: Online World Catalogue of the Tortricidae (Ver. 2.0). http://www.tortricid.net/catalogue.asp.

Horak, M. 1991. Morphology, pp. 1–22. *In*: L. P. S. van der Geest, H. H. Evenhuis (eds.), Tortricid Pests: Their Biology, Natural Enemies and Control. Elsevier, Amsterdam, The Netherlands.

Horak, M. 1998. The Tortricoidea, pp. 199–215. *In*: N. P. Kristensen (ed.), Handbook of Zoology, Lepidoptera, Moths and Butterflies, Vol 1: Evolution, Systematics, and Biogeography. Walter de Gruyter, Berlin, New York.

Horak, M. 2006. Olethreutine moths of Australia (Lepidoptera: Tortricidae). Monographs on Australian Lepidoptera, Vol. 10. 522 pp.

Horak, M. and R. L. Brown. 1991. Taxonomy and phylogeny, pp. 23–50. *In*: L. P. S. van der Geest, H. H. Evenhuis (eds.), Tortricid Pests: Their Biology, Natural Enemies and Control. Elsevier, Amsterdam, The Netherlands.

Pimentel, D., R. Zuniga and D. Morrison. 2005. Update on the environmental and economic costs associated with alien-invasive species in the United States. Ecological Economics. 52: 273–288.

Regier, J.C., A. Zwick, M. P. Cummings, A. Y. Kawahara, S. Cho, S. J. Weller, A. D. Roe, J. Baixeras-Almela, J. W. Brown, C. S. Parr, D. R. Davis, M. E. Epstein, W. Hallwachs, A. Hausmann, D. H. Janzen, I. J. Kitching, M. A. Solis, S.-H. Yen, A. Bazinet, C. Mitter. 2009. Toward reconstructing the evolution of advanced moths and butterflies (Lepidoptera: Ditrysia): an initial molecular study. BMC Evolutionary Biology 9: 280.

Regier, J. C., J. W. Brown, C. Mitter, J. Baixeras, S. Cho, M. P. Cummings and A. Zwick. 2012. A molecular phylogeny for the leaf-roller moths (Lepidoptera: Tortricidae) and its implications for classification and life history evolution. PLoS ONE. 7: e35574. doi:10.1371/journal.pone.0035574.

Zhang, B.-C. 1994. Index of economically important Lepidoptera. CAB International, Wallingford, UK. 599 pp.

Chapter 2

Tortricids of Agricultural Importance to the United States
(Lepidoptera: Tortricidae)*

Introduction

Many tortricids are important agricultural pests and the family contains a large number of pest species. Zhang (1994) lists 687 tortricids as economically important, a number third only to the highly diverse Noctuidae, with 1034 pest species, and Pyralidae (+ Crambidae) with 748 recognized pest species. The economic importance of the family is demonstrated by the amount of publications dedicated to the biology, physiology, identification, and control of tortricid moths, and entire volumes have been published on these subjects (e.g., van der Geest and Evenhuis 1991). Perhaps there is no better example of a ubiquitous tortricid pest than the “worm” that is found after biting into an apple, which is actually the larva of the codling moth, *Cydia pomonella* (L.). In today’s world of global trade and travel, the potential for invasive pests to be transported outside of their native range, either accidentally or intentionally, is higher than ever before. The

---

*Portions of this chapter are published in:
Gilligan, T. M. & M. E. Epstein. 2009. LBAM ID: Tools for diagnosing light brown apple moth and related western U.S. leafrollers (Tortricidae: Archipini). CD-ROM. Center for Plant Health Science and Technology, USDA/APHIS/PPQ, Raleigh, NC.

*A full version of this chapter is published as:
Gilligan, T. M. & M. E. Epstein. 2012. TortAI, Tortricids of Agricultural Importance to the United States (Lepidoptera: Tortricidae). CD-ROM. Identification Technology Program (ITP), USDA/APHIS/PPQ/CPHST, Fort Collins, CO.

introduction of pest species to new locations has the potential to greatly affect local agricultural production, and Pimentel et al. (2000) estimate that crop losses in the United States due to damage by introduced insect pests exceeded $14 billion USD in 1998. In addition to supplying food to its 300,000,000+ residents, U.S. agriculture accounts for over $96 billion USD in annual exports (ERS/USDA 2010).

The most important tortricid pests that threaten U.S. agriculture 1) are generally not established in North America, and 2) have the greatest potential to contribute to the decline of domestic food production and/or agricultural exports. The USDA has compiled a list of the top ten tortricid pests threatening U.S. agriculture. These species have been or are currently being surveyed under the USDA's Cooperative Agriculture Pest Survey Program (CAPS 2010) and are listed as primary target species (Table 1). We have compiled a list of 15 additional tortricids using a combination of prior CAPS targets, recent introductions into North America, and introduction potentials, which are listed as secondary target species (Table 2). The majority (60%) of the target species listed here are native to the Palearctic; others are native to Australia, South America, Africa, or Asia. Two species of *Cryptophlebia*, although native to Hawaii, could significantly affect fruit production if imported into the continental U.S.

Taxonomic resources for tortricid adult identification are numerous. The Palearctic fauna has been well documented, and large monographs have been published that illustrate nearly all of the tortricids described from that region (e.g., Bradley et al., 1973, 1979, Kuznetsov 1978, Razowski 2002, 2003, 2008, etc.). The Oriental fauna has been treated by Yasuda (1969, 1972, 1975), Diakonoff (1968, 1973, 1975), and others. The Australian Olethreutine genera have been recently revised by Horak (2006). Relatively few publications exist on the Neotropical and African faunas, although treatments for genera and tribes have been published by various authors (e.g., Diakonoff 1963, Obraztsov 1966, Powell 1986, Aarvik 2004, etc.). The Nearctic fauna is well documented, especially for the Olethreutinae. Heinrich (1923, 1926), Miller (1987), and Gilligan et al. (2008) provide coverage of most of the North American olethreutine species.

The Nearctic Tortricinae has not been treated in its entirety other than by Freeman (1958) for the Archipini and Powell (1964) for the California tortricines. Most recently, Powell and Opler (2009) treat approximately 25% of the western North American Tortricidae.

Table 1: Primary target species

Genus	Species	Author	Common name	Target status	Region
Adoxophyes	*orana*	(Fischer von Röslerstamm)	summer fruit tortrix	primary	Palearctic
Archips	*podana*	(Scopoli)	fruit tree tortrix	primary	Palearctic
Archips	*xylosteanus*	(Linnaeus)	golden variegated leafroller	primary	Palearctic
Crocidosema	*aporema*	(Walsingham)	bean shoot moth	primary	S. America
Epiphyas	*postvittana*	(Walker)	light brown apple moth	primary	Australia
Eupoecilia	*ambiguella*	(Hübner)	vine moth/grape berry moth	primary	Palearctic
Grapholita	*funebrana*	(Treitschke)	plum fruit moth	primary	Palearctic
Lobesia	*botrana*	([Denis & Schiffermüller])	grape berry moth	primary	Palearctic
Thaumatotibia	*leucotreta*	(Meyrick)	false codling moth	primary	Africa
Tortrix	*viridana*	Linnaeus	European oak leafroller	primary	Palearctic

Table 2: Secondary target species

Genus	Species	Author	Common name	Target status	Region
Acleris	*comariana*	(Lienig & Zeller)	strawberry tortrix	secondary	Holarctic
Archips	*crataegana*	(Hübner)	brown oak tortrix	secondary	Palearctic
Archips	*fuscocupreanus*	Walsingham	apple tortrix	secondary	Oriental
Argyrotaenia	*ljungiana*	(Thunberg)	grape tortrix	secondary	Palearctic
Cacoecimorpha	*pronubana*	(Hübner)	carnation tortrix	secondary	Palearctic
Clepsis	*spectrana*	(Treitschke)	cyclamen tortrix	secondary	Palearctic
Cryptaspasma	*perseana*	Gilligan & Brown	[avocado moth]	secondary	C. America
Cryptophlebia	*illepida*	(Butler)	koa seedworm	secondary	Hawaii
Cryptophlebia	*ombrodelta*	(Lower)	litchi fruit moth	secondary	Aust./Hawaii
Cydia	*coniferana*	(Saxesen)	pine resin moth	secondary	Palearctic
Cydia	*fabiovra*	(Meyrick)	pod moth	secondary	S. America
Cydia	*splendana*	(Hübner)	chestnut tortrix/acorn moth	secondary	Palearctic
Enarmonia	*formosana*	(Scopoli)	cherry bark tortrix	secondary	Palearctic
Pandemis	*cerasana*	(Hübner)	barred fruit tree tortrix	secondary	Palearctic
Pandemis	*heparana*	([Denis & Schiffermüller])	dark fruit tree tortrix	secondary	Palearctic

Although detailed larval descriptions have been published for many economically important tortricids, very few comparative taxonomic works have been produced on tortricid larvae. Swatschek (1958) published the first extensive treatment of the Palearctic Tortricidae, where he described the larvae of more than 350 species. MacKay (1959, 1962) published larval descriptions for more than 280 species of Nearctic Tortricidae in two volumes, providing detailed illustrations and keys to species, species groups, and genera. Yasuda (1969) published descriptions and illustrations of the Japanese tortricid fauna. Other important works including tortricid larval systematics are Brown's (1987) treatment of the family and Dugdale et al.'s (2005) guide to quarantine pests, which includes molecular as well as morphological characters.

Despite the relative abundance of literature, identification of tortricid adults and larvae remains problematic. Adult identification often requires genitalic dissection and prior knowledge of intra- versus interspecific variation in wing patterns, sibling species, and other information that may only be available to a specialist with many years of experience. Larval identification is much more difficult and, in many aspects, more important, since it is the larva that causes economic damage and this is the life stage often intercepted during quarantine inspections. Tortricid larvae are surprisingly homogeneous, with very few distinguishing characters available to separate species, or even genera. Further complicating the problem of larval identification is the paucity of material in collections. Published or informal keys to tortricid larvae often fail in practice due to the low sample size examined during construction of the key.

Tortricids of Agricultural Importance (TortAI) is designed to aid in the identification of tortricid adults encountered during domestic surveys and tortricid larvae encountered during quarantine inspections at U.S. ports of entry. This resource [published as a CD-ROM and also available on the Internet at http://www.tortai.org] includes interactive identification keys, detailed fact sheets, an illustrated glossary, information on dissecting and preparing specimens, identification thumbnail galleries, and a database of DNA barcode sequences. The interactive identification keys run in a web browser as a Lucid3 Java Applet. Because the world tortricid fauna is too large to treat as a whole, this resource is not designed to identify every tortricid encountered, but rather to reliably eliminate or confirm target taxa if or when they are encountered. It may not be possible to identify many tortricid larvae using only morphological characters. A molecular sequence search tool is provided to allow identifiers with sequencing capabilities the ability to confirm DNA barcode sequences for the larvae of most taxa treated here (detailed in Chapter 6).

TortAI is designed for use by persons in the continental United States performing domestic surveys for exotic species or encountering exotic species during port inspections (taxa originating from Hawaii are treated as exotic). Although much of the information can be applied to other parts of the world, identification characters for adults and larvae (outlined below) are specific to the scope of the keys and may lead to ambiguous or misleading results when used outside of this context. TortAI is not designed as a general resource for identifying all tortricid species.

Materials and Methods

Specimens were examined and dissected using a Wild M5A stereomicroscope (Wild Heerbrugg AG, Switzerland). Genitalia dissection methodology follows that summarized in Brown and Powell (1991) except that some preparations were mounted using Euparal mounting medium (Bioquip Products, Rancho Dominguez, CA). Larvae were prepared using the following method (O. Sage, pers. comm.): a small incision was made on the ventral mid-line near the head and the intact larva heated in 10% KOH for approximately 15 minutes; the larva was placed in water and the incision extended the full length of the body; the head capsule was carefully removed using forceps; the internal contents of the larva were removed with a brush and the cleaned larval skin stained lightly with chlorazol black; the skin was placed under glass pieces in a petri dish with 100% ethanol and left to dehydrate for one hour; the skin was then mounted in Euparal on a microscope slide; the head was stored in 80% ethanol. Adult morphological terms and wing pattern descriptions follow those in Horak (1984, 1991, 2006) and Gilligan et al. (2008). Larval terminology follows that in MacKay (1959, 1962). The term "microlepidoptera" is used to refer to the superfamilies in the non-obtectomeran Apoditrysia and below, and the term "macrolepidoptera" is used to refer to the superfamilies in the obtectomeran Apoditrysia and above, as outlined by Kristensen (1998).

Adult, larval, and pupal photographs were taken using a Canon EOS D60, 40D, or 7D digital SLR (Canon U.S.A., Lake Success, NY) mounted on a Visionary Digital BK Lab System (Visionary Digital, Palmyra, VA) or Aristo DA-10-W55 light box (Aristo Lighting Technologies, Waterbury, CT). Microscope slide photos were taken using a Nikon DXM1200 digital camera mounted on a Nikon Labophot2 compound microscope (Nikon Instruments, Melville, NY). Some photographs are a combination of several layers produced with Helicon Focus 4.80 (Helicon Soft Ltd., Kharkov, Ukraine). All photographs were edited using Adobe Photoshop CS3 or CS5 Extended and drawings were produced using Adobe Illustrator CS3 or CS5 (Adobe Systems Inc., San Jose, CA). All photographic images and drawings were produced by the authors unless otherwise credited.

Abbreviations for depositories, institutions, and organizations are as follows: ANIC = Australian National Insect Collection, CSIRO, Canberra; BOLD = Barcode of Life Data Systems; CSCA = California State Collection of Arthropods, CDFA, Sacramento; CSU = Colorado State University; Genbank = NCBI GenBank; Mission Lab = USDA/APHIS/PPQ Moore Air Base, Mission, TX; TMG = Todd M. Gilligan collection, Loveland, CO; UCB = University of California, Berkeley, Essig Museum of Entomology; UCR = University of California, Riverside; USNM = United States National Museum, Smithsonian, Washington, D.C.

Adults and dissections used to produce photos of male and female genitalia for the fact sheets and interactive keys are listed here (slide codes correspond to collection abbreviations): http://idtools.org/id/leps/tortai/TortAI_Genitalia_slide_log.pdf. A partial list of larvae used to define morphological characters for the interactive larval key and fact sheets are listed here: http://idtools.org/id/leps/tortai/TortAI_Larvae_examined_log.pdf.

Information for the species accounts was obtained from the references cited with the exception of the taxonomy and host plants sections, and the following resources are not cited under each individual species account. Taxonomic information was retrieved from the Online World Catalogue of the Tortricidae (Baixeras et al. 2010). Host plant information was obtained from the Food plant database of the leafrollers of the world (Brown et al. 2008). In many cases, additional host plant information was obtained from the references cited for each taxon. All plant family assignments were cross-checked against the USDA Plants database (USDA, NRCS 2012), and common names for each species, when available, were also assigned using the USDA Plants database. Other plant names not in the USDA Plants database were cross-checked against the International Plant Names Index (IPNI 2008).

TortAI Adult Key

The adult key treats 98 taxa (species, genera, or tribes) that are likely to be encountered during domestic surveys performed under the USDA Cooperative Agricultural Pest Survey (CAPS) Program. Taxa are divided into primary targets, secondary targets, and non-targets. The key assumes that the moths being identified were captured using pheromone traps, and it allows the user to restrict taxa based on trap origin (geographic location). Identification is performed using primarily forewing pattern and male genitalia, although other diagnostic characters for the head, hindwings, and female genitalia are also included. Morphological characters used in the key are illustrated and discussed in Appendix I. The online key is located at the following web address: http://idtools.org/id/leps/tortai/keys/TortAIAdults.html.

Ten primary target taxa are treated (Table 1). These taxa were defined by USDA experts for the 2009 Lepidoptera Adult Identification Workshop as the tortricids providing the greatest threat to U.S. agriculture (J. Brown, pers. comm.). Several of these species are present in the U.S. but have a limited distribution. Fifteen secondary target taxa are treated (Table 2). These taxa are defined as those most likely to pose a future threat to U.S. agriculture and/or those that were previously included in USDA CAPS surveys. The majority of these species are present in the U.S. or North America but have a limited distribution. Seventy-three non-target taxa are treated. These taxa are defined as those most likely to be encountered during surveys for, and confused with, the primary and secondary targets. As the North American tortricid fauna includes well over 1,200 described species, it is not expected that all non-targets can be identified using the key, but coverage should be sufficient to eliminate or confirm target species and identify common non-targets. With a few exceptions, the key does not include forest pests, such as the spruce budworm, as these species are unlikely to be encountered during surveys for the targets treated here.

Adult key instructions

The following instructions apply to the TortAI Adult Key running as a Lucid3 Java Applet from the TortAI website. As this is an interactive matrix-based key, a paper/printable version of the key is not available. The adult key is navigated by clicking in a checkbox or by clicking directly on an image for those features that are illustrated. Chosen features are displayed in the bottom left pane. Choosing multiple states for a single feature functions as an “OR” statement (e.g., forewing pattern: costal spot OR tornal spot; not costal spot AND tornal spot). Remaining entities (taxa) are displayed in the top right pane and eliminated entities are displayed in the bottom right pane. Because this key has several dependent features, it may be helpful to reload the key if too many mistakes are made selecting features. The key can be reloaded by choosing “Restart” under the “Key” menu. The Lucid3 “Best” function should be used with caution. The key will function more efficiently if users manually select characters that they are familiar with instead of using “Best.” The “Best” algorithm automatically calculates and selects the feature that would eliminate approximately half of the remaining entities. As the key is designed with several dependencies and many characters are unique to a small group of taxa, the “Best” function automatically skips many important characters. Exclusive use of the “Best” function may not result in a successful identification.

Primary key feature states (characters) are outlined in the following section. Users are forced to start with features 1-4 (in order) by dependencies set in the key.

1. “Is the moth a tortricid?”

   Choose “Yes” if the specimen being keyed is a tortricid, or choose “Unsure” if you are not certain. Choosing “Yes” will automatically skip to #3 (below), “Choose the moth's origin.” Choosing “Unsure” will automatically load several family level characters. The third choice, “Skip to tortricid characters,” is for advanced users only and is not recommended for the novice or first time user (this option skips all family characters as well as geographic origin).
2. “Family level characters”

   This option is only loaded if “Unsure” was selected in #1 (above). Select morphological characters here to determine if the specimen is a tortricid. Six Lepidoptera superfamilies containing taxa that may be easily confused with tortricids are included in the entity list. The next option (#3) will only appear if one or more tortricid species remain in the entity list; if one of the other superfamilies is the only remaining entity, the key is completed and additional resources for Lepidoptera identification should be consulted.
3. “Choose the moth’s origin”

   Select the geographic origin of the specimen you are trying to identify. Because surveys for target species can occur in any location, this option only restricts non-targets. If you do not know the origin or are unsure, select “Unknown.” It is also possible to select “Exotic” for specimens originating from outside of the continental U.S.; however, this option should be used with caution as the key is not designed to identify most exotic non-targets.

4. “Choose the moth’s sex”

   Select the sex of the specimen. Male tortricids have a frenulum consisting of a single bristle, while female tortricids have a frenulum consisting of two or more bristles. Choosing the appropriate sex loads gender-specific characters (such as genitalia). If the sex of the specimen cannot be determined, “unknown” may be selected; however, this option should be used with caution as it will load all characters for both sexes and could lead to incorrect character combinations (e.g., a forewing costal fold in combination with female genitalic characters). Keep in mind that, while pheromone traps primarily target male moths, females are commonly found in traps and the sex of any particular specimen should not be assumed.

5. “Head”

   Three characters are provided for the head. The number of scale rows per antennal segment can be used to separate Tortricinae from Olethreutinae. The other two characters (labial palpi length and ocelli presence) are useful for certain Sparganothini.

6. “Wings”

   Wing patterns are the second most useful diagnostic characters in the key. When choosing forewing patterns, select the predominate feature for the specimen you are examining. In many cases, it may be helpful to select more than one state. Forewing length, the presence or absence of a costal fold (males only), and general hindwing appearance can also be selected. It may be difficult or impossible to make a final determination based only on wing pattern; in most cases a combination of wing pattern and male genitalia will be required for positive identification.

7. “Male genitalia”

   These features will load only if “male” or “unknown” was selected under #4 (above). Male genitalia are the most useful diagnostic characters in the key. Genitalia are grouped by tribal type; these types are fairly consistent and can be selected based on general appearance. The type "other" covers all other types not similar to the four tribes illustrated. Both the Archipini type and Grapholitini type are further divided into genera, which are then divided into species; the other types are divided directly into species. If the genitalia of the specimen you are attempting to key does not match any of the species listed, it may be necessary to back up a level and start again, or accept an identification at the generic or tribal level (Archips sp. or Archipini). Genitalia for all primary and secondary species are illustrated here, thus failure to match a particular species likely means that you can rule out your specimen as a target; however, always consult the fact sheets in detail before making a final determination.

8. "Female genitalia"

   These features will only load if "female" or "unknown" was selected under #4 (above). Because female genitalia are more generalized than male genitalia (with fewer species-specific characters), general characters are listed here that can be used to retain or eliminate remaining taxa. All characters are illustrated and can be selected by clicking on the appropriate image. It is unlikely that two feature states for any single character would be selected in this section. Consult the fact sheets in detail for species-specific characters not listed here.

When the key is used to confirm or eliminate target taxa, the identifier is successful if the targets have all been discarded (regardless of remaining entities), or the key is completed and only a single target is left in the entities remaining. When the key is used to identify non-target taxa, the identifier is successful if all target taxa have been discarded and a single non-target is left in the entities remaining. It may be difficult to positively identify all non-targets using this key; other resources are provided in the fact sheets [on the website]. The identifier should consult the fact sheets for any remaining entities to determine if the description of the species is consistent with the specimen being examined. Positive target species determinations may need to be sent to an expert for verification. The complete data matrix for the adult CAPS key is available at the following web link: http://idtools.org/id/leps/tortai/CAPS_key_matrix.pdf.

TortAI Larval Key

The larval key treats 50 taxa (species, genera, or tribes) that are most commonly intercepted during quarantine surveys by the USDA at U.S. ports of entry. All taxa are treated as targets, although some may be considered non-actionable. Taxa were selected by consulting USDA identification specialists and the USDA PestID database; the key is not designed to identify all exotic tortricid larvae. The key assumes that the larvae are late instar and originated outside of the continental United States (species originating from Hawaii are treated as exotic). Taxa can be restricted by origin (geographic location) and host, and many times this information is necessary to obtain a successful determination. Identification is performed using a variety of morphological characters in conjuction with the origin and host data. Morphological characters used in the key are illustrated and discussed in Appendix I. Specific instructions for the key are provided below. The online key is located at the following web address: http://idtools.org/id/leps/tortai/keys/TortAILarvae.html.

Identifiers unfamiliar with tortricid larvae should consult MacKay (1959, 1962), Brown (1987), and Gilligan et al. (2008) for an overview of larval morphology and techniques. Larval characters vary extensively, even in the same species, both between instars and in the same instar. Most of the characters included in the key are based on late or last instar larvae, and early instars of many species may be impossible to identify. When examining pinacula and setal number, both sides of the larva should be examined as some specimens are asymmetrical. When an asymmetrical individual is found, use the highest number of setae when navigating the keys. Preservation of larvae can obscure characters, and even freshly preserved specimens may discolor and make many of the color or pattern characters in the keys useless. It is also possible for pieces to break off specimens preserved in alcohol, and occasionally setae or the anal comb may be missing because of breakage. Any determination made with the tortricid larval key should be checked by an expert, and molecular analysis may be required to confirm identity in many cases.

Larval key instructions

The following instructions apply to the TortAI Larval Key running as a Lucid3 Java Applet from the TortAI website. As this is an interactive matrix-based key, a paper/printable version of the key is not available. The adult key is navigated by clicking in a checkbox next to the feature state. Chosen features are displayed in the bottom left pane. Choosing multiple states for color or pattern features functions as an "AND" statement (body color: green AND brown = greenish-brown). Choosing multiple states for morphological features functions as an "OR" statement (anal comb: present OR absent). Remaining entities (taxa) are displayed in the top right pane and eliminated entities are displayed in the bottom right pane. Because this key has several dependent features, it may be helpful to reload the key if too many mistakes are made selecting features. The key can be reloaded by choosing "Restart" under the "Key" menu. The Lucid3 "Best" function should be used with caution. The key will function more efficiently if users manually select characters that they are familiar with instead of using "Best." The "Best" algorithm automatically calculates and selects the feature that would eliminate approximately half of the remaining entities. As the key is designed with several dependencies and many characters are unique to a small group of taxa, the "Best" function automatically skips many important characters. Exclusive use of the "Best" function may not result in a successful identification.

Primary key feature states (characters) are outlined in the following section. Users are forced to start with features 1-3 (in order) by dependencies set in the key. Feature #4 (hosts) is also a dependency for all larval origins except "Canada" and "Unknown."

1. “Is the larva a tortricid?”

    Choose “Yes” if the specimen being keyed is a tortricid, or choose “Unsure” if you are not certain. Choosing “Yes” will automatically skip to #3 (below), “Choose the larval origin.” Choosing “Unsure” will automatically load several family level characters.

2. “Family level characters”

    This option is only available if “Unsure” was selected in #1 (above). Select morphological characters here to determine if the specimen is a tortricid. Three Lepidoptera superfamilies containing taxa that may be commonly confused with tortricids are included in the entity list. Two general categories are also included to account for character states that do not fit with any specific entities. The next option (#3) will only appear if one or more tortricid species remain in the entity list; if one of the other superfamilies or general categories is the only remaining entity, the key is completed and additional resources for larval identification should be consulted.

3. “Choose the larval origin”

    Select the geographic origin of the specimen you are trying to identify. If you do not know the origin or are unsure, select “Unknown;” however, selecting “Unknown” may reduce your chances of a successful determination as host information will not be loaded. There are also no regular hosts associated with larvae originating from Canada.

4. “Hosts (origin)”

    This option will load if any origin other than “Canada” or “Unknown” is selected in #3 (above). Choose the particular host on which the larva was intercepted, if this information is known. Choosing a host is optional, and choices here do not affect other characters in the key. Do not guess at a host if it unknown or not listed. The host lists are the export products on which a particular tortricid species or group is commonly intercepted. In many cases, selecting a host will narrow the entity list to one taxon (e.g. Africa: *Capsicum* = *Thaumatotibia leucotreta*). This does not mean that the single entity is the only larva found on this combination of origin/host, but rather that it is the most likely choice. All entities that remain after choosing a specific host should be checked for matching morphological characters. Choosing a host option is required to display the remaining features #5-8 (below).

5. “Head”

    All of the head features listed here are instar-dependent. The general color, markings, and mandible morphology can vary between instars, and all of the feature states in this section are coded assuming a late or last instar larva.

6. “Thorax”

    The thorax includes several diagnostic structures, such as the prothoracic shield and the prothoracic prespiracular pinaculum. These features (especially the prespiracular pinaculum shape) can be used to narrow the remaining taxa to several species groups. As with the head, markings and coloration on the legs and prothoracic shield change over time, and they are coded here assuming a late instar larva.

7. “Abdomen”

    The majority of diagnostic characters are found on the abdomen, and as many features should be scored here as possible. Markings, coloration, and crochet count may change between instars, but the general chaetotaxy should remain fairly consistent. All taxa are scored assuming a late instar larva.

8. “Feeding habits”

    If known, choose the feeding habits of the larva. This feature separates borers from external feeders.

The identifier is successful when the key is completed and one or more entities remain. In some cases it may be impossible to key a larva to a single entity, especially if location and/or intercepted host information is not known. The identifier should consult the fact sheets for any remaining entities to determine if the description of the species is consistent with the specimen being examined. DNA data may be necessary to provide or confirm identifications for many intercepted tortricid larvae. The complete data matrix for the larval key is available at the following web link: http://idtools.org/id/leps/tortai/Port_key_matrix.pdf.

Species Accounts

Tortricidae: Tortricinae: Tortricini

Acleris Hübner

Overview

Acleris is a large genus in the Tortricini that contains approximately 250 species (Baixeras et al. 2008). Most species diversity occurs in the Palearctic and Nearctic. Forewing pattern is quite variable in many species and some, such as *A. hastiana*, have been described as the most variable of all Lepidoptera in terms of markings and coloration. The high diversity of wing patterns has lead to the application of over 800 names to this group describing the various forms, aberrations, and varieties (Bradley et al. 1973). Many species have patches of raised scales on the forewings.

Several species are of economic concern. The budworms, *A. variana* and *A. gloverana*, are important forest pests that can cause serious damage to fir, spruce, and other Pinaceae. *Acleris comariana* is a pest of strawberry and other Rosaceae and has been recently introduced into the Pacific Northwest. *Acleris minuta* is a minor pest of cranberry, apple, and plum. Other species are minor pests of fruit trees.

It is assumed that any *Acleris* larvae arriving at U.S. ports would originate from Europe, although there are no specific records for this genus. Identification of larvae is difficult and molecular diagnostics may be necessary to confirm identity. Eleven species are treated here, primarily to aid in identification as non-target captures during CAPS surveys. All are present in the U.S., although some, such as *A. comariana* and *A. holmiana*, have limited distribution.

Acleris comariana **(Lienig & Zeller)**

Nomenclature

Acleris comariana (Lienig & Zeller) (Tortricidae: Tortricinae: Tortricini)

Common names: strawberry tortrix

Synonyms: *baracola* (*Acalla*), *burnneana* (ab.), *comparana* (ab.), *fasciana* (ab.), *fuscana* (ab.), *latifasciana* (ab.), *meincki* (*Acalla*), *potentillana* (ab.), *proteana* (ab.)

Adult Recognition

Seven forms or aberrations have been described, many of which are identical in appearance to other *Acleris* species such as *A. laterana* and *A. extensana*. Wing pattern among the different forms varies considerably, but all have a dark triangular costal patch which ranges in color from brown to red to black. The general form of the male genitalia is sufficient to place individuals in the *A. comariana/laterana* complex. Female genitalia are less diagnostic but *A. comariana* has a much reduced signum. Males lack a forewing costal fold. FWL: 5.0-8.5 mm.

Acleris comariana is most closely related to *A. laterana*, which is referred to as the synonym *A. latifasciana* in many publications. Because Sheldon (1925) described many of the *A. comariana* forms using names identical to *Acleris* species names, literature discussions of differences in species and forms must be carefully followed to avoid confusion. Razowski (2002) illustrates differences in genitalia that can be used to separate *A. comariana* from other closely related European *Acleris*.

Larval Morphology

Early instar larvae are white with a black head, prothoracic shield, and legs. Abdomen color varies in later instars, but all have a darker subdorsal line and conspicuous brown pinacula. The head is yellowish brown along with the prothoracic shield, which has a dark brown or black posterior margin.

Larvae of *A. laterana* can be distinguished from *A. comariana* by the dark brown to black posterior margin of the prothoracic shield, which has two subdorsal spots in *A. laterana*, and brown pinacula with are concolorous with the abdomen and not conspicuous in *A. laterana* (Bradley et al. 1973). *Acleris laterana* has not been recorded from North America.

Biology

The following life history information was compiled from Bradley et al. (1973) and Turner (1968). *Acleris comariana* completes two generations in Europe, with adults present from mid-June to July and again from late August to early November. This species prefers wetlands and bogs where many of its host plants occur.

Females deposit eggs on lower parts of the plant, usually on the stipules or petioles. Eggs from the first generation hatch in approximately 10 days; those of the second generation turn a reddish color and hatch the following spring. Larvae feed inside tied or folded leaves although they may also feed on flowers of some hosts. In strawberry, flower-feeding causes distortion of fruit or prevents it from developing. Pupation occurs in a cocoon that is constructed in a rolled leaf or between the stem and petioles.

Acleris comariana feeds on a variety of plants in the family Rosaceae (Table 3). Records of A. *laterana* (= *latifasciana*) on azaleas and rhododendron (Ericaceae: *Rhododendron* L.) from continental Europe are thought to actually refer to *A. comariana*.

Distribution

In the Palearctic, *A. comariana* is widely distributed across Europe to China and Japan. It is also found in Canada and has been presumably introduced into the United States (Washington) and New Zealand (Bradley et al. 1973, Dugdale et al. 2005, Razowski 2002).

Table 3: *Acleris comariana* host plants

Family	**Genus/species**	**Common name**
Ericaceae	*Rhododendron* L.	rhododendron
Rosaceae	*Comarum* L.	comarum
Rosaceae	*Comarum palustre* L.	purple marshlocks
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Fragaria* L.	strawberry
Rosaceae	*Fragaria vesca* L.	woodland strawberry
Rosaceae	*Geum* L.	avens
Rosaceae	*Geum rivale* L.	purple avens
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Malus toringo* (Siebold) Siebold ex de Vriese	toringo crab
Rosaceae	*Potentilla* L.	cinquefoil

***Acleris flavivittana* (Clemens)**

Nomenclature

Acleris flavivittana (Clemens) (Tortricidae: Tortricinae: Tortricini)

Common names: masked leaf roller

Synonyms: *perspicuana* (*Teras*)

Adult Recognition

Forewing pattern is variable, with ground color varying from dark gray to brown, red, or black. The dorsal margin is pale yellow or white in some individuals. Males lack a forewing costal fold. A genitalic dissection may be necessary to separate this species from other *Acleris*. FWL: 7.5-10.0 mm.

Larval Morphology

Last instar larvae are 12-17 mm in length. The head is dark brown ventrally and light brown dorsally, although the degree of contrast between the two halves can vary. Other diagnostic features of the larvae include the prothoracic shield, which is shaded posterolaterally with dark brown to black and the thoracic legs, with are dark on the prothorax and pale on the other two segments (Chapman and Lienk 1971).

Biology

Life history information is not well documented. Chapman and Lienk (1971) suggest that only one generation is produced in the northeastern U.S. Larvae complete development in late July to late August, and adults are present August and September. Overwintering probably occurs as a mid-instar larva.

Larvae feed between two leaves, skeletonizing the top surface of one the leaves. Larvae will feed on apples if given an opportunity, although this species has not been recorded damaging cultivated apples. In addition to apple, *A. flavivittana* has been recorded feeding on pin cherry (Chapman and Lienk 1971) (Table 4).

Table 4: *Acleris flavivittana* host plants

Family	**Genus/species**	**Common name**
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus pensylvanica* L. f.	pin cherry

Distribution

Acleris flavivittana occurs in the northeastern United States and southern Canada. It has also been recorded from North Carolina.

***Acleris gloverana* (Walsingham)**

Nomenclature

Acleris gloverana (Walsingham) (Tortricidae: Tortricinae: Tortricini)

Common names: western blackheaded budworm

Adult Recognition

Forewing pattern is variable and Powell (1962) describes eight different forms. The most common forms are illustrated here. Most forms are also present in *A. variana*. Males lack a forewing costal fold. FWL: 8.0-10.5 mm.

Acleris gloverana is nearly identical to *A. variana* in both biology and morphology. The two species are geographically separated: *A. gloverana* occurs in western North America and *A. variana* occurs in eastern North America (Otvos 1991). Powell (1962) outlined minor genitalic differences that can be used to separate the two species. In males, the sacculus is broader and less elongate in *A. gloverana*, while it is narrower and more elongate in *A. variana*. In *A. gloverana* females, the sterigma is broad, with lobes connected, and a cestum is absent. In *A. variana* females, the sterigma is narrow with the lobes separate, and a weakly sclerotized cestum is present. In addition, females of *A. variana* have large abdominal scale tufts that are used to cover the eggs; these are lacking in *A. gloverana* females.

Larval Morphology

Larvae are approximately 11-15 mm in length and are green with a black head that turns brown in the final instar. The prothoracic shield is green to brown and heavily shaded with black on the posterolateral margins. An anal comb is present with 6-10 teeth (EPPO 1997, Powell 1962).

Biology

The following life history information is summarized from Otvos (1991), Powell (1962), and Powell and Opler (2009). *Acleris gloverana* completes one generation per year. Adults are present in late July through September.

Females lay single eggs on the underside of needles in the upper regions of host trees. Eggs overwinter until the following spring, and larvae hatch starting in mid-May. First instars feed inside opening buds, mid-instars web together needles to create a nest, and last instars are free-feeding. Males and 50% of females complete 4 larval instars while the remaining females complete 5 instars. Pupation occurs in webbed needles. Adults eclose in approximately two weeks.

Acleris gloverana feeds on a variety of fir, hemlock, larch, and spruce (Table 5). This species can be a serious forest pest, and severe outbreaks can cover millions of acres. In coastal forests, extensive defoliation by *A. gloverana* can result in up to 50% tree mortality. Outbreaks tend to occur after periods of low rainfall.

Table 5: *Acleris gloverana* host plants

Family	**Genus/species**	**Common name**
Pinaceae	*Abies balsamea* (L.) Mill.	balsam fir
Pinaceae	*Abies concolor* (Gord. & Glend.) Lindl. ex Hildebr.	white fir
Pinaceae	*Abies* Mill.	fir
Pinaceae	*Larix* Mill.	larch
Pinaceae	*Picea* A. Dietr.	spruce
Pinaceae	*Picea sitchensis* (Bong.) Carriere	Sitka spruce
Pinaceae	*Pseudostuga* Carriere	Douglas-fir
Pinaceae	*Pseudotsuga menziesii* (Mirb.) Franco	Douglas-fir
Pinaceae	*Tsuga* Carriere	hemlock
Pinaceae	*Tsuga heterophylla* (Raf.) Sarg.	western hemlock

Distribution

Acleris gloverana occurs from Alaska and northwestern Canada south to northern California and east to western Montana (Powell 1962, Powell and Opler 2009).

***Acleris hastiana* (Linnaeus)**

Nomenclature

Acleris hastiana (Linnaeus) (Tortricidae: Tortricinae: Tortricini)

Synonyms: a highly variable forewing pattern has led to the description of over 125 synonyms, including many forms, aberrations, and varieties

Adult Recognition

This is possibly the most polymorphic tortricid, with over 100 named forms representing different phenotypes (Bradley et al. 1973, Obraztsov 1963). Forewing patterns can be divided into those with longitudinal markings and those with transverse markings, although many forms show a combination of these two classes. According to Powell (1964), California individuals usually have uniform brown, black, or dull-red forewings with a gray basal band. Males lack a forewing costal fold. FWL: 7.5-12.0 mm.

Acleris hastiana can be confused with other species of *Acleris* due to its highly variable forewing pattern. Although it may be possible to diagnose *A. hastiana* using only wing pattern, a genitalic dissection should be used to confirm identity. Razowski (2002) illustrates differences in genitalia that can be used to separate *A. hastiana* from other similar European *Acleris*, such as *A. abietana*, *A. aspersana*, *A. cristana*, *A. shepherdana* and *A. umbrana*.

Larval Morphology

Late instar larvae are pale green or yellow with a brownish-yellow head and pale thoracic legs. The prothoracic shield is concolorous with the abdomen. In earlier instars, the head and prothoracic shield are black (Bradley et al. 1973).

Biology

The following life history information is summarized from Bradley et al. (1973), Obraztsov (1963), and Powell (1964). *Acleris hastiana* completes one or two annual generations. Adults are present in June and July and again in August through October. Overwintering occurs as a second generation adult.

Females deposit individual eggs on bark or branches in the spring (first generation) and on terminal shoots in the summer (second generation). First instar larvae bore into buds. Later intars feed within shelters constructed by webbing together terminal leaves. Pupation occurs in a flimy silken cocoon that is constructed in folded or webbed leaves or on the ground. In North America, this species has been recorded feeding on blueberry (*Vaccinium*), bog rosemary (*Andromeda*), *Ceanothus*, huckleberry (*Gaylussacia*), oak (*Quercus*), *Rhododendron*, and willow (*Salix*) (Table 6).

Distribution

Acleris hastiana is Holarctic and is widely distributed across Europe and Asia to Japan and North Africa. In North America this species is distributed from the northeastern United

States across southern Canada to British Columbia and south along the Pacific Coast to California (Powell 1964, Razowski 2002).

Table 6: *Acleris hastiana* host plants

Family	Genus/species	Common name
Betulaceae	*Betula papyrifera* Marsh.	paper birch
Ericaceae	*Andromeda polifolia* L.	bog rosemary
Ericaceae	*Rhododendron* L.	rhododendron
Ericaceae	*Vaccinium* L.	blueberry
Fagaceae	*Quercus dumosa* Nutt.	coastal sage scrub oak
Myricaceae	*Myrica gale* L.	sweetgale
Rhamnaceae	*Ceanothus thyrsiflorus* Eschsch.	blueblossom
Rosaceae	*Prunus* L.	
Salicaceae	*Populus alba* L.	white poplar
Salicaceae	*Populus balsamifera* L.	balsam poplar
Salicaceae	*Populus tremuloides* Michx.	quaking aspen
Salicaceae	*Populus* L.	cottonwood
Salicaceae	*Salix aurita* L.	eared willow
Salicaceae	*Salix caprea* L.	goat willow
Salicaceae	*Salix cinerea* L.	large gray willow
Salicaceae	*Salix fragilis* L.	crack willow
Salicaceae	*Salix hookeriana* Barratt ex Hook.	dune willow
Salicaceae	*Salix laevigata* Bebb	red willow
Salicaceae	*Salix lasiolepis* Benth.	arroyo willow
Salicaceae	*Salix repens* L.	creeping willow
Salicaceae	*Salix scouleriana* Barratt ex Hook.	Scouler's willow
Salicaceae	*Salix sessilifolia* Nutt.	northwest sandbar willow
Salicaceae	*Salix sitchensis* Sanson ex Bong.	Sitka willow
Salicaceae	*Salix* X *sepulcralis* Simonkai [*alba* X ?*pendulina*]	weeping willow
Salicaceae	*Salix* L.	willow

Acleris holmiana **(Linnaeus)**

Nomenclature

Acleris holmiana (Linnaeus) (Tortricidae: Tortricinae: Tortricini)

Adult Recognition

Adults are yellowish brown to orange or red with a conspicuous, white, semitriangular costal patch. Males lack a forewing costal fold. FWL: 5.5-8.0 mm. Male genitalia are characterized by a reduced uncus and elongate valva with an apical membranous lobe. Female genitalia are characterized by a small corpus bursae with a single scobinate signum.

Some (aberrant) individuals of *Archips argyrospila* can appear similar, although the costal patch is usually trapezoidal rather than triangular in *A. argyrospila* and males have a forewing costal fold (Mutuura and Munroe 1978). A genitalic dissection can easily separate *A. holmiana* from *A. argyrospila*.

Acleris holmiana was collected in large numbers as a non-target during *E. postvittana* pheromone trap surveys conducted in Washington (E. LaGasa, pers. comm.).

Larval Morphology

Last instar larvae are yellowish to pale green. The head is light brown to reddish brown with black posterior shading. The prothoracic shield is black or dark brown with black posterolateral margins. An anal comb is present (Bradley et al. 1973).

Biology

In North America, *A. holmiana* completes two, and possibly three, generations per year. Adults are present in May and again in June and July. Larvae create a shelter by webbing together two leaves at their margins. Pupation occurs in the larval shelter. Overwintering occurs in the egg stage (Mutuura and Monroe 1978, Obraztsov 1963).

This species is a minor pest of fruit trees (Rosaceae) in Europe (Bradley et al. 1973). In addition to Rosaceae, it has been recorded feeding on buffalobur nightshade (Solanaceae) in Washington (E. LaGasa, pers. comm.). A list of documented host plants is provided in Table 7.

Table 7: *Acleris holmiana* host plants

Family	**Genus/species**	**Common name**
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Cydonia* Mill.	cydonia
Rosaceae	*Malus domestica* Borkh.	apple
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Malus sylvestris* (L.) Mill.	European crab apple
Rosaceae	*Prunus* L.	[various]
Rosaceae	*Pyrus* L.	pear
Rosaceae	*Rosa* L.	rose
Rosaceae	*Rubus* L.	blackberry
Rosaceae	*Sorbus aucuparia* L.	European mountain ash
Solanaceae	*Solanum rostratum* Dunal	buffalobur nightshade

Distribution

A native of Europe and Asia Minor, *A. holmiana* was discovered in North America (British Columbia) in 1977. It was first detected in the U.S. (Washington) in 1994. It has apparently not spread outside of the Pacific Northwest.

Acleris minuta **(Robinson)**

Nomenclature

Acleris minuta (Robinson) (Tortricidae: Tortricinae: Tortricini)

Common names: yellow-headed fireworm, lesser apple leaf-folder

Synonyms: *cinderella* (*Tortrix*), *malivorana* (*Tortrix*), *vacciniivorana* (*Tortrix*), *variolana* (*Teras*)

Adult Recognition

Forewing color is uniform, and adults occur in two forms: a summer form which is yellow or orange, and an overwintering form, which is gray and is sometimes referred to as the "cinderella" form (Chapman and Lienk 1971). Males lack a forewing costal fold. FWL: 6.5-9.5 mm.

Male genitalia are characterized by long, thin socii and a wide valva with the distal end of the sacculus greatly produced into a rounded lobe. Female genitalia are characterized by a wide ostium, partially twisted ductus bursae, and scobinate signum.

Larval Morphology

Last instar larvae are greenish yellow and approximately 12 mm in length (Chapman and Lienk 1971).

Biology

The following life history information is summarized from Chapman and Lienk (1971). *Acleris minuta* completes three generations over most of its range although only two generations have been recorded in northern localities. Adults of the first and second generation are orange or yellow and are present in June and August. Adults of the third generation are gray and are present in October; this species overwinters as a third generation adult.

Females deposit eggs singly on bark in the spring or on leaves in the summer. Eggs hatch in 7-10 days and first instar larvae feed on the underside of leaves; later instars web together leaves to create a shelter or fold single leaves. In the past, this species has been recorded as a pest of apple, plum, and cranberry, and larvae feed mostly on plants in the families Ericaceae and Rosaceae (Table 8). The common name "fireworm" refers to the larval damage during heavy infestations, which has the appearance of being caused by fire. *Acleris minuta* is not currently considered a significant pest.

Distribution

Acleris minuta is widely distributed in eastern North America (Chapman and Lienk 1971).

Table 8: *Acleris minuta* host plants

Family	**Genus/species**	**Common name**
Ericaceae	*Calluna* Salisb.	heather
Ericaceae	*Kalmia angustifolia* L.	sheep laurel
Ericaceae	*Kalmia* L.	laurel
Ericaceae	*Vaccinium macrocarpon* Aiton	cranberry
Ericaceae	*Vaccinium* L.	blueberry
Myricaceae	*Myrica gale* L.	sweetgale
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus* L.	peach
Rosaceae	*Prunus* L.	plum
Rosaceae	*Pyrus* L.	pear
Salicaceae	*Salix* L.	willow

Acleris nivisellana **(Walsingham)**

Nomenclature

Acleris nivisellana (Walsingham) (Tortricidae: Tortricinae: Tortricini)

Common names: apple leaf twister

Adult Recognition

The following forewing pattern elements are consistent across most individuals: ground color white; large dark brown to black, triangular costal patch; brownish-yellow patch extending from the costal patch to the dorsal margin; and large patch of dark-brown to black on distal one-third of the wing. Males lack a forewing costal fold. FWL: 6.0-7.5 mm.

Acleris nivisellana can appear similar in forewing pattern to certain forms of other *Acleris* species, such as *A. forbesana* and *A. variegana*. Most other similar species lack the large patch of dark scales on the distal one-third of the wing that is found in *A. nivisellana*.

Larval Morphology

Mid- to late instar larvae are approximately 9-16 mm long. Abdominal color varies, while the head is brown to dark brown posteriorly and dark brown to black anteriorly. The prothoracic shield has two large posterolateral black spots, one on each side, and is otherwise concolorous with the abdomen (Chapman and Lienk 1971).

Biology

Acleris nivisellana appears to complete two generations per year, with adults present in June and again in mid-August and September. Larval feeding occurs in a silken chamber on the lower surface of leaves along the midrib. Larvae skeletonize the leaves and may partly sever the midrib, causing injured leaves to have a characteristic twisted appearance. Larvae have not been recorded feeding on fruit or other parts of the plant (Chapman and Lienk 1971). A list of documented host plants is provided in Table 9.

Table 9: *Acleris nivisellana* host plants

Family	**Genus/species**	**Common name**
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Physocarpus malvaceus* (Greene) Kuntze	mallow ninebark
Rosaceae	*Prunus pensylvanica* L. f.	pin cherry
Rosaceae	*Sorbus* L.	mountain ash
Rosaceae	*Sorbus scopulina* Greene	Greene's mountain ash

Distribution

Acleris nivisellana is distributed from the Northeastern United States across southern Canada to British Columbia and south to California. In the United States it is found mainly north of the 40th parallel (Powell 1964).

Acleris robinsoniana **(Forbes)**

Nomenclature

Acleris robinsoniana (Forbes) (Tortricidae: Tortricinae: Tortricini)

Common names: Robinson's acleris moth

Synonyms: *clemensiana* (*Peronea*)

Adult Recognition

The forewing pattern of *A. robinsoniana* varies greatly. Obraztsov (1963) described six distinct forms that were present in the type series; the nominate form is brown with a pale-yellow streak along the dorsal margin. In one of the more common forms the forewing is brown on the distal half and gray or white on the basal half with a dark-brown to black mark on the dorsum (Powell 1964). Males lack a forewing costal fold. FWL: 8.0-9.0 mm.

Acleris robinsoniana may appear similar to other species of *Acleris*, specifically certain forms of *A. fragariana*, *A. inana*, *A. keiferi*, *A. maculidorsana*, and *A. variegana*. A genitalic dissection can be used to confirm species identity.

Larval Morphology

Larvae are yellowish or light green. Late instar individuals have a brown head, a dark-brown prothoracic shield, and brown to dark-brown legs. Earlier instars lack these markings and the prothoracic shield and legs are concolorous with the abdomen (Obraztsov 1963, Powell 1964).

Biology

Acleris robinsoniana completes one or two generations per year (Powell 1964). Adults are usually present in May or June. Early instar larvae fold or tie newly developing leaves and feed on the upper surface. Later instars fold leaves and consume the apical half. Individuals overwinter as adults. Larvae of this species have only been recorded feeding on California wildrose and quaking aspen (Table 10) (Powell 1964).

Table 10: *Acleris robinsoniana* host plants

Family	Genus/species	Common name
Rosaceae	*Rosa californica* Cham. & Schltdl.	California wildrose
Salicaceae	*Populus tremuloides* Michx.	quaking aspen

Distribution

Acleris robinsoniana is distributed from the northeastern United States across southern Canada to British Columbia and south to California (Powell 1964).

***Acleris senescens* (Zeller)**

Nomenclature

Acleris senescens (Zeller) (Tortricidae: Tortricinae: Tortricini)

Adult Recognition

Adults have elongate, narrow forewings. Several different forms exist, although most are a shade of gray with black lines and spots for markings (Powell 1964). Males lack a forewing costal fold. FWL: 10.0-12.5 mm. Forewing pattern is similar to certain forms of several other *Acleris* species, such as *A. chalybeana* and *A. hastiana*. A genitalic dissection can be used to confirm identity.

Biology

Adults are present in early spring and late fall in California, suggesting two generations per year (Powell 1964). Life history information for this species is undocumented. Larvae of *A. senescens* have been recorded feeding on plants in the families Betulaceae, Rosaceae, and Salicaceae.

Distribution

Acleris senescens is distributed along the Pacific Coast of North America from British Columbia south to California (Powell 1964).

Table 11: *Acleris senescens* host plants

Family	Genus/species	Common name
Betulaceae	*Alnus rubra* Bong.	red alder
Betulaceae	*Betula* L.	birch
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus virginiana* L.	chokecherry
Salicaceae	*Populus tremuloides* Michx.	quaking aspen
Salicaceae	*Salix hookeriana* Barratt ex Hook.	dune willow
Salicaceae	*Salix* L.	willow
Salicaceae	*Salix lasiolepis* Benth.	arroyo willow

***Acleris variana* (Fernald)**

Nomenclature

Acleris variana (Fernald) (Tortricidae: Tortricinae: Tortricini)

Common names: eastern blackheaded budworm

Synonyms: *angusana* (*Peronea*)

Adult Recognition

The forewing pattern of *A. variana* is quite variable and many forms are shared with *A. gloverana*. Powell (1962) describes eight different forms for *A. gloverana* and an additional form for *A. variana*. The most common forms are illustrated here. Males lack a forewing costal fold. FWL: 7.5-9.5 mm.

Acleris variana is nearly identical to *A. gloverana* in both biology and morphology. The two species are geographically separated: *A. gloverana* occurs in western North America and *A. variana* occurs in eastern North America. Powell (1962) outlined minor genitalic differences that can be used to separate the two species. In males, the sacculus is broader and less elongate in *A. gloverana*, while it is narrower and more elongate in *A. variana*. In *A. gloverana* females, the sterigma is broad, with lobes connected, and a cestum is absent. In *A. variana* females, the sterigma is narrow with the lobes separate, and a weakly sclerotized cestum is present. In addition, females of *A. variana* have large abdominal scale tufts that are used to cover the eggs; these are lacking in *A. gloverana* females.

Larval Morphology

Larvae are approximately 11-15 mm in length and are bright green with a black head. The black head is lost in the final instar, where both the head and prothoracic shield are brown. An anal comb is present with 6-10 teeth (EPPO 1997, Powell 1962).

Biology

The following life history information is summarized from Otvos (1991), Powell (1962), and Powell and Opler (2009). *Acleris variana* completes one generation per year. Adults are present in August and September. The *A. variana* life cycle is nearly identical to that of *A. gloverana*.

Females lay eggs on the underside of needles in the upper section of host trees. Eggs overwinter until the following spring, and larvae hatch starting in mid-May. First intars feed inside opening buds, mid-instars web together needles to create a nest, and last instars are free-feeding. Males and 50% of females complete 4 larval instars while the remaining females complete 5 instars. Pupation occurs in webbed needles. Adults eclose in approximately two weeks. Larval damage is similar to that of *A. gloverana*, although outbreaks of *A. variana* do not tend to cause tree mortality. Periods of low rainfall tend to trigger increases in *A. variana* populations. A list of documented host plants is provided in Table 12.

Table 12: *Acleris variana* host plants

Family	**Genus/species**	**Common name**
Cupressaceae	*Thuja plicata* Donn ex D. Don	western redcedar
Pinaceae	*Abies alba* Mill.	silver fir
Pinaceae	*Abies amabilis* (Douglas ex Louden) Douglas ex Forbes	Pacific silver fir
Pinaceae	*Abies balsamea* (L.) Mill.	balsam fir
Pinaceae	*Abies grandis* (Douglas ex D. Don) Lindl.	grand fir
Pinaceae	*Abies lasiocarpa* (Hook.) Nutt.	subalpine fir
Pinaceae	*Abies* Mill.	fir
Pinaceae	Larix Mill.	larch
Pinaceae	*Larix occidentalis* Nutt.	western larch
Pinaceae	*Picea* A. Dietr.	spruce

Table 12: *Acleris variana* host plants

Family	**Genus/species**	**Common name**
Pinaceae	*Picea abies* (L.) Karst.	Norway spruce
Pinaceae	*Picea engelmannii* Parry ex Engelm.	Engelmann spruce
Pinaceae	*Picea glauca* (Moench) Voss	white spruce
Pinaceae	*Picea mariana* (Mill.) Britton, Sterns & Poggenb.	black spruce
Pinaceae	*Picea pungens* Engelm.	blue spruce
Pinaceae	*Picea rubens* Sarg.	red spruce
Pinaceae	*Picea sitchensis* (Bong.) Carriere	Sitka spruce
Pinaceae	*Pinus contorta* Douglas ex Loudon	lodgepole pine
Pinaceae	*Pseudostuga* Carriere	Douglas-fir
Pinaceae	*Pseudotsuga menziesii* (Mirb.) Franco	Douglas-fir
Pinaceae	*Tsuga canadensis* (L.) Carriere	eastern hemlock
Pinaceae	*Tsuga heterophylla* (Raf.) Sarg.	western hemlock
Pinaceae	*Tsuga mertensiana* (Bong.) Carriere	mountain hemlock
Salicaceae	*Populus balsamifera* L.	balsam poplar

Distribution

Acleris variana is distributed in coniferous forests from eastern Canada and the northeastern United States west to Alberta (Powell 1962, Powell and Opler 2009).

***Acleris variegana* (Denis & Schiffermüller)**

Nomenclature

Acleris variegana (Denis & Schiffermüller) (Tortricidae: Tortricinae: Tortricini)

Common names: garden rose tortrix

Synonyms: *albana* (*Peronea*), *alpicolana* (form), *argentana* (form), *asperana* (*Pyralis*), *blandiana* (*Tortrix*), *brunneana* (form), *caeruleoatrana* (ab.), *cirrana* (*Peronea*), *costimaculana* (*Peronea*), *fuscana* (form), *insignata* (*Tortrix*), *nyctemerana* (*Tortrix*), *osbeckiana* (*Tortrix*), *uniformis* (ab.)

Adult Recognition

The forewing pattern of *A. variegana* is variable; Bradley et al. (1973) lists eight forms as occuring in the United Kingdom. Powell (1964) describes three distinct phenotypes found in western North America: 1) forewing white on inner half with a dark dorsal triangle, outer half dark-purplish; 2) forewing divided into white basal and blue-black or purplish distal area; 3) and forewing ground color tan with a basal dorsal triangle and purplish outer costal triangle. Males lack a forewing costal fold. FWL: 7.0-10.0 mm. *Acleris variegana* may appear similar to certain forms of other *Acleris* species, including *A. keiferi*, *A. maculidorsana*, and *A. robinsoniana* in North America and *A. permutana* in Europe. A genitalic dissection can be used to confirm identity.

Larval Morphology

Larvae are yellowish or light green. The head is brown and the prothoracic shield is concolorous with the abdomen. Legs are brown or pale. Most individuals exhibit a black spot located laterally on the head and a dark posterolateral mark on each side of the prothoracic shield (Bradley et al. 1973).

Biology

Acleris variegana completes two generations per year in North America. Adults fly from April to July and again from August to November. Overwintering occurs as a second generation adult (Powell 1964).

Eggs are laid singly or in small batches on leaves of the host plant. Early instar larvae tie together two leaves and skeletonize them from the inside; later instars consume the entire leaf. Pupation occurs in the larval shelter or among dead leaves on the ground (Bradley et al. 1973, Powell 1964). Larvae have been recorded feeding on a variety of host plants, primarily those in the family Rosaceae (Table 13).

Distribution

Acleris variegana is widely distributed throughout Europe, northwestern Africa, central Asia, and China. It was first reported from North America in the early 1900's where it is present on the East and West Coasts (Powell and Opler 2009).

Table 13: *Acleris variegana* host plants

Family	**Genus/species**	**Common name**
Berberidaceae	*Berberis vulgaris* L.	common barberry
Betulaceae	*Carpinus betulus* L.	European hornbeam
Betulaceae	*Corylus avellana* L.	common filbert
Ericaceae	*Vaccinium* L.	blueberry
Rosacae	*Sanguisorba minor* Scop.	small burnet
Rosaceae	*Cotoneaster* Medik.	cotoneaster
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Dasiphora fruticosa* (L.) Rydb. ssp. *floribunda* (Pursh) Kartesz	shrubby cinquefoil
Rosaceae	*Malus domestica* Borkh.	apple
Rosaceae	*Malus sylvestris* (L.) Mill.	European crab apple
Rosaceae	*Prunus armeniaca* L.	apricot
Rosaceae	*Prunus avium* (L.) L.	sweet cherry
Rosaceae	*Prunus domestica* L.	European plum
Rosaceae	*Prunus* L.	
Rosaceae	*Prunus spinosa* L.	blackthorn
Rosaceae	*Pyracantha* M. Roem.	firethorn
Rosaceae	*Pyrus communis* L.	common pear
Rosaceae	*Pyrus* L.	pear
Rosaceae	*Rosa californica* Cham. & Schltdl.	California wildrose
Rosaceae	*Rosa* L.	rose
Rosaceae	*Rubus idaeus* L.	American red raspberry
Rosaceae	*Rubus* L.	blackberry
Rosaceae	*Rubus ursinus* Cham. & Schltdl.	California blackberry
Rosaceae	*Sorbus aucuparia* L.	European mountain ash
Solanaceae	*Solanum lycopersicum* L. var. *lycopersicum*	garden tomato
Ulmaceae	*Ulmus* L.	elm

Tortricidae: Tortricinae: Cochylini

Overview

The tribe Cochylini contains approximately 1,000 described species in 80 genera (Baixeras et al. 2008). Its members are present worldwide, although the greatest species diversity occurs in the Holarctic and Neotropics. The group is defined by wing venation, wing pattern, and to a lesser extent, genitalic characters (Horak and Brown 1991). The wing pattern of many species is similar, with yellow or white ground color and one or two distinct, reddish brown fasciae. Often the median fascia only extends partway from dorsum to costa, creating the appearance of a dorsal patch. Male genitalia are characterized by triangular or rectangular valvae and a very large aedeagus. Female genitalia are characterized by a short, broad, ductus bursae, and a corpus bursae with numerous wrinkles, sclerotizations, or spines.

The group contains a number of economically important species. In much of the economic literature it is still treated as a distinct family or subfamily (Cochylidae or Cochylinae), although it is currently considered a tribe within the Tortricinae. Brown (2006) outlines the history of placement of this group. Only two species are treated here. *Eupoecilia ambiguella* is an important pest of grapes in the Palearctic, and its larvae cause damage similar to that of *Lobesia botrana*. *Henricus umbrabasana* is a California species that is commonly collected as a non-target in *Epiphyas postvittana* traps.

Larvae are primarily internal feeders in seeds, stalks, and roots. Larval morphology for the group is unusual in that larvae share the following character states with those of Grapholitini (Olethreutinae): D1 and SD1 are on the same pinaculum on A9, and the L pinaculum on T1 is enlarged and extends beneath the spiracle. In some species (e.g., *E. ambiguella*), the L-group does not extend past (posterior to) the spiracle as it does in many of the olethreutines with a similar state (*Cryptophlebia*, some *Cydia*, *Ecdytolopha*, *Gymnandrosoma*, and *Thaumatotibia*). Other larval characters include an anal comb with 2-10 teeth and the L pinaculum on A9 bisetose.

Cochylini larvae are commonly intercepted at U.S. ports of entry on products arriving from Central America. One commonly recorded host is monkeypod (*Pithecellobium dulce*) from Mexico (J. Brown, pers. comm.).

Eupoecilia ambiguella **(Hübner)**

Nomenclature

Eupoecilia ambiguella (Hübner) (Tortricidae: Tortricinae: Cochylini)

Common names: European grape berry moth (preferred), European grape vine moth, grape berry moth

Synonyms: *omphaciella* (*Tinea*), *roserana* (*Tortrix*), *turbinaris* (*Clysia*)

Adult Recognition

The forewing is yellow or yellowish orange with a well-defined dark-brown to black median fascia. Males and females exhibit no sexual dimorphism in wing pattern although females may be slightly larger than males. Males lack a forewing costal fold. FWL: 6.0-7.5 mm. Male genitalia are distinguished by a reduced uncus, short socii, prominent transtilla, distally triangular valva, and large aedeagus. Female genitalia are distinguished by a broad, short ductus burase and a corpus bursae with numerous sclerotizations and spines (Gilligan and Epstein 2011).

Adults may be confused with other species of *Eupoecilia* or European Cochylini, although *E. ambiguella* is the only cochylid commonly associated with grape. A genitalic dissection can be used to confirm *E. ambiguella* identity.

Larval Morphology

Late instar larvae are approximately 10-12 mm in length. The head, prothoracic shield, and legs are dark brown to black. Body color varies from brown to yellow and green. Pinacula are large, conspicuous, and brown. The anal shield is pale brown (Gilligan and Epstein 2011).

Larvae cause damage similar to *Lobesia botrana* and the two species can be found sympatrically. Other tortricid grape pests include: *Argyrotaenia franciscana*, *Argyrotaenia ljungiana*, *Epiphyas postvittana*, *Paralobesia viteana*, *Platynota stultana*, and *Proeulia* spp. Larvae of *E. ambiguella* can be separated from the larvae of other tortricid grape-feeding pests by the L-group (or prespiracular) pinaculum on T1, which extends horizonally beneath the spiracle in *E. ambiguella*.

Biology

The life cycle of *E. ambiguella* is similar to that of *Lobesia botrana*, with the exception of two generations for *E. ambiguella* versus three or more generations for *L. botrana*. Over most of its range, adults are present in May and June for the first generation and again in August and September for the second generation (Bradley et al. 1973, EPPO 2007, Roehrich and Boller 1991).

Females deposit eggs singly on buds, pedicels, and flowers during the first generation, and on grape berries during the second generation. Early instar larvae burrow into the buds or berries and feed internally; later instars web together buds or berries, and a single larva can feed on up to a dozen berries. Pupation occurs in leaves for the first generation and under bark for the second generation. Overwintering occurs as a second generation pupa. Development time is highly dependent on temperature and humidity. The optimum relative humidity level for development is 70% or higher; eggs will fail to hatch at low relative humidity levels (EPPO 2007, Roehrich and Boller 1991).

Economic losses on grape are caused by direct feeding damage and secondary infections. Feeding damage is similar to that of *Lobesia botrana*. Larvae of the first generation cause minor damage by feeding on flower buds, while those of the second generation cause the most damage by feeding on grape berries. The most significant losses are due to secondary infection of feeding sites on berries and clusters by *Botrytis cinerea*. Economic thresholds vary with the type of grape and cultivar. Although grape is the most economically important host, *E. ambiguella* has been recorded from plants in several families (Table 14).

Table 14: *Eupoecilia ambiguella* host plants

Family	**Genus/species**	**Common name**
Aceraceae	*Acer campestre* L.	hedge maple
Araliaceae	*Eleutherococcus* Maxim.	ginseng
Araliaceae	*Hedera helix* L.	English ivy
Araliaceae	*Hedera* L.	ivy
Caprifoliaceae	*Lonicera* L.	honeysuckle
Caprifoliaceae	*Lonicera periclymenum* L.	European honeysuckle
Caprifoliaceae	*Lonicera ramosissima* Franch. & Sav. ex Maxim.	
Caprifoliaceae	*Symphoricarpos* Dunham.	snowberry
Caprifoliaceae	*Viburnum* L.	viburnum
Cornaceae	*Cornus* L.	dogwood
Cornaceae	*Cornus mas* L.	Cornelian cherry
Cuscutaceae	*Cuscuta* L.	dodder
Cuscutaceae	*Cuscuta reflexa* Roxb.	giant dodder
Grossulariaceae	*Ribes* L.	currant
Oleaceae	*Ligustrum* L.	privet
Oleaceae	*Syringa* X *persica* L.	Persian lilac
Rhamnaceae	*Frangula alnus* Mill.	glossy buckthorn
Rhamnaceae	*Rhamnus* L.	buckthorn
Rosaceae	*Prunus* L.	
Vitaceae	*Parthenocissus quinquefolia* (L.) Planch.	Virginia creeper
Vitaceae	*Vitis vinifera* L.	wine grape

Distribution

Eupoecilia ambiguella is widely distributed across the Palearctic, where it is more common in cooler and humid climates. It has been reported from Brazil (EPPO 2007), but this record could not be verified. It is not considered established outside of Europe and Asia.

Henricus umbrabasana **(Kearfott)**

Nomenclature

Henricus umbrabasana (Kearfott) (Tortricidae: Tortricinae: Cochylini)

Synonyms: *umbrabasana* (*Commophila*)

Adult Recognition

Forewings are yellowish white with a well-defined brown to dark-brown basal patch and a black spot in the median fascia. Males lack a forewing costal fold. FWL: 7.5-9.0 mm.

In California, adult *H. umbrabasana* are commonly found in *Epiphyas postvittana* pheromone traps. Distinguishing between individuals of these two species by wing pattern may be difficult if the forewings are heavily stained from sticky traps. The two species can be separated by the male forewing costal fold: present in *E. postvittana*, absent in *H. umbrabasana*.

Larval Morphology

Mature larvae are dark maroon with a brown or black head and black prothoracic shield.

Biology

Henricus umbrabasana completes a single generation per year. Adults are present from May through September.

Eggs are laid during the summer or fall and overwinter until the next spring. Larvae emerge in March and feed on young leaves and shoots of *Quercus* (Table 15) inside a shelter covered in frass and debris. Pupation occurs within a cocoon covered in frass and debris (Opler 1974).

Table 15: *Henricus umbrabasana* host plants

Family	**Genus/species**	**Common name**
Fagaceae	*Quercus agrifolia* Nee	California live oak
Fagaceae	*Quercus chrysolepis* Liebm.	canyon live oak
Fagaceae	*Quercus lobata* Nee	valley oak

Distribution

Henricus umbrabasana is recorded from California and southwestern Washington (E. LaGasa, pers. comm.). It likely also occurs in Oregon.

Tortricidae: Tortricinae: Cnephasiini

Overview

The Cnephasiini contains approximately 250 species that are distributed throughout the Holarctic (Horak and Brown 1991). Forewing pattern varies within the tribe, but many North American representatives are yellow, gray, or white. Male genitalia are characterized by an uncus covered in small spines. Female genitalia are characterized by papillae anales that are modified to cover eggs with debris, and a band-shaped, poorly-defined signum. This is the only tribe in the Tortricinae in which most of its members have a 12-carbon sex pheromone, a condition usually found in the Olethreutinae (Roelofs and Brown 1982).

North American representatives of the genus include *Cnephasia*, *Decodes*, and *Eana*. *Cnephasia* is a Palearctic genus with three members introduced into North America: *C. asseclana* and *C. stephensiana*, and *C. longana* (Razowski 1987, 2002). *Cnephasia longana* is a pest of cereal crops in Europe and has been reported as a pest of strawberries in the Pacific Northwest (Glas 1991). *Decodes* contains 18 species, many of which are present in the American Southwest and northern Mexico (Powell and Opler 2009). *Decodes basiplagana* and *D. fragariana* are oak feeders; the former is present in the East while the latter is present in the West. *Eana* is a small genus with members in the Palearctic and Nearctic. Most species are have unmarked, white forewings; however, none are treated here.

Cnephasiini larvae are occasionally intercepted at U.S. ports of entry from Europe. It is assumed that a majority of these are *Cnephasia*, but a genus-level identification is rarely confirmed.

***Cnephasia longana* (Haworth)**

Nomenclature

Cnephasia longana (Haworth) (Tortricidae: Tortricinae: Cnephasiini)

Common names: omnivorous leaf-tier, strawberry fruitworm

Synonyms: *cadizensis* (form), *capillana* (*Sphaleroptera*), *egenana* (*Tortrix*), *expallidana* (*Tortrix*), *gratana* (*Sciaphila*), *ictericana* (*Tortrix*), *insolatana* (*Tortrix*), *loeviana* (*Sciaphila*), *luridalbana* (*Tortrix*), *lutosana* (*Tortrix*), *minor* (form), *ongana* (*Cnephasia*), *stratana* (*Sciaphila*)

Adult Recognition

Cnephasia longana is sexually dimorphic: males are a uniform white to yellowish brown while females are variably marked with light to dark brown. Some females have a strong fasciate pattern. Males lack a forewing costal fold. FWL: 7.5-10.8 mm.

The yellowish, narrow, elongate forewings separate this species from most of the other Tortricidae covered here. Other Palearctic *Cnephasia*, such as *C. gueneeana*, are similar, and a genitalic dissection can be used to confirm identity.

Larval Morphology

Late instar larvae are greenish brown or yellowish green with pale lateral lines and are approximately 14-18 mm in length. The head is pale brown with dark brown margins and the prothoracic shield is concolorous with the rest of the body (Bradley et al. 1973, CPC 2007).

Biology

The following life history information is summarized from Glas (1991), Powell (1964), and Powell and Opler (2009). Adults are present from late March to early July in North America. Females lay eggs singly or in small batches in depressions or cracks in the bark of trees and cover them with bits of debris. After hatching, first instar larvae search out suitable cracks or crevices in bark, construct a silk hibernaculum, and hibernate for the remainder of the summer and following winter. Larvae leave the hibernaculum in the spring and disperse to nearby herbaceous plants by ballooning with the aid of silk threads. After encountering a suitable host, larvae mine within leaves close to the ground. Later instars web terminal parts of the plant, including the shoots, buds, and/or flowers. In some instances larvae may bore into fruit (such as strawberries) causing economic damage.

Cnephasia longana larvae have been recorded feeding on a large number of herbaceous plants. In Europe, the larvae can be a pest of cereal crops (Glas 1991). In Oregon and Washington, *C. longana* larvae have been reported as a pest of strawberries (Powell 1964). Larvae have been recorded feeding on plants in more than 20 families (Table 16).

Table 16: *Cnephasia longana* host plants

Family	**Genus/species**	**Common name**
Apiaceae	*Selinum carvifolium* Crantz	
Asteraceae	*Achillea* L.	yarrow
Asteraceae	*Achillea millefolium* L.	common yarrow
Asteraceae	*Agoseris heterophylla* (Nutt.) Greene	annual agoseris
Asteraceae	*Anthemis cotula* L.	stinking chamomile
Asteraceae	*Anthemis* L.	chamomile
Asteraceae	*Aster* L.	aster
Asteraceae	*Centaurea* L.	knapweed
Asteraceae	*Chrysanthemum* L.	daisy
Asteraceae	*Cirsium* Mill.	thistle
Asteraceae	*Erigeron glaucus* Ker Gawl.	seaside fleabane
Asteraceae	*Hypochaeris* L.	cat's ear
Asteraceae	*Leucanthemum vulgare* Lam.	oxeye daisy
Asteraceae	*Ligularia* Cass.	summer ragwort
Asteraceae	*Senecio jacobaea* L.	stinking willie
Asteraceae	*Sonchus arvensis* L.	field sowthistle
Asteraceae	*Tripolium pannonicum* (Jacq.) Dobrocz.	sea aster
Asteraceae	*Wyethia angustifolia* (DC.) Nutt.	California compassplant
Berberidaceae	*Mahonia pinnata* (Lag.) Fedde	wavyleaf barberry
Boraginaceae	*Amsinckia* Lehm.	fiddleneck
Boraginaceae	*Amsinckia menziesii* (Lehm.) A. Nelson & J.F. Macbr. var. *intermedia* (Fisch. & C.A. Mey.) Ganders	common fiddleneck
Caryophyllaceae	*Lychnis* L.	campion
Convolvulaceae	*Convolvulus* L.	bindweed
Crassulaceae	*Sempervivum tectorum* L.	common houseleek
Fabaceae	*Lupinus albifrons* Benth.	silver lupine
Fabaceae	*Lupinus* L.	lupine

Table 16: *Cnephasia longana* host plants

Family	**Genus/species**	**Common name**
Fabaceae	*Psoralea* L.	
Fabaceae	*Vicia americana* Muhl. ex Willd.	American vetch
Fabaceae	*Vicia* L.	vetch
Geraniaceae	*Erodium cicutarium* (L.) L'Her. ex Aiton	redstem stork's bill
Geraniaceae	*Geranium* L.	geranium
Hydrophyllaceae	*Phacelia distans* Benth.	distant phacelia
Lamiaceae	*Stachys* L.	hedgenettle
Linaceae	*Linum* L.	flax
Onagraceae	*Camissonia* Link	suncup
Papaveraceae	*Eschscholzia californica* Cham.	California poppy
Plantaginaceae	*Plantago lanceolata* L.	narrowleaf plantain
Plumbaginaceae	*Armeria maritima* (Mill.) Willd.	thrift seapink
Polygonaceae	*Eriogonum latifolium* Sm.	seaside buckwheat
Polygonaceae	*Eriogonum* Michx.	buckwheat
Polygonaceae	*Rumex acetosella* L.	common sheep sorrel
Polygonaceae	*Rumex* L.	dock
Ranunculaceae	*Ranunculus* L.	buttercup
Rosaceae	*Fragaria* L.	strawberry
Rosaceae	*Prunus armeniaca* L.	apricot
Scrophulariaceae	*Castilleja* Mutis ex L. f.	Indian paintbrush
Scrophulariaceae	*Collinsia multicolor* Lindl. & Paxton	San Francisco blue eyed Mary
Scrophulariaceae	*Scrophularia californica* Cham. & Schltdl.	California figwort
Violaceae	*Viola* L.	violet
Zygophyllaceae	*Fagonia* L.	fagonbush

Distribution

Cnephasia longana is native to the Palearctic where it is widely distributed across Europe, northwestern Africa, and Asia. It was introduced into western North America in the late 1920's, where it is currently present in southern British Columbia, Washington, Oregon, and California (Powell 1964, Powell and Opler 2009).

***Decodes basiplagana* (Walsingham)**

Nomenclature

Decodes basiplagana (Walsingham) (Tortricidae: Tortricinae: Cnephasiini)

Adult Recognition

Forewings are gray with a strongly arched costa. Forewing pattern varies greatly, with some individuals expressing well-defined dark-gray and black fasciate markings, and others being unmarked. Males lack a forewing costal fold. FWL: 8.0-9.0 mm.

Decodes fragariana is similar, but males have an aedeagus that is curved more than 90 degrees and females lack a signum in the corpus burase. *Decodes montanus* is similar to both species, but the male aedeagus is more strongly curved than in *D. fragariana* and females of *D. montanus* have a band of dark scales on the seventh sternite that is lacking in the other two species (Powell 1964). Wing pattern is not diagnostic and cannot be used to reliably separate the *Decodes* species listed here.

Biology

The life history of *D. basiplagana* is unknown, although it is assumed to be similar to that of *D. fragariana*. Larvae have been recorded feeding on oak (Table 17.)

Table 17: *Decodes basiplagana* host plants

Family	**Genus/species**	**Common name**
Fagaceae	*Quercus* L.	oak
Fagaceae	*Quercus lobata* Nee	valley oak

Distribution

Decodes basiplagana is recorded from the eastern U.S. and ranges west into central Texas, Colorado, Utah, Arizona, and central California (Powell 1964, Powell and Opler 2009).

***Decodes fragariana* (Busck)**

Nomenclature

Decodes fragariana (Busck) (Tortricidae: Tortricinae: Cnephasiini)

Synonyms: *elapsa* (*Peronea*)

Adult Recognition

Forewings are gray with a strongly arched costa. Forewing pattern varies, with some specimens marked with well-defined dark-gray fasciae and others unmarked. Males lack a forewing costal fold. FWL: 6.0-8.5 mm.

Decodes basiplagana is similar, but males have an aedeagus that is curved less than 90 degrees and females have a signum in the corpus burase. *Decodes montanus* is similar to both species, but the male aedeagus is more strongly curved than in *D. fragariana* and females of *D. montanus* have a band of dark scales on the seventh sternite that is lacking in the other two species (Powell 1964). Wing pattern is not diagnostic and cannot be used to reliably separate the *Decodes* species listed here.

Biology

Decodes fragariana completes one generation per year in California. Adults are present in late August through October.

Females deposit eggs on rough twigs or bark and cover them with debris. The eggs enter diapause and overwinter until the following spring. Larvae hatch in the spring, construct a silken feeding tube on the bottom of a leaf or between two leaves, and feed on new growth until mature. Mature larvae drop to the ground and pupae in silk cocoons covered in soil or other particles. Adults eclose in the fall (Powell 1964, Powell and Opler 2009).

This species was originally described as feeding on strawberry; however, *D. fragariana* has subsequently been recorded feeding only on live oak (Table 18). Powell and Opler (2009) suggest that the individuals found in association with strawberry were simply pupating after having completed larval development on nearby oak trees.

Distribution

Decodes fragariana ranges from British Columbia south to San Diego, California and east to Utah, Colorado, and New Mexico (Powell 1964, Powell and Opler 2009).

Table 18: *Decodes fragariana* host plants

Family	**Genus/species**	**Common name**
Fagaceae	[leaf litter]	
Fagaceae	*Quercus agrifolia* Nee	California live oak
Fagaceae	*Quercus douglasii* Hook. & Arn.	blue oak
Fagaceae	*Quercus dumosa* Nutt.	coastal sage scrub oak
Fagaceae	*Quercus durata* Jeps.	leather oak
Fagaceae	*Quercus garryana* Douglas ex Hook.	Oregon white oak
Fagaceae	*Quercus kelloggii* Newb.	California black oak
Fagaceae	*Quercus* L.	oak
Fagaceae	*Quercus lobata* Nee	valley oak
Fagaceae	*Quercus turbinella* Greene	Sonoran scrub oak
Fagaceae	*Quercus wislizeni* A. DC.	interior live oak
[Rosaceae]	[*Fragaria* L.]	[strawberry]

Tortricidae: Tortricinae: Archipini

Overview

The Archipini is the largest tribe within the Tortricinae, with more than 1,600 described species in 150 genera (Baixeras et al. 2008). Its members are present in all regions but are uncommon in the neotropics. Forewing pattern varies greatly; however, a large number of species have a generalized fasciate pattern, with a well-expressed basal and median fascia, and a dark costal spot. Male genitalia are characterized by an elongate, variably-shaped uncus, well-developed gnathos, and rounded, sometimes membranous or folded, valvae. Females of nearly all Archipini have a daggar-shaped signum with a capitulum; this character is the only true synapomorphy for the group (Horak and Brown 1991).

This group contains the most pest species of any tortricid tribe. Larvae are mostly polyphagous, external feeders, and some species, such as the light brown apple moth

(*Epiphyas postvittana*), have been recorded feeding on more than 500 species of plants (Brown et al. 2010). Larvae of many species have few diagnostic morphological characters, and it may be difficult to separate larvae of various genera such as *Archips*, *Argyrotaenia*, *Clepsis*, etc. As such, many larval interceptions at U.S. ports of entry are not identified below the subfamily level except in certain cases of specific host/origin associations.

Adoxophyes **Meyrick**

Overview

Adoxophyes is a genus in the Archipini that contains approximately 50 species (Baixeras et al. 2008). Most species diversity occurs in Asia and Australasia, although the genus is present in all regions except the neotropics. Forewing markings are consistent across the genus, with many species exhibiting a generalized fasciate pattern with horizontal reticulations. Genitalic differences between species are often minor, and it is difficult to separate closely related taxa using only morphology (Sakamaki and Hayakawa 2004).

The most economically important species is *A. orana*, which is a major pest of fruit crops in the Palearctic. Larvae are highly polyphagous but cause most economic damage to apple and pear. Several subspecies and related species are present in Asia, where the tea-feeding populations are generally thought to be distinct from the apple-feeding populations (Lee et al. 2005, Zhou et al. 1997).

It is not known how many *Adoxophyes* larvae arrive at U.S. ports due to a lack of diagnostic characters for the genus. Molecular diagnostics are necessary to identify larvae in most cases. Four species are treated here, including two Nearctic non-targets.

Adoxophyes furcatana **(Walker)**

Nomenclature

Adoxophyes furcatana (Walker) (Tortricidae: Tortricinae: Archipini)

Adult Recognition

Adults are pale yellow with light-brown markings. Hindwings are white. Males have a forewing costal fold. FWL: 7.5-10.5 mm.

Adoxophyes negundana and *A. furcatana* are the only two representatives of the genus *Adoxophyes* in the Nearctic. These two species are difficult to separate using forewing pattern: in general, the median fascia is narrower in *A. furcatana*, although this character does not appear to be consistent across all individuals. Freeman (1958) states that male genitalia of *A. furcatana* have more cornuti (greater than four) and less sharply elbowed gnathos arms than those of *A. negundana*.

Both species of Nearctic *Adoxophyes* are similar to *A. orana* and might be confused with this Palearctic species if it were discovered in North America. It is not known if Nearctic *Adoxophyes* are attracted to *A. orana* pheromone.

Larval Morphology

Larval morphology is undocumented for this species.

Biology

Adults are present June to August. Larvae feed in rolled sycamore leaves (Table 19) (Freeman 1958).

Table 19: *Adoxophyes furcatana* host plants

Family	**Genus/species**	**Common name**
Platanaceae	*Platanus* L.	sycamore

Distribution

Adoxophyes furcatana is widely distributed in eastern North America (Freeman 1958).

Adoxophyes honmai **Yasuda**

Nomenclature

Adoxophyes honmai Yasuda (Tortricidae: Tortricinae: Archipini)

Common names: smaller tea tortrix

Adult Recognition

Forewing pattern is similar to *A. orana* and other Asian *Adoxophyes*, and the various species cannot be separated based solely on wing pattern. Although tea pests are not a top concern for U.S. agriculture, this species is treated here as part of the *A. orana* complex in Asia. FWL: 8.0-9.5 mm.

Genitalia of *A. honmai* are similar to those of other *Adoxophyes*. According to Yasuda (1998), male *A. honmai* can be distinguished from *A. orana* based on the following combination of characters: uncus dorsally truncate and narrow; apical membranous projection on uncus small and shrunken; brachiola (membranous apical projection off valva) “finger-shaped;” and transtilla lobes not touching at the proximal ends. Females are distinguished from *A. orana* by a smaller bursa copulatrix and longer ductus bursae.

Adoxophyes honmai is similar to other species of *Adoxophyes* in Asia which include: *A. dubia* (Japan), *A. orana beijingensis* (China), *A. orana fasciata* (Japan), *A. orana orana* (China, Korea, and Europe), and *A. privatana* (China). All are difficult to separate and molecular diagnostics may be required for a positive identification (Lee et al. 2005, Yasuda 1998, Zhou et al. 1997).

Larval Morphology

Similar to other *Adoxophyes*, late instar larvae vary in body color and are approximately 20 mm in length. Head and prothoracic shield are black in early instars and become brown to yellowish brown in later instars. Legs are unmarked (Sakamaki and Hayakawa 2004).

Sakamaki and Hayakawa (2004) attempted to separate the immature stages of the three Japanese species of *Adoxophyes* using morphology but the differences they found are too minor to be consistently diagnostic.

Biology

Adoxophyes honmai completes 4-5 generations per year in Japan. Adults are present April through June, June and July, July and August, and August through November. Adults are capable of long dispersal and can fly up to 5 km per night (Shirai and Kosugi 2000).

Females lay eggs in masses on the underside of tea leaves. First instar larvae disperse to feed on young leaves or shoot tips. Later instars web together leaves to create a nest in which pupation occurs. Larvae complete 5-6 instars and overwintering occurs in the larval stage although there is no diapause (Tamaki 1991).

Adoxophyes honmai is the most common pest of tea in Japan along with *Homona magnanima*. Larvae of *A. honmai* cause damage that is uniformly distributed on the tea bush, where the damage caused by *H. magnanima* is restricted to one area of the plant (Tamaki 1991). Sakamaki and Hayakawa (2004) recorded *A. dubia* as feeding on tea in Japan, sometimes occuring sympatrically with *A. honmai*. In addition to tea, *A. honmai* has been recorded feed on host plants in several other families (Table 20).

Table 20: *Adoxophyes honmai* host plants

Family	**Genus/species**	**Common name**
Araliaceae	*Hedera thombea* (Miq.) Bean	
Asteraceae	*Solidago altissima* L.	Canada goldenrod
Caprifoliaceae	*Viburnum suspensum* Lindl.	viburnum
Fagaceae	*Quercus phillyraeoides* A. Gray	ubame oak
Magnoliaceae	*Michelia champaca* L.	michelia
Myrtaceae	*Eucalyptus* L'Her.	gum
Rosaceae	[unspecified]	cherry
Theaceae	*Camellia sinensis* (L.) Kuntze	tea

Distribution

Adoxophyes honmai has been recorded from Japan and Korea (Lee et al 2005). Other tea-feeding *Adoxophyes* species are present in Asia, and it is unknown if these are conspecific with *A. honmai*.

***Adoxophyes negundana* (McDunnough)**

Nomenclature

Adoxophyes negundana (McDunnough) (Tortricidae: Tortricinae: Archipini)

Adult Recognition

Adults are pale yellow with light-brown markings. Hindwings are white. Males have a forewing costal fold. FWL: 7.5-9.5 mm.

Adoxophyes negundana and *A. furcatana* are the only two Nearctic representatives of the genus *Adoxophyes*. These two species are difficult to separate using forewing pattern. In general, the median fascia is broader in *A. negundana*, although this character does not appear to be consistent across all individuals. Freeman (1958) states that male genitalia of *A. negundana* have fewer cornuti (only four) and more sharply elbowed gnathos arms than those of *A. furcatana*.

Both species of Nearctic *Adoxophyes* are similar to *A. orana* and may be confused with this Palearctic species if it were discovered in North America. It is not known if Nearctic *Adoxophyes* are attracted to *A. orana* pheromone.

Larval Morphology

Larval morphology is undocumented for this species.

Biology

Adults are present in June through early September. Larvae feed in the rolled leaves of boxelder (*Acer negundo*) (Table 21) (Freeman 1958).

Table 21: *Adoxophyes negundana* host plants

Family	Genus/species	Common name
Aceraceae	*Acer negundo* L.	boxelder

Distribution

Adoxophyes negundana is found from Ontario to Manitoba, south to Florida and west to Utah (Freeman 1958, McDunnough 1923).

Adoxophyes orana (Fischer von Roslerstamm)

Nomenclature

Adoxophyes orana (Fischer von Roslerstamm) (Tortricidae: Tortricinae: Archipini)

Common names: summer fruit tortrix, smaller tea tortrix, apple peel tortrix

Synonyms: *minor* (*Archips*), *reticulana* (*Tortrix*), *sutschana* (*Capua*), *tripsiana* (*Tortrix*)

Subspecies: *A. orana beijingensis* (China), *A. orana fasciata* (Japan), *A. orana orana* (Europe, China, Korea)

Adult Recognition

Forewings are pale yellowish brown with brown to dark-brown fasciate markings. Males are smaller with brighter markings and a forewing costal fold; females are larger, darker, and have duller markings. Male genitalia are distinguished by reduced socii, a well developed gnathos, a rounded valva, and transtilla with a spiny basal lobe. Female genitalia have a long ductus bursae and a hooklike signum in the rounded corpus bursae. FWL: 7.0-8.5 mm (male); 8.0-11.0 mm (female).

Adults may appear similar to other *Adoxophyes* and species in other genera such as *Choristoneura* and *Clepsis*. *Adoxophyes* in Asia include: *A. dubia* (Japan), *A. honmai* (Japan and Korea), *A. orana beijingensis* (China), *A. orana fasciata* (Japan), *A. orana orana* (China, Korea, and Europe), and *A. privatana* (China). All are difficult to separate using morphology and molecular diagnostics may be required for a positive identification (Lee et al. 2005).

Larval Morphology

Last instar larvae vary in body color. Head and prothoracic shield are black in early instars and become brown to yellowish brown in later instars. Legs are unmarked (Sakamaki and Hayakawa 2004).

Larvae appear similar to those of many other tortricids, including *Choristoneura rosaceana*, *Epiphyas postvittana*, and many Archipini. Molecular diagnostics may be required to positively identify *A. orana* larvae. Sakamaki and Hayakawa (2004) attempted to separate the immature stages of the three Japanese *Adoxophyes* using morphology but the differences they found are too minor to be consistently diagnostic.

Biology

The following life history information is summarized from Beeke and De Jong (1991), Bradley et al. (1973), and Davis et al. (2005). *Adoxophyes orana* completes 2-3 generations per year. Adults are present in late May and June for the first generation, in July through September for the second generation, and in October for the third generation. Flight times vary with latitude and climatic conditions.

Females lay eggs in masses of 4-150 individual eggs. Eggs are laid primarily on leaves, but females may utilize fruits and tree trunks in outbreak conditions. First instar larvae hatch in 8-20 days and feed under a silk web on the underside of a leaf. Later instars feed inside rolled leaves or web leaves to fruit and feed on the surface. Larvae complete five instars and pupation occurs in the final larval nest. Second or third instar larvae of the last generation hibernate until spring and complete development by feeding on buds and young leaves. This species is considered a major pest of fruit crops throughout its range. Larvae are highly polyphagous and have been recorded feeding on the leaves and fruits of plants in many different families (Table 22). Most economic loss is caused by damage to apple and pear.

Table 22: *Adoxophyes orana* host plants

Family	**Genus/species**	**Common name**
Anacardiaceae	*Pistacia* L.	pistache
Betulaceae	*Alnus glutinosa* (L.) Gaertn.	European alder
Betulaceae	*Alnus* Mill.	alder
Betulaceae	*Betula* L.	birch
Betulaceae	*Corylus avellana* L.	common filbert
Betulaceae	*Corylus* L.	hazelnut
Cannabaceae	*Humulus* L.	hop
Cannabaceae	*Humulus lupulus* L.	common hop
Caprifoliaceae	*Lonicera* L.	honeysuckle
Caprifoliaceae	*Lonicera periclymenum* L.	European honeysuckle
Ebenaceae	*Diospyros kaki* L. f.	Japanese persimmon
Ericaceae	*Vaccinium uliginosum* L.	bog blueberry
Fabaceae	*Albizia lebbeck* (L.) Benth.	woman's tongue
Fabaceae	*Arachis hypogaea* L.	peanut
Fabaceae	*Glycine max* (L.) Merr.	soybean
Fagaceae	*Castanea* Mill.	chestnut
Fagaceae	*Castanopsis fissa* (Champ.) Rehd. & Wils.	
Fagaceae	*Lithocarpus glaber* (Thunb.) Nakai	Japanese oak
Fagaceae	*Quercus acutissima* Carruth.	sawtooth oak
Fagaceae	*Quercus* L.	oak
Fagaceae	*Quercus mongolica* Fisch. ex Ledeb.	mongolian oak
Fagaceae	*Quercus robur* L.	English oak
Grossulariaceae	*Ribes* L.	currant
Malvaceae	*Gossypium hirsutum* L. var. *hirsutum*	upland cotton
Moraceae	*Morus alba* L.	white mulberry
Pinaceae	*Larix* Mill.	larch
Rosaceae	*Eriobotrya japonica* (Thunb.) Lindl.	loquat
Rosaceae	*Malus domestica* Borkh.	apple
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Malus sylvestris* (L.) Mill.	European crab apple
Rosaceae	*Prunus avium* (L.) L.	sweet cherry
Rosaceae	*Prunus cerasus* L.	sour cherry
Rosaceae	*Prunus* L.	
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Prunus salicina* Lindl.	Japanese plum
Rosaceae	*Pyrus* L.	pear
Rosaceae	*Pyrus pyrifolia* (Burm. f.) Nakai	Chinese pear
Rosaceae	*Pyrus ussuriensis* Maxim.	
Rosaceae	*Rosa* L.	rose
Rosaceae	*Rubus idaeus* L.	American red raspberry

Table 22: *Adoxophyes orana* host plants

Family	**Genus/species**	**Common name**
Rosaceae	*Rubus* L.	blackberry
Rutaceae	*Citrus* L.	citrus
Salicaceae	*Populus* L.	cottonwood
Salicaceae	*Salix cinerea* L.	large gray willow
Salicaceae	*Salix* L.	willow
Sapindaceae	*Litchi chinensis* Sonn. [excluded]	lychee
Solanaceae	*Solanum dulcamara* L.	climbing nightshade
Theaceae	*Camellia sinensis* (L.) Kuntze	tea

Although tea (*Camellia sinensis*) is listed as a host for *A. orana*, it is questionable if this record refers to the same species of *Adoxophyes*. Zhou et al. (1997) determined that the fruit-feeding populations of *A. orana* in northern China were a different subspecies (*A. orana beijingensis*) than the cotton- and tea-feeding populations in southern China (*A. orana orana*). In Japan, *A. orana* has long been divided into an "apple form" and "tea form," referring to populations that utilize different hosts. Yasuda (1998) described the "tea form" of *Adoxophyes* in Japan as a new species, *A. honmai*, and treated the "apple form" of *A. orana* in Japan as a new subspecies (*A. orana fasciata*). Tea-feeding *Adoxophyes* in Korea were determined to also be *A. honmai* using molecular methods (Lee et al. 2005). It is unknown if the Chinese tea-feeding *A. orana orana* is actually a separate species or if it is conspecific with *A. honmai*.

Distribution

A European native, this species is widely distributed throughout Europe and several countries in Asia (China, Japan, and Korea) (Bradley et al. 1973, Lee et al. 2005).

Archips **Hübner**

Overview

The genus *Archips* contains more than 100 described species that are distributed throughout the Holarctic and Oriental regions (Baixeras et al. 2008). The forewing pattern of many species is similar, with a mark in the subbasal area, a well-defined median fascia, and a costal spot. Male genitalia are characterized by a fingerlike or spatulate uncus, reduced socii, well-developed gnathos, and rounded valvae. Female genitalia are characterized by a funnellike sterigma, single signum in the corpus bursae, and many species have a cestum in the ductus bursae (Horak and Brown 1991).

This group contains a number of important pest species, such as *A. podana* and *A. xylosteana*. Larvae feed primarily on deciduous trees and shrubs, including apple, apricot, plum, pear, peach, and many others. However, most species are external leaf feeders, and fruit damage is caused by incidental feeding or when leaves are webbed to fruit.

It is difficult to distinguish between different species of *Archips* (or many Archipini) using larval morphology. As such, most *Archips* species intercepted at U.S. ports of entry are not identified beyond the subfamily level. Ten species are treated here. Seven are common non-targets that may be encountered during surveys for *A. podana*, *A. xylosteana*, or other Tortricinae.

***Archips argyrospila* (Walker)**

Nomenclature

Archips argyrospila (Walker) (Tortricidae: Tortricinae: Archipini)

Common names: fruit-tree leaf roller, apple leaf roller

Synonyms: *furvana* (*Tortrix*), *vsignatana* (*Tortrix*)

Subspecies: *A. argyrospila columbiana* (British Columbia), *A. argyrospila vividana* (Colorado)

Adult Recognition

Forewing color is a variable combination of reddish brown, dark brown, and tan. The majority of individuals have two contrasting triangular to semi-rectangular pale-tan patches on the costa. Females are generally lighter in color than males. Males have a forewing costal fold. FWL: 6.0-10.2 mm (male); 8.5-11.7 mm (female).

The names associated with *A. argyrospila* are considered a species complex by some authors (Chapman and Lienk 1971, Freeman 1958). This group includes *A. eleagnanus*, *A. mortuana*, *A. myricana*, and two subspecies (*A. a. columbiana* and *A. a. vividana*). According to Freeman (1958), genitalic characters for the group include: an aedeagus with hooked apex and two long cornuti in the male and a bulbous sterigma and antrum in the female.

Larval Morphology

Last instar larvae are 15-24 mm in length with a translucent green or gray abdomen. The head is reddish brown to dark brown and may be mottled in some individuals; head markings are too variable to be diagnostic. The prothoracic shield is amber with brown lateral shading. Prothoracic legs are brown or black while the other thoracic legs are pale and unmarked (Chapman and Lienk 1971). Late instar *A. argyrospila* larvae may be confused with larvae of *Choristoneura rosaceana* and other *Archips* species.

Biology

The following life history information is summarized from Chapman and Lienk (1971), Freeman (1958), and Powell (1964). *Archips argyrospila* completes a single generation per year. Adults are present from mid-May through July.

Eggs are laid in masses on the twigs of the host and covered by the female with a substance that hardens to create a smooth, hard surface. Eggs are laid in June and July and do not hatch until the following year. First instar larvae hatch in late Februrary to mid-May and bore into buds. Later instars roll or tie leaves together or to fruit and partially emerge from the shelter to feed. Larvae may feed on leaves, flowers, buds, or fruits of the host. Pupation occurs within the larval shelter and adults eclose in 10-12 days. The adult flight period lasts approximately 3 weeks.

During the first half of the 20th century, outbreaks of *A. argyrospila* would completely defoliate large areas of vegetation. The species was brought under control with the introduction of pesticides in the mid-1950's. *Archips argyrospila* has been recorded from a long list of plants, many of which are not primary hosts (Table 23). Under outbreak conditions the larvae feed on any plant near the primary host.

Table 23: *Archips argyrospila* host plants

Family	**Genus/species**	**Common name**
Aceraceae	*Acer* L.	maple
Aceraceae	*Acer negundo* L.	boxelder
Aceraceae	*Acer rubrum* L.	red maple
Aceraceae	*Acer spicatum* Lam.	mountain maple
Anacardiaceae	*Rhus* L.	sumac
Anacardiaceae	*Toxicodendron pubescens* Mill.	Atlantic poison oak
Betulaceae	*Betula papyrifera* Marshall	paper birch
Betulaceae	*Carpinus* L.	hornbeam
Caprifoliaceae	*Viburnum* L.	viburnum
Cornaceae	*Cornus glabrata* Benth.	brown dogwood
Cornaceae	*Cornus* L.	dogwood
Ericaceae	*Arbutus* L.	madrone
Ericaceae	*Arctostaphylos* Adans.	manzanita
Ericaceae	*Ledum groenlandicum* Oeder	bog Labrador tea
Ericaceae	*Rhododendron* L.	rhododendron
Ericaceae	*Vaccinium* L.	blueberry
Fabaceae	*Amorpha fruticosa* L.	desert false indigo
Fabaceae	*Medicago sativa* L.	alfalfa
Fabaceae	*Melilotus* Mill.	sweetclover
Fabaceae	*Robinia pseudoacacia* L.	black locust
Fabaceae	*Vicia* L.	vetch
Fagaceae	*Quercus agrifolia* Nee	California live oak
Fagaceae	*Quercus alba* L.	white oak
Fagaceae	*Quercus coccinea* Munchh.	scarlet oak
Fagaceae	*Quercus douglasii* Hook. & Arn.	blue oak
Fagaceae	*Quercus dumosa* Nutt.	coastal sage scrub oak
Fagaceae	*Quercus garryana* Douglas ex Hook.	Oregon white oak
Fagaceae	*Quercus kelloggii* Newb.	California black oak
Fagaceae	*Quercus lobata* Nee	valley oak
Fagaceae	*Quercus rubra* L.	red oak
Fagaceae	*Quercus wislizeni* A. DC.	interior live oak
Fagaceae	*Quercus* L.	oak
Grossulariaceae	*Ribes nigrum* L.	European black currant
Grossulariaceae	*Ribes uva-crispa* L.	European gooseberry
Grossulariaceae	*Ribes* L.	currant
Hippocastanaceae	*Aesculus californica* (Spach) Nutt.	California buckeye

Table 23: *Archips argyrospila* host plants

Family	**Genus/species**	**Common name**
Hippocastanaceae	*Aesculus hippocastanum* L.	horse chestnut
Hydrophyllaceae	*Eriodictyon* Benth.	yerba santa
Hydrophyllaceae	*Eriodictyon californicum* (Hook. & Arn.) Torr.	California yerba santa
Juglandaceae	*Carya* Nutt.	hickory
Juglandaceae	*Juglans* L.	walnut
Liliaceae	*Allium cepa* L.	garden onion
Myricaceae	*Myrica* L.	sweetgale
Myrtaceae	*Comptonia peregrina* (L.) J. M. Coult.	sweet fern
Oleaceae	*Fraxinus* L.	ash
Oleaceae	*Fraxinus latifolia* Benth.	Oregon ash
Pinaceae	*Abies concolor* (Gord. & Glend.) Lindl. ex Hildebr.	white fir
Pinaceae	*Pseudotsuga macrocarpa* (Vasey) Mayr	bigcone Douglas-fir
Pinaceae	*Pseudotsuga menziesii* (Mirb.) Franco	Douglas-fir
Platanaceae	*Platanus* L.	sycamore
Rhamnaceae	*Ceanothus cuneatus* (Hook.) Nutt.	buckbrush
Rhamnaceae	*Ceanothus incanus* Torr. & A. Gray	coast whitethorn
Rosaceae	*Cercocarpus* Kunth	mountain mahogany
Rosaceae	*Cercocarpus montanus* Raf.	alderleaf mountain mahogany
Rosaceae	*Cercocarpus montanus* Raf. var. *glaber* (S. Watson) F. L. Martin	birchleaf mountain mahogany
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Malus domestica* Borkh.	apple
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus armeniaca* L.	apricot
Rosaceae	*Prunus avium* (L.) L.	sweet cherry
Rosaceae	*Prunus ilicifolia* (Nutt. ex Hook. & Arn.) D. Dietr.	hollyleaf cherry
Rosaceae	*Prunus pensylvanica* L. f.	pin cherry
Rosaceae	*Prunus virginiana* L.	chokecherry
Rosaceae	*Prunus* L.	
Rosaceae	*Pyrus communis* L.	common pear
Rosaceae	*Pyrus* L.	pear
Rosaceae	*Rosa californica* Cham. & Schltdl.	California wildrose
Rosaceae	*Rosa* L.	rose
Rosaceae	*Rubus deliciosus* Torr.	delicious raspberry
Rosaceae	*Rubus* L.	blackberry
Rutaceae	*Citrus* L.	citrus
Rutaceae	*Citrus* X *sinensis* (L.) Osbeck (pro sp.) [*maxima* X *reticulata*]	sweet orange
Salicaceae	*Populus balsamifera* L.	balsam poplar
Salicaceae	*Populus grandidentata* Michx.	bigtooth aspen

Table 23: *Archips argyrospila* host plants

Family	**Genus/species**	**Common name**
Salicaceae	*Populus tremuloides* Michx.	quaking aspen
Salicaceae	*Populus* L.	cottonwood
Salicaceae	*Salix lasiolepis* Benth.	arroyo willow
Salicaceae	*Salix sessilifolia* Nutt.	northwest sandbar willow
Salicaceae	*Salix* L.	willow
Tiliaceae	*Tilia americana* L.	American basswood
Ulmaceae	*Ulmus americana* L.	American elm
Ulmaceae	*Ulmus* L.	elm
Vitaceae	*Vitis* L.	grape

Distribution

Archips argyrospila is native to North America and is found throughout the continental United States and southern Canada.

Archips cerasivorana **(Fitch)**

Nomenclature

Archips cerasivorana (Fitch) (Tortricidae: Tortricinae: Archipini)

Common names: ugly-nest caterpillar

Adult Recognition

Forewing color varies from bright orange to yellow, often with faint purplish markings. The hindwing is yellow or yellowish brown. Males have a forewing costal fold. FWL: 7.5-9.5 mm (male); 9.0-12.0 mm (female).

Archips rileyana is very similar to *A. cerasivorana*, both in appearance and larval habits. Freeman (1958) considered *A. rileyana* a subspecies of *A. cerasivorana*, but Obraztsov (1959) demonstrated that both adults and larvae of the two species can be separated. *Archips cerasivorana* is characterized by an uncus with parallel sides in the male, a large blunt signum in the female, and smaller, sometimes body-colored pinacula on the larval abdomen. *Archips* *rileyana* is characterized by a spatulate uncus in the male, a moderate pointed signum in the female, and very large, conspicuous, black pinacula on the larval abdomen.

Larval Morphology

Last instar larvae are 19-26 mm in length with a yellow to dark yellowish-green abdomen. The head, prothoracic shield, thoracic legs, and anal shield are dark brown to black. An anal comb is absent.

Larvae of *Archips rileyana* are very similar to those of *A. cerasivorana*. Larvae of *A. rileyana* have very large, conspicuous, black pinacula, whereas those of *A. cerasivorana* are smaller, and range from body-colored to black (Obraztsov 1959).

Biology

The following life history information is summarized from Chapman and Lienk (1971), Obraztsov (1959), and Powell and Opler (2009). *Archips cerasivorana* completes one generation per year. Adults are present in July and August.

Females lay eggs in masses of 25-200 eggs at the base of shoots, often near the ground. Eggs overwinter and first instar larvae hatch in May. Unlike many tortricids, the larvae are social, and feeding occurs in silken nests on terminals of the host plants. A typical nest may contain 30-200 larvae and reach up to 30 inches in diameter. The nest is expanded when the colony needs additional food and feeding always occurs under the protection of the nest. Early stages skeletonize leaves while later stages consume entire leaves. Pupation occurs in chambers constructed in the nest from frass and silk.

Archips cerasivorana is primarily associated with cherry and its most common host is *Prunus virginiana* (chokecherry). During outbreak conditions the larvae may utilize other hosts (Table 24).

Table 24: Archips cerasivorana host plants

Family	**Genus/species**	**Common name**
Aceraceae	*Acer saccharinum* L.	silver maple
Betulaceae	*Alnus incana* (L.) Moench	gray alder
Betulaceae	*Betula papyrifera* Marshall	paper birch
Betulaceae	*Corylus* L.	hazelnut
Fagaceae	*Quercus macrocarpa* Michx.	bur oak
Fagaceae	*Quercus palustris* Munchh.	pin oak
Oleaceae	*Fraxinus* L.	ash
Pinaceae	*Pinus sylvestris* L.	Scots pine
Rosaceae	*Amelanchier* Medik.	serviceberry
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Prunus avium* (L.) L.	sweet cherry
Rosaceae	*Prunus emarginata* (Douglas ex Hook.) D. Dietr.	bitter cherry
Rosaceae	*Prunus ilicifolia* (Nutt. ex Hook. & Arn.) D. Dietr.	hollyleaf cherry
Rosaceae	*Prunus* L.	
Rosaceae	*Prunus pensylvanica* L. f.	pin cherry
Rosaceae	*Prunus pumila* L.	sandcherry
Rosaceae	*Prunus serotina* Ehrh.	black cherry
Rosaceae	*Prunus virginiana* L.	chokecherry
Rosaceae	*Prunus virginiana* L. var. *demissa* (Nutt.) Torr.	western chokecherry
Rosaceae	*Rosa* L.	rose
Salicaceae	*Populus balsamifera* L.	balsam poplar
Salicaceae	*Populus* L.	cottonwood
Salicaceae	*Populus tremuloides* Michx.	quaking aspen
Salicaceae	*Salix* L.	willow
Tiliaceae	*Tilia americana* L.	American basswood

Distribution

Archips cerasivorana occurs across southern Canada and the northern half of the United States, although there are records from some southern States (Chapman and Lienk 1971, Powell and Opler 2009).

***Archips crataegana* (Hübner)**

Nomenclature

Archips crataegana (Hübner) (Tortricidae: Tortricinae: Archipini)

Common names: brown oak tortrix

Synonyms: *confluens* (ab.), *roborana* (*Tortrix*), *rubromaculata* (var.)

Subspecies: *A. crataegana endoi* (Japan)

Adult Recognition

The male forewing is light brown with dark-brown markings. The female forewing is generally darker with more obscured markings and a pronounced apex. The median fascia does not extend to the costa in the male, while it is continuous from the dorsal margin to the costa in the female. The hindwing of both sexes is grayish brown with yellow apical shading present in some females. Males have a forewing costal fold. FWL: 9.0-10.5 mm (male); 11.0-13.0 mm (female).

Yasuda (1975) described *A. crataegana endoi* as a subspecies from Japan. He states that it can be separated from the nominate subspecies by its larger size, longer costal fold in the male, and shape of the male sacculus.

Archips crataegana is most similar to *A. xylosteana*. Males of the two species can be separated by the median fasica, which is continuous to from dorsum to costa in *A. xylosteana*, and does not reach the costa in *A. crataegana* (Bradley et al. 1973). Other *Archips* species, including *A. grisea* and forms of *A. podana* and *A. rosana*, can appear similar to *A. crataegana*. A genitalic dissection can be used to confirm species identity.

Larval Morphology

Larvae are approximately 23 mm in length. The abdomen is dark green to black with black pinacula and long white setae. The head, prothoracic shield, thoracic legs, and anal shield are black. An anal comb is present with 6-8 teeth (Bradley et al. 1973).

Biology

Archips crataegana completes one generation per year. Adults are present June through August. Females deposit eggs on bark in masses of approximately 30 individual eggs. Egg masses are usually deposited high on the tree and appear similar to bird droppings. Eggs overwinter and first instar larvae hatch the following spring. Early instars feed on the underside of the leaf while later instars roll leaves. Pupation occurs in the final larval feeding site (Bradley et al. 1973).

Larvae of *A. crataegana* have been reported feeding on a variety of tree species (Table 25). Host records from East Asia may refer to *A. crataegana endoi* (Yasuda 1975), or possibly another species (Razowski 2002).

Distribution

Archips crataegana is distributed across the western Palearctic. Yasuda (1975) described *A. crataegana endoi* from Japan, although Razowski (2002) states that records of *A. crataegana* from East Asia are of a different species.

Table 25: *Archips crataegana* host plants

Family	**Genus/species**	**Common name**
Betulaceae	*Alnus japonica* (Thunb.) Steud.	Japanese alder
Betulaceae	*Betula platyphylla* Sukaczev	Asian white birch
Fagaceae	*Quercus dentata* Thunb.	Daimyo oak
Fagaceae	*Quercus robur* L.	English oak
Fagaceae	*Quercus* L.	oak
Moraceae	*Morus* L.	mulberry
Oleaceae	*Fraxinus excelsior* L.	European ash
Rosaceae	*Crataegus pinnatifida* Bunge	Chinese haw
Rosaceae	*Malus baccata* (L.) Borkh.	Siberian crab apple
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Prunus sargentii* Rehder	
Rosaceae	*Prunus tomentosa* Thunb.	Nanking cherry
Rosaceae	*Prunus* X *yedoensis* Matsum. (pro sp.) [*subhirtella* X *speciosa*]	
Rosaceae	*Prunus* L.	
Rosaceae	*Pyrus pyrifolia* (Burm. f.) Nakai	Chinese pear

Table 25: *Archips crataegana* host plants

Family	**Genus/species**	**Common name**
Rosaceae	*Pyrus* L.	pear
Rutaceae	*Citrus* L.	citrus
Salicaceae	*Populus maximoniczii* A. Henry	Japanese poplar
Salicaceae	*Populus* L.	cottonwood
Salicaceae	*Salix* L.	willow
Tiliaceae	*Tilia* L.	basswood
Ulmaceae	*Ulmus* L.	elm

Archips fuscocupreanus **Walsingham**

Nomenclature

Archips fuscocupreanus Walsingham (Tortricidae: Tortricinae: Archipini)

Common names: apple tortrix

Synonyms: *ishidaii* (*Loxotaenia*), *punicae* (*Cacoecia*), *rosaceana* (*Archips*)

Adult Recognition

Forewing color is dull brown with dark-brown to reddish-brown markings. Pattern expression can vary between individuals, but the median fascia is complete from costa to dorsum. Hindwings are brownish gray. Males have a forewing costal fold. FWL: 7.0-9.5 mm (male); 8.0-12.5 mm (female).

Some individuals resemble *A. xylosteana*, but the two species can be separated by the broader costal fold and continuous median fasica (from costa to dorsum) in *A. fuscocupreanus*. Other *Archips* species, including *A. crataegana*, *A. grisea*, and forms of *A. podana* and *A. rosana*, can appear similar to *A. fuscocupreanus*. A genitalic dissection can be used to confirm identity.

Larval Morphology

Late instar larvae are approximately 22 mm in length. The abdomen is pale green to grayish green with dark dorsal and lateral lines and conspicuous pale pinacula. The head is orange or brownish orange. The prothoracic shield is brownish orange with black posterolateral margins. The thoracic legs and thoracic pinacula are black (Yasuda 1975).

Biology

Archips fuscocupreanus completes one generation per year, with adults present in mid-June through mid-July in the northeastern U.S.

Females lay black egg masses on the trunks and limbs of trees. The eggs overwinter, and first instar larvae hatch the following spring. Early instars feed on young leaves. Later instars construct a shelter by webbing together leaves and consume leaves, flowers, and occasionally fruit. Pupation occurs in webbed leaves (Maier 2003).

Larvae of *A. fuscocupreanus* are highly polyphagous and have been recorded feeding on plants in more than 23 families. Many larval hosts are in the Rosaceae, and *A. fuscocupreanus* is an important pest of apple in East Asia (Yasuda 1975). A partial host list is presented in Table 26; Maier (2003) provides a list of other minor hosts found in the northeastern U.S.

Table 26: *Archips fuscocupreanus* host plants

Family	**Genus/species**	**Common name**
Aceraceae	*Acer* L.	maple
Aquifoliaceae	*Ilex verticillata* (L.) Gray	common winterberry
Asteraceae	*Erigeron annuus* (L.) Pers.	eastern daisy
Betulaceae	*Alnus* Mill.	alder
Betulaceae	*Betula platyphylla* Sukaczev	Asian white birch
Betulaceae	*Corylus* L.	hazelnut
Celastraceae	*Celastrus orbiculatus* Thunb.	Oriental bittersweet
Cornaceae	*Cornus* L.	dogwood
Ebenaceae	*Diospyros kaki* L. f.	Japanese persimmon
Elaeagnaceae	*Elaeagnus umbellata* Thunb.	autumn olive
Ericaceae	*Rhododendron* L.	rhododendron
Euphorbiaceae	*Ricinus communis* L.	castorbean
Fabaceae	*Glycine max* (L.) Merr.	soybean
Fabaceae	*Phaseolus* L.	bean
Fagaceae	*Castanea crenata* Siebold & Zucc.	Japanese chestnut

Table 26: *Archips fuscocupreanus* host plants

Family	**Genus/species**	**Common name**
Fagaceae	*Castanea* Mill.	chestnut
Fagaceae	*Quercus acutissima* Carruth.	sawtooth oak
Fagaceae	*Quercus glauca* Thunb.	ring-cup oak
Fagaceae	*Quercus mongolica* Fisch. ex Ledeb.	mongolian oak
Fagaceae	*Quercus serrata* Thunb.	bao li
Fagaceae	*Quercus variabilis* Blume	Chinese cork oak
Hydrangeaceae	*Deutzia* Thunb.	pride-of-Rochester
Juglandaceae	*Juglans* L.	walnut
Moraceae	*Morus alba* L.	white mulberry
Moraceae	*Morus bombycis* Koidz.	
Pinaceae	*Larix* Mill.	larch
Pinaceae	*Picea* A. Dietr.	spruce
Rhamnaceae	*Rhamnus* L.	buckthorn
Rosaceae	*Amelanchier canadensis* (L.) Medik.	Canadian serviceberry
Rosaceae	*Cydonia oblonga* Mill.	quince
Rosaceae	*Fragaria* L.	strawberry
Rosaceae	*Malus baccata* (L.) Borkh.	Siberian crab apple
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus armeniaca* L.	apricot
Rosaceae	*Prunus avium* (L.) L.	sweet cherry
Rosaceae	*Prunus laurocerasus* L.	cherry laurel
Rosaceae	*Prunus mume* Siebold & Zucc.	Japanese apricot
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Prunus salicina* Lindl.	Japanese plum
Rosaceae	*Prunus sargentii* Rehder	
Rosaceae	*Prunus serotina* Ehrh.	black cherry
Rosaceae	*Prunus* X *yedoensis* Matsum. (pro sp.) [*subhirtella* X *speciosa*]	
Rosaceae	*Pyrus pyrifolia* (Burm. f.) Nakai	Chinese pear
Rosaceae	*Pyrus ussuriensis* Maxim.	
Rosaceae	*Pyrus* L.	pear
Rosaceae	*Rosa multiflora* Thunb.	multiflora rose
Rosaceae	*Rosa rugosa* Thunb.	rugosa rose
Rosaceae	*Rubus* L.	blackberry
Rosaceae	*Sorbus* L.	mountain ash
Salicaceae	*Salix* L.	willow
Ulmaceae	*Ulmus* L.	elm

Distribution

A native of East Asia, *Archips fuscocupreanus* has been introduced to the northeastern United States (Connecticut, Massachusetts, New Jersey, New York, and Rhode Island) and Pacific Northwest (Washington) (Maier 2003, Maier and Mastro 1998).

Archips grisea **(Robinson)**

Nomenclature

Archips grisea (Robinson) (Tortricidae: Tortricinae: Archipini)

Common names: black-shield leaf roller

Synonyms: *brauniana* (*Cacoecia*)

Adult Recognition

Forewing color is primarily brownish gray, with dark-brown to black markings that are more prominently expressed in the male. Hindwings are a uniform gray. Males have a forewing costal fold. FWL: 7.5-9.0 mm (male); 8.0-11.0 mm (female).

Adult *A. grisea* are unlikely to be confused with other native Nearctic *Archips*. General forewing pattern is similar to some forms of *A. podana*, *A. crataegana*, and *A. rosana*, although the grayish appearance of the forewing and hindwing should be sufficient to distinguish *A. grisea* from these other species.

Larval Morphology

Larvae are 15-23 mm in length with a green abdomen. The prothoracic shield and head are uniformly black in the last instar. The prothoracic legs are brown to black while the other thoracic legs are pale green and unmarked (Chapman and Lienk 1971).

The life cycle and larval habits of *A. grisea* are similar to those of *A. argyrospila*, *A. mortuana*, and *Choristoneura rosaceana*. Larvae of *A. grisea* are distinguished from these other species by a uniformly black prothoracic shield. *Archips cerasivorana* larvae also have a uniformly black prothoracic shield, but the larvae live and feed in communal nests.

Biology

Archips grisea completes one generation per year. Adults are present in June and July. This species has the same basic life history information as *Archips argyrospila* and can often be found in association with *A. argyrospila* infestations (Chapman and Lienk 1971).

Females lay eggs in masses of approximately 50-60 individual eggs on small diameter branches of the host. Eggs overwinter and first instar larvae hatch the following spring. Larvae feed on leaves and young fruit and pupation occurs within the final larval feeding site. Chapman and Lienk (1971) state that apple and hawthorn are the primary larval hosts in the northeastern U.S. Other host plants are listed in Table 27.

Table 27: *Archips grisea* host plants

Family	**Genus/species**	**Common name**
Asteraceae	*Rudbeckia* L.	coneflower
Fagaceae	*Quercus* L.	oak
Juglandaceae	*Carya* Nutt.	hickory
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus virginiana* L.	chokecherry
Rosaceae	*Pyrus* L.	pear

Distribution

Archips grisea is distributed in the eastern United States, from Maine south to Texas. There is a record from Utah and Chapman and Lienk (1971) speculate this might have been the result of an introduction of infested plants from the East.

***Archips mortuana* (Kearfott)**

Nomenclature

Archips mortuana (Kearfott) (Tortricidae: Tortricinae: Archipini)

Common names: dusky-back leaf roller

Adult Recognition

The male forewing pattern is similar to that of *Archips argyrospila*, with at least one, and usually two, semi-rectangular pale tan patches on the costa. These costal patches are greatly reduced in the female, with the remaining pattern resembling the male or reduced to a patternless rusty gray. Males have a forewing costal fold. FWL: 7.0-9.0 mm.

Archips mortuana is part of the *A. argyrospila* species complex, and it was once hypothesized to simply be a dark form of *A. argyrospila* (Powell 1964). Chapman and Lienk (1971) demonstrated that the sex pheromone for *A. mortuana* is different from *A. argyrospila*, providing evidence that the two are indeed separate species.

Larval Morphology

Larvae are 14-22 mm in length. Early to mid-instars are dark green dorsally and ligher green laterally; this color difference is reduced in the last instar. Pinacula are pale and contrast with the dark green dorsal coloration. The head is yellowish brown and may be unmarked or have dark-brown to black mottling. The prothoracic shield is greenish brown with variable degrees of posterior shading (Chapman and Lienk 1971).

Biology

Archips mortuana completes one generation per year. Adults are present in June and July. This species has the same basic life history information as *A. argyrospila* (Chapman and Lienk 1971).

Females lay eggs in masses of approximately 60-80 individual eggs. Eggs overwinter and first instar larvae hatch the following spring. Larvae feed primarily on leaves and pupation occurs within the final larval feeding site. Chapman and Lienk (1971) consider apple and hawthorn to be the primary larval hosts in the northeastern U.S. A list of documented host plants is provided in Table 28.

Table 28: *Archips mortuana* host plants

Family	**Genus/species**	**Common name**
Apiaceae	*Cicuta* L.	water hemlock
Betulaceae	*Alnus* Mill.	alder
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus virginiana* L.	chokecherry
Rosaceae	*Spiraea* L.	meadowsweet
Salicaceae	*Populus balsamifera* L.	balsam poplar
Salicaceae	*Populus tremuloides* Michx.	quaking aspen
Salicaceae	*Salix bebbiana* Sarg.	Bebb willow
Salicaceae	*Salix* L.	willow

Distribution

Archips mortuana is found in the eastern half North America. There are also individuals recorded from Utah and California, but Chapman and Lienk (1971) doubt the accuracy of these records.

Archips podana **(Scopoli)**

Nomenclature

Archips podana (Scopoli) (Tortricidae: Tortricinae: Archipini)

Common names: fruit tree tortrix

Synonyms: *ameriana* (*Cacoecia*), *congenerana* (*Tortrix*), *fulvana* (*Tortrix*), *meridana* (*Archips*), *pyrastrana* (*Tortrix*), *sauberiana* (var.), *vulpeculana* (*Tortrix*)

Adult Recognition

Archips podana is sexually dimorphic. The male forewing is pale purplish brown with a dark-brown basal patch and well-defined median fascia. The female forewing is pale brown to purplish brown with darker reticulate markings. The hindwing in both sexes is gray basally and yellowish orange apically; the yellowish-orange coloration is usually more pronounced in the female. Dark forms of both sexes exist, where the entire forewing is overlaid with purplish-gray scaling. Males have a forewing costal fold. FWL: 10.0-12.0 mm.

Male genitalia are characterized by a well-developed uncus, reduced socii, rounded valva, and a well defined sacculus ending in a point. Female genitalia are characterized by a long ductus bursae with cestum and a single signum in the rounded corpus bursae.

Archips podana males can appear similar to males of several other *Archips* species, including: *A. betulana*, *A. crataegana*, *A. grisea*, *A. magnoliana*, *A. oporana*, and *A. xylosteana*. The yellowish-orange coloration on the apical half of the hindwing serves to separate *A. podana* from these other species.

Other similar species of *Archips* with yellowish-orange apical shading on the hindwing include *A. oporana* and *A. rosana*, but the amount of shading is much greater in most *A. podana* individuals. Female *A. podana* may be confused with female *Choristoneura rosaceana*, which has similar markings on both the forewing and hindwing. A genitalic dissection will easily separate these two species.

Larval Morphology

Last instar larvae are approximately 15-20 mm in length. The abdomen is pale green to pale gray with pale conspicuous pinacula. The head is reddish brown. The prothoracic shield is reddish brown anteriorly and shaded with black posteriorly; a thin white line on the anterior margin of the shield separates it from the head. Thoracic legs are brown or black (Bradley et al. 1973, LaGasa et al. 2003). Larvae of *A. podana* are similar to those of *Adoxophyes orana*, *A. rosana*, and *Choristoneura rosaceana*.

Biology

The following life history information is summarized from Bradley et al. (1973) and LaGasa et al. (2003). *Archips podana* completes a single generation per year over most of its range. In warmer climates a partial second generation may develop. Adults are present from the end of June through early August.

Females lay eggs in masses of 50-100 individual eggs on the surfaces of leaves. Eggs are covered by a waxy secretion that both protects and camouflages the egg mass. First instar larvae hatch in 17-23 days and begin feeding on leaves. Second and third instars feed on the surface of fruit, often creating a shelter by webbing leaves to fruit. Larvae overwinter in the third instar and continue development the following spring. Fourth instars feed on developing buds and subsequent instars feed on foliage in shelters constructed of webbed leaves. Pupation occurs in the final larval shelter.

Archips podana is considered a major pest of apple and pear in parts of Europe. Larvae are moderately polyphagous and feed on a variety of deciduous trees and shrubs (Table 29).

Distribution

Archips podana is distributed throughout Europe. Records from East Asia are of another species (Razowski 2002). It has been introduced into the Pacific Northwest where it is present in Washington and British Columbia (LaGasa et al. 2003).

Table 29: *Archips podana* host plants

Family	**Genus/species**	**Common name**
Betulaceae	*Alnus glutinosa* (L.) Gaertn.	European alder
Betulaceae	*Corylus* L.	hazelnut
Caprifoliaceae	*Lonicera periclymenum* L.	European honeysuckle
Celastraceae	*Euonymus japonicus* Thunb.	Japanese spindletree
Ericaceae	*Vaccinium* L.	blueberry
Fabaceae	*Trifolium* L.	clover
Fagaceae	*Fagus* L.	beech
Fagaceae	*Quercus robur* L.	English oak
Fagaceae	*Quercus* L.	oak
Grossulariaceae	*Ribes* L.	currant
Pinaceae	*Picea* A. Dietr.	spruce
Rosaceae	*Cydonia oblonga* Mill.	quince
Rosaceae	*Malus domestica* Borkh.	apple
Rosaceae	*Malus sylvestris* (L.) Mill.	European crab apple
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Padus avium* Mill.	
Rosaceae	*Prunus* L.	cherry

Table 29: *Archips podana* host plants

Family	**Genus/species**	**Common name**
Rosaceae	*Prunus* L.	plum
Rosaceae	*Pyrus* L.	pear
Rosaceae	*Rosa rugosa* Thunb.	rugosa rose
Rosaceae	*Rubus idaeus* L.	American red raspberry
Rosaceae	*Rubus* L.	blackberry
Rosaceae	*Rubus* L.	raspberry

***Archips purpurana* (Clemens)**

Nomenclature

Archips purpurana (Clemens) (Tortricidae: Tortricinae: Archipini)

Synonyms: *guritana* (*Cacoecia*), *gurgitana* (*Tortrix*), *lintneriana* (*Tortrix*)

Adult Recognition

Forewing color varies from brown to dark purplish brown, with brown to dark-brown markings and reticulations. The costal margin of the forewing is sinuate, especially in the female. The male forewing costal fold is expressed as a rolling of the costal margin that does not completely extend to the base of the wing. FWL: 8.5-11.0 mm (male); 10.5-12.5 mm (female). Adults are unlikely to be confused with other species of Nearctic Tortricidae due to the strongly sinuate costal margin of the forewing.

Larval Morphology

Last instar larvae are 20-30 mm in length. The abdomen is pale bluish green. The head is yellowish brown. The prothoracic shield is yellowish brown, with pair of semirectangular black spots on the posterolateral margin. The thoracic legs are pale and unmarked. Late instar larvae can be easily confused with those of *Clepsis melaleucanus*, which are similar in appearance and occur in the same habitat (Chapman and Lienk 1971).

Biology

Archips purpurana completes a single generation over most of its range. Adults present in June and July.

Females lay eggs in masses, presumably on the upper surface of leaves. Early instar larvae feed on foliage, occasionally webbing leaves to fruit. Overwintering occurs as a third instar larva, either in fallen leaves on the ground or possibly in the host tree. In the spring, larvae crawl up into any non-coniferous host to complete development. Pupation occurs in the final larval feeding site (Chapman and Lienk 1971).

Larvae of *A. purpurana* are highly polyphagous and have been recorded feeding on plants in approximately 20 families (Table 30). Although larvae have been recorded from economically important species such as apple, it is likely that *A. purpurana* is a general feeder that utilizes any readily available host (Chapman and Lienk 1971).

Table 30: *Archips purpurana* host plants

Family	**Genus/species**	**Common name**
Anacardiaceae	*Rhus* L.	sumac
Asteraceae	*Erigeron annuus* (L.) Pers.	eastern daisy
Asteraceae	*Solidago* L.	goldenrod
Betulaceae	*Betula papyrifera* Marshall	paper birch
Betulaceae	*Betula populifolia* Marsh.	gray birch
Betulaceae	*Betula* L.	birch
Caprifoliaceae	*Viburnum* L.	viburnum
Cornaceae	*Cornus canadensis* L.	bunchberry dogwood
Crassulaceae	*Sedum* L.	stonecrop
Crassulaceae	*Sempervivum tectorum* L.	common houseleek
Ericaceae	*Vaccinium* L.	blueberry
Fabaceae	*Lupinus* L.	lupine
Fagaceae	*Quercus macrocarpa* Michx.	bur oak
Geraniaceae	*Geranium* L.	geranium
Grossulariaceae	*Ribes* L.	currant
Lauraceae	*Sassafras* Nees & Eberm.	sassafras
Liliaceae	*Maianthemum racemosum* (L.) Link	feathery false lily of the valley
Oleaceae	*Fraxinus* L.	ash
Rosaceae	*Fragaria* L.	strawberry
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus pensylvanica* L. f.	pin cherry
Rosaceae	*Prunus virginiana* L.	chokecherry
Rosaceae	*Prunus* L.	

Table 30: *Archips purpurana* host plants

Family	**Genus/species**	**Common name**
Rosaceae	*Rubus plicatus* Weihe & Nees	shrubby blackberry
Rosaceae	*Rubus* L.	raspberry
Rosaceae	*Spiraea* L.	meadowsweet
Salicaceae	*Populus tremuloides* Michx.	quaking aspen
Salicaceae	*Salix* L.	willow
Solanaceae	*Mandragora* L.	mandrake
Tiliaceae	*Tilia americana* L.	American basswood
Violaceae	*Viola* L.	violet

Distribution

Archips purpurana is distributed throughout eastern North America.

Archips rosana **(Linnaeus)**

Nomenclature

Archips rosana (Linnaeus) (Tortricidae: Tortricinae: Archipini)

Common names: rose tortrix, European leafroller

Synonyms: *acerana* (*Tortrix*), *ameriana* (*Tortrix*), *avellana* (*Tortrix*), *hewittana* (*Cacoecia*), *laevigana* (*Tortrix*), *nebulana* (*Lozotaenia*), *obscura* (ab.), *ochracea* (ab.), *orientana* (var.), *oxyacanthana* (*Tortrix*), *splendana* (var.), *variana* (*Pyralis*)

Adult Recognition

Forewings are light brown to brown with dark-brown markings that vary in expression. Hindwings are grayish brown with yellowish-orange apical shading. Males have a forewing costal fold. FWL: 7.0-9.0 mm (male); 8.5-10.5 mm (female).

Some phenotypes of *Archips rosana* can appear similar to several other *Archips* species, including: *A. betulana*, *A. crataegana*, *A. grisea*, *A. oporana*, *A. podana*, and *A. xylosteana*. Forewing markings in most *A. rosana* individuals are more subdued than in these other species.

Larval Morphology

Last instar larvae are 15-20 mm in length. The abdomen is dull pale green to dark olive green with pale pinacula. The head is reddish brown to dark brown. The greenish-brown prothoracic shield is shaded posteriorly with dark brown to black; the degree of shading varies. Thoracic legs are brown to black (Bradley et al. 1973, Chapman and Lienk 1971). Larvae of *A. rosana* are similar to those of *Adoxophyes orana*, *A. podana*, and *Choristoneura rosaceana*.

Biology

Archips rosana completes one generation per year. Adults are present in late June to August.

The life cycle of *A. rosana* is similar to that of *Archips argyrospila*. Females lay eggs in masses of approximately 60 individual eggs on woody parts of the host plant. Eggs overwinter and larvae hatch the following spring. Larvae feed on leaves, flowers, and small fruits, rolling leaves in later instars. Pupation occurs in the final larval feeding site (Chapman and Lienk 1971).

Archips rosana is a pest of fruit trees and other Rosaceae in Europe and the Pacific Northwest. Larvae are polyphagous, feeding on members of more than 15 plant families (Table 31). Chapman and Lienk (1972) speculate that primary hosts are apple, currant, hawthorn, pear, and privet.

Table 31: *Archips rosana* host plants

Family	**Genus/species**	**Common name**
Aceraceae	*Acer circinatum* Pursh	vine maple
Aceraceae	*Acer macrophyllum* Pursh	bigleaf maple
Aceraceae	*Acer rubrum* L.	red maple
Aceraceae	*Acer* L.	maple
Betulaceae	*Alnus glutinosa* (L.) Gaertn.	European alder
Betulaceae	*Alnus incana* (L.) Moench	gray alder
Betulaceae	*Alnus rhombifolia* Nutt.	white alder
Betulaceae	*Alnus rubra* Bong.	red alder
Betulaceae	*Alnus* Mill.	alder

Table 31: *Archips rosana* host plants

Family	**Genus/species**	**Common name**
Betulaceae	*Corylus avellana* L.	common filbert
Betulaceae	*Corylus cornuta* Marsh. var. californica (A. DC.) Sharp	California hazelnut
Betulaceae	*Corylus* L.	hazelnut
Caprifoliaceae	*Viburnum opulus* L.	European cranberrybush
Caprifoliaceae	*Viburnum* L.	viburnum
Cornaceae	*Cornus nuttallii* Audubon ex Torr. & A. Gray	Pacific dogwood
Cornaceae	*Cornus racemosa* Lam.	gray dogwood
Cornaceae	*Cornus sericea* L.	redosier dogwood
Ericaceae	*Vaccinium* L.	blueberry
Fabaceae	*Caragana arborescens* Lam.	Siberian peashrub
Fagaceae	*Quercus robur* L.	English oak
Fagaceae	*Quercus rubra* L.	red oak
Fagaceae	*Quercus* L.	oak
Grossulariaceae	*Ribes nigrum* L.	European black currant
Grossulariaceae	*Ribes* L.	currant
Juglandaceae	*Carya* Nutt.	hickory
Oleaceae	*Ligustrum vulgare* L.	European privet
Oleaceae	*Ligustrum* L.	privet
Oleaceae	*Syringa* L.	lilac
Pinaceae	[unspecified]	
Rhamnaceae	*Rhamnus* L.	buckthorn
Rosaceae	*Crataegus douglasii* Lindl.	black hawthorn
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Malus domestica* Borkh.	apple
Rosaceae	*Malus sylvestris* (L.) Mill.	European crab apple
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus armeniaca* L.	apricot
Rosaceae	*Prunus avium* (L.) L.	sweet cherry
Rosaceae	*Prunus domestica* L.	European plum
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Prunus virginiana* L.	chokecherry
Rosaceae	*Prunus* L.	
Rosaceae	*Pyrus communis* L.	common pear
Rosaceae	*Pyrus* L.	pear
Rosaceae	*Rosa rugosa* Thunb.	rugosa rose
Rosaceae	*Rosa* L.	rose
Rosaceae	*Rubus idaeus* L.	American red raspberry
Rosaceae	*Rubus parviflorus* Nutt.	thimbleberry
Rosaceae	*Rubus* L.	blackberry
Rutaceae	*Citrus* L.	citrus

Table 31: *Archips rosana* host plants

Family	**Genus/species**	**Common name**
Salicaceae	*Populus* L.	cottonwood
Salicaceae	*Salix* L.	willow
Ulmaceae	*Ulmus* L.	elm
Urticaceae	*Urtica dioica* L.	stinging nettle

Distribution

Archips rosana is native to the Palearctic. It was introduced into North America prior to 1890 where it is currently present in the Northeast and Pacific Northwest (Powell and Opler 2009).

Archips semiferanus **(Walker)**

Nomenclature

Archips semiferanus (Walker) (Tortricidae: Tortricinae: Archipini)

Common names: oak leafroller

Synonyms: *flaccidana* (*Tortrix*)

Adult Recognition

Forewings are pale golden brown with variable reddish-brown markings. A broad, reddish-brown median fascia is continuous from the costa to the dorum in many individuals. In other individuals the fasciae are a subdued pale golden brown. The hindwings are light brown. Males have a forewing costal fold. FWL: 8.5-11.5 mm. The ventral surface of the female abdomen is covered in dark purplish-gray scales that are used to cover the egg masses.

Archips semiferanus adults are most similar to those of *Archips negundana*, although most *A. negundana* individuals are lighter with white hindwings, and females exhibit differences in the scales on the ventral surface of the abdomen. The two species are similar enough to be confused in the early literature; Freeman (1958) described their differences and separated them by primary host: *A. semiferanus* on Quercus and *A. negundana* on Acer.

Larval Morphology

Last instar larvae are 16-26 mm in length with a pale-green abdomen. The head is brown to brownish green and ranges from unmarked to heavily marked with black. The prothoracic shield is pale brownish green and markings range from a pair of spots on the anterolateral corners to more extensive dark patterns. Thoracic legs are pale and unmarked (Chapman and Lienk 1971).

Biology

Archips semiferanus completes one generation per year. Adults are present in late June through August.

Females lay eggs in masses of 40-50 individual eggs, often in slight depressions on the bark of the host. The egg masses are covered with specialized scales located on the ventral surface of the female abdomen. Eggs overwinter and first instar larvae hatch the following spring. Larvae feed on all types of new growth, including small fruits, and pupation occurs in the final larval feeding site (Chapman and Lienk 1971).

The primary host of *Archips semiferanus* is oak, but it has been recorded from plants in several other families (Table 32). Chapman and Lienk (1972) note that records on apple are always in the vicinity of oak. Records for boxelder and maple likely refer to *Archips negundana*.

Table 32: *Archips semiferanus* host plants

Family	**Genus/species**	**Common name**
Aceraceae	*Acer negundo* L.	boxelder
Aceraceae	*Acer* L.	maple
Fagaceae	*Quercus rubra* L.	red oak
Fagaceae	*Quercus* L.	oak
Hamamelidaceae	*Hamamelis* L.	witchhazel
Oleaceae	*Fraxinus pennsylvanica* Marsh.	green ash
Polygonaceae	*Polygonum* L.	knotweed
Rosaceae	*Malus sylvestris* (L.) Mill.	European crab apple
Rosaceae	*Malus* Mill.	apple

Distribution

Archips semiferanus is widely distributed in eastern North America and ranges as far west as Colorado (Freeman 1958).

Archips xylosteana **(Linnaeus)**

Nomenclature

Archips xylosteana (Linnaeus) (Tortricidae: Tortricinae: Archipini)

Common names: apple leaf roller, brown oak tortrix, variegated golden tortrix

Synonyms: *characterana* (*Tortrix*), *densata* (*Phalaena*), *gilvana* (*Tortrix*), *hybnerana* (*Pyralis*), *obliquana* (*Pyralis*), *pallens* (var.), *westriniana* (*Tortrix*)

Subspecies: *A. xylosteana sabrinae* (France)

Adult Recognition

Forewing ground color is pale pinkish brown. Forewing markings are dark reddish brown and include a basal patch and a well-defined median fascia that is narrow at the costa and broadened at the dorsum. The subterminal fascia gives the appearance of a costal spot on many individuals. Hindwings are grayish brown. Males have a forewing costal fold. FWL: 7.0-10.0 mm (male); 9.0-11.0 mm (female).

Male genitalia are characterized by a large, parallel-sided uncus, rounded valva, and well-defined sacculus with a short projection. Female genitalia are characterized by a very long ductus bursae with cestum and a signum in the round corpus bursae.

Archips xylosteana can appear similar to other *Archips*, including: *A. betulana*, *A. crataegana*, *A. grisea*, *A. magnoliana*, *A. oporana*, *A. podana*, and *A. rosana*. A genitalic dissection can be used to confirm identity.

Larval Morphology

Last instar larvae are greenish gray to whitish gray. The head and prothoracic shield are black or dark brown and are separated by a white line. Thoracic legs are dark brown or black (Bradley et al. 1973, Yasuda 1975).

Biology

The following life history information is summarized from Bradley et al. (1973), Hoebeke et al. (2008), and Yasuda (1975). *Archips xylosteana* completes a single generation per year. Adults are present in late June to mid-August.

Females deposit eggs in masses on branches or tree trunks. Eggs overwinter and first instar larvae hatch the following spring. Early instars feed on leaves and buds while later instars produce a leaf roll and skeletonize the leaf from inside. Pupation occurs from mid-May to June in a folded leaf and adults emerge in 9-12 days.

In Europe, this species is considered a minor pest of fruit trees, where it can cause damage to buds and new foliage. Larvae may also feed on fruitlets causing superficial damage. A list of documented host plants is provided in Table 33.

Table 33: *Archips xylosteana* host plants

Family	**Genus/species**	**Common name**
Aceraceae	*Acer* L.	maple
Betulaceae	*Alnus* Mill.	alder
Betulaceae	*Betula* L.	birch
Betulaceae	*Corylus avellana* L.	common filbert
Caprifoliaceae	*Lonicera periclymenum* L.	European honeysuckle
Caprifoliaceae	*Lonicera* L.	honeysuckle
Clusiaceae	*Hypericum* L.	St. Johnswort
Cornaceae	*Cornus controversa* Hemsl. ex Prain	giant dogwood
Fagaceae	*Castanea crenata* Siebold & Zucc.	Japanese chestnut
Fagaceae	*Castanea* Mill.	chestnut
Fagaceae	*Quercus acutissima* Carruth.	sawtooth oak
Fagaceae	*Quercus cerris* L.	European turkey oak
Fagaceae	*Quercus dentata* Thunb.	Daimyo oak
Fagaceae	*Quercus robur* L.	English oak

Table 33: *Archips xylosteana* host plants

Family	Genus/species	Common name
Fagaceae	*Quercus serrata* Thunb.	bao li
Fagaceae	*Quercus* L.	oak
Oleaceae	*Fraxinus excelsior* L.	European ash
Pinaceae	*Abies* Mill.	fir
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Malus domestica* Borkh.	apple
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Prunus armeniaca* L.	apricot
Rosaceae	*Prunus avium* (L.) L.	sweet cherry
Rosaceae	*Prunus serrulata* Lindl.	Japanese flowering cherry
Rosaceae	*Prunus* L.	
Rosaceae	*Pyrus pyrifolia* (Burm. f.) Nakai	Chinese pear
Rosaceae	*Pyrus ussuriensis* Maxim.	
Rosaceae	*Rubus* L.	raspberry
Rosaceae	*Sorbus aucuparia* L.	European mountain ash
Rutaceae	*Citrus* L.	citrus
Salicaceae	*Salix cinerea* L.	large gray willow
Salicaceae	*Salix* L.	willow
Tiliaceae	*Tilia* L.	basswood
Ulmaceae	*Ulmus davidiana* Planch.	Japanese elm
Ulmaceae	*Ulmus* L.	elm

Distribution

Archips xylosteana is distributed throughout Europe, Asia (China, Iran, Japan, Kazakhstan, Korea, Siberia, Turkey), and northern Africa (Algeria). It was first discovered in North America (Newfoundland, Canada) in 2005 (Hoebeke et al. 2008).

Argyrotaenia **Stephens**

Overview

The genus *Argyrotaenia* contains approximately 100 described species (Baixeras et al. 2008). The majority of species diversity occurs in the Nearctic and Neotropical regions. Wing patterns vary widely across the genus, although there are a number of species with golden-brown forewings and a large reddish-brown median fascia. Male genitalia are characterized by a spatulate or fingerlike uncus and rounded valvae with a long, thin sacculus. Female genitalia are characterized by a daggar-shaped signum and a sclerotized plate in the anterior end of the ductus bursae.

A single species, *A. ljungiana*, is present in the Palearctic. Larvae are polyphagous and feed on a variety of plants, sometimes causing economic damage to grape, apple, and tea. *Argyrotaenia franciscana* is a common species in California, Oregon, and Washington that has been recorded feeding on plants in more than 40 families. It is an occasional pest of crops and nursery stock in California, but is more often encountered as a non-target during *Epiphyas postvittana* surveys. The other species treated here are common non-targets encountered in general tortricid surveys.

It is difficult to distinguish between different species of *Argyrotaenia* using larval morphology. As such, most *Argyrotaenia* larvae intercepted at U.S. ports of entry are not identified beyond the subfamily or genus level.

***Argyrotaenia franciscana* (Walsingham)**

Nomenclature

Argyrotaenia franciscana (Walsingham) (Tortricidae: Tortricinae: Archipini)

Common names: orange tortrix, apple skinworm

Synonyms: *citrana* (*Tortrix*), *kearfotti* (*Argyrotaenia*)

Argyrotaenia franciscana and *A. citrana* are considered separate species in much of the economic literature. Landry et al. (1999) concluded that they form a single species based on molecular data.

Subspecies: *A. franciscana insulana* (Anacapa Island, California)

Adult Recognition

In California, *A. franciscana* has two common phenotypes that were previously separated into two species (Landry et al. 1999, Powell 1964). The typical *A. citrana* has broad, orange to orange-brown forewings with a well defined dark median fascia and outer spot on the costa, and the hindwings are white to gray. The typical *A. franciscana* has narrow, gray to brownish-gray forewings with a dark, variably defined median fascia and an outer spot on the costa, and the hindwings are primarily gray. Wing pattern and size can be quite variable although most individuals show remnants of a median fascia and outer costal spot. Males lack a forewing costal fold. FWL: 6.0-10.0 mm.

Choristoneura rosaceana males have been captured in *A. franciscana* pheremone traps; however, adults of these two species are unlikely to be confused.

Larval Morphology

The larval head and prothoracic shield are light brown and unmarked. Abdomen color vaires with host plant, but larvae are usually pale to dark green.

Larval damage by *Pandemis pyrusana* may resemble that of *A. franciscana* and the larvae are similar in appearance (Powell 1964). *Argyrotaenia franciscana* larvae are similar to those of many other Archipini that have a brown, unmarked head and prothoracic shield.

Biology

The following life history information is summarized from Powell (1964) and Powell and Opler (2009). *Argyrotaenia franciscana* is bivoltine or multivoltine, depending on location. In warmer inland areas of California, larvae aestivate during the summer and only two generations are completed. In cooler coastal areas there may be up to 5 continuous overlapping generations with adults present year-round.

Eggs are laid on smooth surfaces of leaves, fruits, and twigs in masses that contain approximately 200 individual eggs. Early instar larvae skeletonize leaves under a silk shelter; later instars roll, fold, or web leaves together or to fruits. Larvae complete 5-7 instars in a period of 20-30 days. Larvae or pupae overwinter in dead leaves, in mummified fruits, under buds, or on weedy herbaceous plants in the same vicinity as the host. Pupation occurs in the final larval shelter.

Larvae can cause economic damage by directly feeding on developing fruit in citrus, apple, and grape. Larvae may also feed on stems, causing fruit to drop. *Argyrotaenia franciscana* has been described as one of the most polyphagous tortricid species in North America (Powell 1964). Its host list includes plants in more than 40 families, many of which are important crops (Table 34).

Table 34: *Argyrotaenia franciscana* host plants

Family	**Genus/species**	**Common name**
Anacardiaceae	*Schinus molle* L.	Peruvian peppertree
Aquifoliaceae	*Ilex opaca* Aiton	American holly
Araliaceae	*Hedera helix* L.	English ivy
Asteraceae	*Achillea millefolium* L.	common yarrow
Asteraceae	*Artemisia californica* Less.	coastal sagebrush
Asteraceae	*Artemisia douglasiana* Besser	Douglas' sagewort
Asteraceae	*Baccharis pilularis* DC.	coyotebrush
Asteraceae	*Baccharis* L.	baccharis
Asteraceae	*Cirsium occidentale* (Nutt.) Jeps.	cobwebby thistle
Asteraceae	*Coreopsis gigantea* (Kellogg) H. M. Hall	giant coreopsis
Asteraceae	*Coreopsis* L.	tickseed
Asteraceae	*Corethrogyne* DC.	sandaster
Asteraceae	*Encelia californica* Nutt.	California brittlebush
Asteraceae	*Erigeron glaucus* Ker Gawl.	seaside fleabane
Asteraceae	*Eriophyllum staechadifolium* Lag.	
Asteraceae	*Gnaphalium* L.	cudweed
Asteraceae	*Grindelia camporum* Greene	Great Valley gumweed
Asteraceae	*Grindelia hirsutula* Hook. & Arn.	hairy gumweed
Asteraceae	*Grindelia* Willd.	gumweed
Asteraceae	*Isocoma veneta* (Kunth) Greene	goldenbush
Asteraceae	*Lessingia* Cham.	lessingia
Asteraceae	*Pericallis hybrida* B. Nord.	common ragwort
Asteraceae	*Pseudognaphalium biolettii* Anderb.	two-color rabbit-tobacco
Asteraceae	*Pseudognaphalium californicum* (DC.) Anderb.	ladies' tobacco
Asteraceae	*Senecio jacobaea* L.	stinking willie

Table 34: *Argyrotaenia franciscana* host plants

Family	**Genus/species**	**Common name**
Asteraceae	*Solidago simplex* Kunth	Mt. Albert goldenrod
Asteraceae	*Solidago* L.	goldenrod
Begoniaceae	*Begonia* L.	begonia
Berberidaceae	*Mahonia pinnata* (Lag.) Fedde	wavyleaf barberry
Berberidaceae	*Mahonia* Nutt.	barberry
Buddlejaceae	*Buddleja* L.	butterflybush
Caprifoliaceae	*Lonicera involucrata* (Richardson) Banks ex Spreng.	twinberry honeysuckle
Caprifoliaceae	*Sambucus nigra* L. ssp. *cerulea* (Raf.) R. Bolli	blue elderberry
Caprifoliaceae	*Symphoricarpos albus* (L.) S. F. Blake	common snowberry
Caryophyllaceae	*Dianthus caryophyllus* L.	carnation
Caryopyllaceae	*Spergularia macrotheca* (Hornem.) Heynh.	sticky sandspurry
Chenopodiaceae	*Chenopodium* L.	goosefoot
Commelinaceae	*Tradescantia zebrina* hort. ex Bosse	inchplant
Convolvulaceae	*Calystegia macrostegia* (Greene) Brummitt	island false bindweed
Convolvulaceae	*Convolvulus* L.	bindweed
Crassulaceae	*Dudleya farinosa* (Lindl.) Britt. & Rose	powdery liveforever
Crassulaceae	*Sedum spathulifolium* Hook.	broadleaf stonecrop
Cupressaceae	*Chamaecyparis lawsoniana* (A. Murray) Parl.	Port Orford cedar
Cupressaceae	*Cupressus macrocarpa* Hartw. ex Gord.	Monterey cypress
Cupressaceae	*Sequoia sempervirens* (Lamb. ex D. Don) Endl.	redwood
Cupressaceae	*Thuja plicata* Donn ex D. Don	western redcedar
Dennstaedtiaceae	*Pteridium aquilinum* (L.) Kuhn	western brackenfern
Dryopteridaceae	*Dryopteris arguta* (Kaulf.) Watt	coastal woodfern
Ericaceae	*Arbutus menziesii* Pursh	Pacific madrone
Ericaceae	*Arbutus* L.	madrone
Ericaceae	*Arctostaphylos imbricata* Eastw.	San Bruno Mountain manzanita
Ericaceae	*Vaccinium ovatum* Pursh	California huckleberry
Fabaceae	*Acacia* Mill.	acacia
Fabaceae	*Astragalus miguelensis* Greene	San Miguel milkvetch
Fabaceae	*Cytisus scoparius* (L.) Link	Scotch broom
Fabaceae	*Lotus scoparius* (Nutt.) Ottley	common deerweed
Fabaceae	*Lotus* L.	trefoil
Fabaceae	*Lupinus arboreus* Sims	yellow bush lupine
Fabaceae	*Lupinus chamissonis* Eschsch.	Chamisso bush lupine
Fabaceae	*Lupinus* L.	lupine
Fagaceae	*Quercus agrifolia* Nee	California live oak
Fagaceae	*Quercus douglasii* Hook. & Arn.	blue oak
Fagaceae	*Quercus dumosa* Nutt.	coastal sage scrub oak
Fagaceae	*Quercus* L.	oak

Table 34: *Argyrotaenia franciscana* host plants

Family	**Genus/species**	**Common name**
Geraniaceae	*Erodium* L'Her. ex Aiton	stork's bill
Geraniaceae	*Geranium* L.	geranium
Geraniaceae	*Pelargonium* L'Her. ex Aiton	geranium
Grossulariaceae	*Ribes amarum* McClatchie	bitter gooseberry
Grossulariaceae	*Ribes malvaceum* Sm.	chaparral currant
Grossulariaceae	*Ribes menziesii* Pursh	canyon gooseberry
Grossulariaceae	*Ribes sanguineum* Pursh	redflower currant
Grossulariaceae	*Ribes* L.	currant
Hippocastanaceae	*Aesculus californica* (Spach) Nutt.	California buckeye
Hydrophyllaceae	*Eriodictyon californicum* (Hook. & Arn.) Torr.	California yerba santa
Hydrophyllaceae	*Phacelia malvifolia* Cham.	stinging phacelia
Juglandaceae	*Juglans regia* L.	English walnut
Juglandaceae	*Juglans* L.	walnut
Lamiaceae	*Lavandula angustifolia* Mill.	English lavender
Lamiaceae	*Monardella villosa* Benth.	coyote mint
Lamiaceae	*Monardella* Benth.	monardella
Lamiaceae	*Stachys bullata* Benth.	California hedgenettle
Lamiaceae	*Stachys* L.	hedgenettle
Lauraceae	*Persea americana* Mill.	avocado
Lauraceae	*Umbellularia californica* (Hook. & Arn.) Nutt.	California laurel
Liliaceae	*Asparagus* L.	asparagus
Liliaceae	*Calochortus catalinae* S. Watson	Santa Catalina mariposa lily
Liliaceae	*Calochortus* Pursh	mariposa lily
Malvaceae	*Sphaeralcea ambigua* A. Gray	desert globemallow
Myricaceae	*Morella californica* (Cham.) Wilbur	California wax myrtle
Myrtaceae	*Eucalyptus* L'Her.	gum
Onagraceae	*Epilobium canum* (Greene) P.H. Raven ssp. Canum	hummingbird trumpet
Onagraceae	*Epilobium* L.	willowherb
Onagraceae	*Oenothera* L.	evening primrose
Pinaceae	*Abies* Mill.	fir
Pinaceae	*Cedrus deodara* (Roxb.) G. Don f.	Deodar cedar
Pinaceae	*Picea* A. Dietr.	spruce
Pinaceae	*Picea pungens* Engelm.	blue spruce
Pinaceae	*Pinus radiata* D. Don	Monterey pine
Pinaceae	*Pinus* L.	pine
Pinaceae	*Pseudotsuga menziesii* (Mirb.) Franco	Douglas-fir
Pinaceae	*Pseudostuga* Carriere	Douglas-fir
Pinaceae	*Tsuga canadensis* (L.) Carriere	eastern hemlock
Pinaceae	*Tsuga Carriere*	hemlock

Table 34: *Argyrotaenia franciscana* host plants

Family	**Genus/species**	**Common name**
Poaceae	*Coix lacryma-jobi* L.	Job's tears
Polygonaceae	*Eriogonum latifolium* Sm.	seaside buckwheat
Polygonaceae	*Eriogonum parvifolium* Sm.	seacliff buckwheat
Proteaceae	*Macadamia* F. Muell.	macadamia
Ranunculaceae	*Aquilegia* L.	columbine
Rhamnaceae	*Ceanothus arboreus* Greene	feltleaf ceanothus
Rhamnaceae	*Ceanothus oliganthus* Nutt.	hairy ceanothus
Rhamnaceae	*Ceanothus sorediatus* Hook. & Arn.	jimbrush
Rhamnaceae	*Ceanothus thyrsiflorus* Eschsch.	blueblossom
Rhamnaceae	*Ceanothus* L.	ceanothus
Rhamnaceae	*Frangula californica* (Eschsch.) A. Gray	California buckthorn
Rosaceae	*Adenostoma* Hook. & Arn.	chamise
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Eriobotrya japonica* (Thunb.) Lindl.	loquat
Rosaceae	*Fragaria vesca* L.	woodland strawberry
Rosaceae	*Fragaria vesca* L. ssp. *californica* (Cham. & Schltdl.) Staudt	California strawberry
Rosaceae	*Heteromeles arbutifolia* (Lindl.) M. Roem.	toyon
Rosaceae	*Heteromeles arbutifolia* (Lindl.) M. Roem. var. *arbutifolia*	toyon
Rosaceae	*Horkelia californica* Cham. & Schltdl.	California horkelia
Rosaceae	*Lyonothamnus floribundus* A. Gray ssp. *aspleniifolius* (Greene) P. H. Raven	fern-leaf Catalina ironwood
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Malus sylvestris* (L.) Mill.	European crab apple
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Potentilla* L.	cinquefoil
Rosaceae	*Prunus armeniaca* L.	apricot
Rosaceae	*Prunus avium* (L.) L.	sweet cherry
Rosaceae	*Prunus domestica* L.	European plum
Rosaceae	*Prunus dulcis* (Mill.) D. A. Webb	sweet almond
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Prunus* L.	
Rosaceae	*Pyracantha* M. Roem.	firethorn
Rosaceae	*Rosa* L.	rose
Rosaceae	*Rubus parviflorus* Nutt.	thimbleberry
Rosaceae	*Rubus ursinus* Cham. & Schltdl.	California blackberry
Rosaceae	*Rubus vitifolius* Cham. & Schltdl.	Pacific dewberry
Rosaceae	*Rubus* L.	cranberry
Rutaceae	*Citrus* L.	citrus
Salicaceae	*Salix lasiolepis* Benth.	arroyo willow
Salicaceae	*Salix* L.	willow

Table 34: *Argyrotaenia franciscana* host plants

Family	**Genus/species**	**Common name**
Sapindaceae	*Filicium decipiens* Thwaites	
Scrophulariaceae	*Castilleja affinis* Hook. & Arn.	coast Indian paintbrush
Scrophulariaceae	*Castilleja exserta* (A. Heller) T.I. Chuang & Heckard ssp. *exserta*	exserted Indian paintbrush
Scrophulariaceae	*Castilleja* Mutis ex L. f.	Indian paintbrush
Scrophulariaceae	*Diplacus aurantiacus* (W. Curtis) Jeps. ssp. *aurantiacus*	orange bush monkeyflower
Scrophulariaceae	*Scrophularia californica* Cham. & Schltdl.	California figwort
Scrophulariaceae	*Scrophularia* L.	figwort
Scrophulariaceae	*Veronica* L.	speedwell
Solanaceae	*Solanum douglasii* Dunal	greenspot nightshade
Solanaceae	*Solanum pseudocapsicum* L.	Jerusalem cherry
Thymelaeaceae	*Dirca occidentalis* A. Gray	western leatherwood
Urticaceae	*Urtica* L.	nettle
Verbenaceae	*Lantana* L.	lantana
Vitaceae	*Vitis vinifera* L.	wine grape

Distribution

Argyrotaenia franciscana is found in California, Oregon, and Washington, primarily in the cooler coastal areas and river valleys (Powell and Opler 2009).

Argyrotaenia ljungiana **(Thunberg)**

Nomenclature

Argyrotaenia ljungiana (Thunberg) (Tortricidae: Tortricinae: Archipini)

Common names: grape tortrix

Synonyms: *cognatana* (*Argyrotaenia*), *fuscociliana* (*Argyrotaenia*), *lepidana* (*Argyrotaenia*), *micantana* (*Olethreutes*), *micanthana* (*Olethreutes*), *politana* (*Tortrix*), *pulchellana* (*Tortrix*), *sylvana* (*Tortrix*)

Adult Recognition

Forewing ground color ranges from pale brown to silvery white. Markings are dark reddish brown to gray and usually include a well-defined median fascia and outer costal spot. Hindwings are grayish brown. Males lack a forewing costal fold. FWL: 5.5-8.5 mm.

Adults may appear similar to other species of *Argyrotaenia*, including *A. velutinana* and *A. pinatubana*. A genitalic dissection is necessary to confirm identity. Male *A. velutinana* have a distal, pointed projection from the median sclerotized portion of the valva that is absent in *A. ljungiana*. The uncus of male *A. pinatubana* is broader and slightly expanded distally compared to the uncus of *A. ljungiana*.

Larval Morphology

Late instar larvae are pale green with a yellowish brown head. The prothoracic shield is yellowish green with a black posterolateral mark and black shading on the posterior margin. An anal comb with 6-8 teeth is present (Bradley et al. 1973).

Biology

The following life history information is summarized from Bradley et al. (1973) and Ovsyannikova and Grichanov (2009). *Argyrotaenia ljungiana* completes 1-2 generations in most of Europe and 2-3 generations in Transcaucasia (Caucasus Mountains). Adults are present April-May and June-July, and again in September if there is a third generation.

Females deposit eggs in masses of 40-50 individual eggs on the upper surface of leaves. Early instar larvae skeletonize the underside of leaves along the midrib. Later instars roll or fold leaves and may also feed on buds and fruits. Pupation occurs in debris on the ground, in webbed leaves, or in bark crevices. Overwintering occurs in the pupal stage.

This species is considered a pest of fruit and field crops and ornamentals in southern Europe and the European part of the former USSR. It has been recorded damaging a variety of plants, with most economic losses occuring in apple, grape, and tea (Table 35).

Distribution

Argyrotaenia ljungiana is distributed from Western Europe east to China and Japan (Bradley et al. 1973, Razowski 2002).

Bradley et al. (1973) reported this species as occuring in North America; the source of this record is probably related to a misidentification of *A. pinatubana* as *Tortrix politana* (= *ljungiana*) by Zeller, which was then reported by Packard (1890). Freeman (1944) correctly synonymizes the Packard *T. politana* under *A. pinatubana*.

Table 35: *Argyrotaenia ljungiana* host plants

Family	**Genus/species**	**Common name**
Asteraceae	*Centaurea* L.	knapweed
Asteraceae	*Chrysanthemum* L.	daisy
Asteraceae	*Tripolium pannonicum* (Jacq.) Dobrocz.	sea aster
Betulaceae	*Betula* L.	birch
Ericaceae	*Calluna vulgaris* (L.) Hull	heather
Ericaceae	*Erica* L.	heath
Ericaceae	*Vaccinium uliginosum* L.	bog blueberry
Ericaceae	*Vaccinium* L.	blueberry
Fabaceae	*Colutea arborescens* L.	bladder senna
Gentianaceae	*Gentiana pneumonanthe* L.	
Lamiaceae	*Lavandula* L.	lavender
Myricaceae	*Myrica gale* L.	sweetgale
Pinaceae	*Abies* Mill.	fir
Pinaceae	*Larix decidua* Mill.	European larch
Pinaceae	*Picea* A. Dietr.	spruce
Pinaceae	*Pinus* L.	pine
Poaceae	*Zea mays* L.	corn
Ranunculaceae	*Ranunculus* L.	buttercup
Rosaceae	*Drayas octopetala* L.	
Rosaceae	*Malus sylvestris* (L.) Mill.	European crab apple
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus armeniaca* L.	apricot
Rosaceae	*Prunus* L.	
Rosaceae	*Pyracantha* M. Roem.	firethorn
Rutaceae	*Citrus* L.	citrus
Theaceae	*Camellia sinensis* (L.) Kuntze	tea
Vitaceae	*Vitis vinifera* L.	wine grape

***Argyrotaenia mariana* (Fernald)**

Nomenclature

Argyrotaenia mariana (Fernald) (Tortricidae: Tortricinae: Archipini)

Common names: gray-banded leaf roller

Adult Recognition

Forewing ground color is light gray to white. Markings include a dark-brown to black cresent on the costa, a light-brown patch in the area of the median fascia, and sparse pale-brown shading on the basal half of the wing in some individuals. The hindwing is gray. Males lack a forewing costal fold FWL: 7.0-10.0 mm.

The general forewing coloration and pattern is similar to some species of *Acleris*, such as *A. forbesana* and *A. nivisellana*. A genitalic dissection can be used to confirm identity.

Larval Morphology

Last instar larvae are approximately 17-23 mm in length and are largely unmarked. The abdomen is light green, the head and prothoracic shield are yellowish green, and the anal shield may be lightly mottled with dark green spots (Chapman and Lienk 1971).

The larvae can be confused with those of many other tortricids, including other *Argyrotaenia*, *Epiphyas postvittana*, and *Choristoneura rosaceana*. According to Chapman and Lienk (1971), the markings on the anal shield can be used to separate *A. mariana* from other *Argyrotaenia* species.

Biology

Argyrotaenia mariana completes a single generation per year. Adults are present in May and June.

Females lay eggs in masses of 20-120 individual eggs on the upper surface of leaves and smooth bark. First instar larvae immediately disperse from the egg mass on silken threads, which transport them to different parts of the plant or entirely different plants. Early instars skeletonize the underside of leaves along the midrib. Later instars roll or web leaves, or web leaves to fruit. This species has been recorded as an infrequent pest of apple in the Northeast (Chapman and Lienk 1971).

Although larvae of *A. mariana* have been recorded from a variety of plants (Table 36), Chapman and Lienk (1971) speculate that its primary hosts are restricted to the family Rosaceae.

Table 36: *Argyrotaenia mariana* host plants

Family	**Genus/species**	**Common name**
Aceraceae	*Acer* L.	maple
Asteraceae	*Prenanthes trifoliolata* (Cass.) Fernald	gall of the earth
Asteraceae	*Prenanthes* L.	rattlesnakeroot
Betulaceae	*Alnus* Mill.	alder
Betulaceae	*Betula alleghaniensis* Britton	yellow birch
Betulaceae	*Betula papyrifera* Marshall	paper birch
Betulaceae	*Betula populifolia* Marsh.	gray birch
Betulaceae	*Betula* L.	birch
Caprifoliaceae	*Viburnum* L.	viburnum
Ericaceae	*Kalmia angustifolia* L.	sheep laurel
Ericaceae	*Vaccinium uliginosum* L.	bog blueberry
Ericaceae	*Vaccinium* L.	blueberry
Fagaceae	*Quercus* L.	oak
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Malus sylvestris* (L.) Mill.	European crab apple
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus virginiana* L.	chokecherry
Rosaceae	*Pyrus* L.	pear
Salicaceae	*Populus tremuloides* Michx.	quaking aspen
Salicaceae	*Salix* L.	willow
Ulmaceae	*Ulmus americana* L.	American elm

Distribution

Argyrotaenia mariana is found in eastern North America, where it is most common in the Northeast and southern Canada (Chapman and Lienk 1971, Freeman 1958).

***Argyrotaenia quadrifasciana* (Fernald)**

Nomenclature

Argyrotaenia quadrifasciana (Fernald) (Tortricidae: Tortricinae: Archipini)

Common names: lesser all-green leaf roller, four-banded leaf roller

Adult Recognition

Forewings are bright yellow with variable orange to orange-brown markings and reticulations. There are often two conspicuous orange fasciae running from costa to dorum, but these may be obscured by the reticulated pattern in some individuals. The male hindwings are dark brown, while the female hindwings are golden brown. Males lack a forewing costal fold. FWL: 6.5-8.5 mm.

Adults of *A. quercifoliana* may appear similar to those of *A. quadrifasciana*. The two species can be distinguished by forewing color: bright yellow in *A. quadrifasciana* versus pale yellow in *A. quercifoliana*. In addition, the hindwings of *A. quercifoliana* are white, while those of *A. quadrifasciana* are dark brown or golden brown (Freeman 1958).

Larval Morphology

Last instar larvae are 12-17 mm in length. The abdomen, head, and prothoracic shield are yellowish green and unmarked (Chapman and Lienk 1971). The green unmarked larva can be confused with the larva of many other tortricids, including other species of *Argyrotaenia*, *Epiphyas postvittana*, and *Choristoneura rosaceana*.

Biology

Argyrotaenia quadrifasciana completes one generation per year. Adults are present in late June and July.

Females deposit eggs in masses that contain approximately 50 individual eggs on the upper surface of leaves of the host plant. Early instar larvae skeletonize the underside of a leaf near the midrib under a patch of silk. Third instar larvae leave the feeding site and construct a hibernaculum in the crotches of twigs and bark crevices, where they overwinter until the following spring. In the spring, larvae leave the hibernaculum and construct a nest by webbing together terminal shoot growth, where they feed inside on shoots and buds. Pupation occurs in the final larval feeding site (Chapman and Lienk 1971). A list of documented host plants is provided in Table 37.

Table 37: *Argyrotaenia quadrifasciana* host plants

Family	**Genus/species**	**Common name**
Rosaceae	*Amelanchier alnifolia* (Nutt.) Nutt. ex M. Roem.	Saskatoon serviceberry
Rosaceae	*Amelanchier* Medik.	serviceberry
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Malus sylvestris* (L.) Mill.	European crab apple
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus pensylvanica* L. f.	pin cherry
Rosaceae	*Prunus serotina* Ehrh.	black cherry
Rosaceae	*Prunus virginiana* L.	chokecherry
Rosaceae	*Prunus* L.	cherry
Rosaceae	*Pyrus* L.	pear

Distribution

Argyrotaenia quadrifasciana is distributed in eastern North America, where it is more common in the Northeast and southern Canada (Chapman and Lienk 1971, Freeman 1958).

***Argyrotaenia quercifoliana* (Fitch)**

Nomenclature

Argyrotaenia quercifoliana (Fitch) (Tortricidae: Tortricinae: Archipini)

Synonyms: *trifurculana* (*Tortrix*)

Adult Recognition

Forewings are pale yellow with brown to light-brown reticulated markings. Most specimens have two brown fascial remnants that form parallel lines from costa to dorsum. Hindwings are white. Males lack a forewing costal fold. FWL: 7.5-9.5 mm (male); 9.0-11.5 mm (female).

Adults of *A. quadrifasciana* may appear similar to those of *A. quercifoliana*. The two species can be distinguished by forewing color: bright yellow in *A. quadrifasciana* versus pale yellow in *A. quercifoliana*. In addition, the hindwings of *A. quercifoliana* are white, while those of *A. quadrifasciana* are dark brown or golden brown. According to Freeman (1944), some individuals exhibit a solid patch of light brown on the forewing and resemble *A. alisellana*.

Biology

Adults are present in May through July. Larvae feed on leaves of oak and various other hosts (Table 38) (Freeman 1944, 1958). Full life history information for this species has not been reported.

Table 38: *Argyrotaenia quercifoliana* host plants

Family	**Genus/species**	**Common name**
Aceraceae	*Acer spicatum* Lam.	mountain maple
Anacardiaceae	*Rhus* L.	sumac
Fagaceae	*Quercus alba* L.	white oak
Fagaceae	*Quercus macrocarpa* Michx.	bur oak
Fagaceae	*Quercus palustris* Munchh.	pin oak
Fagaceae	*Quercus rubra* L.	red oak
Fagaceae	*Quercus velutina* Lam.	black oak
Fagaceae	*Quercus* L.	oak
Hamamelidaceae	*Hamamelis virginiana* L.	American witchhazel
Hamamelidaceae	*Hamamelis* L.	witchhazel
Rhamnaceae	*Rhamnus cathartica* L.	common buckthorn
Rhamnaceae	*Rhamnus* L.	buckthorn
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Rubus* L.	blackberry

Distribution

Argyrotaenia quercifoliana is widely distributed in eastern North America from southern Canada to Florida (Freeman 1944, 1958).

Argyrotaenia velutinana **(Walker)**

Nomenclature

Argyrotaenia velutinana (Walker) (Tortricidae: Tortricinae: Archipini)

Common names: red-banded leaf roller (RBLR)

Synonyms: *incertana* (*Tortrix*), *lutosana* (*Tortrix*), *triferana* (*Cacoecia*)

Adult Recognition

Forewing ground color ranges from pale brown to golden brown. The most conspicuous wing marking is the reddish-brown median fascia, which is the basis for the species' common name. Other markings can be quite variable, although generally there is a dark mark or partial fascia at the base of the wing, a reddish-brown outer costal spot, and a row of near-white scales along the termen that may extend to the median fascia in some individuals. Hindwings are grayish brown. Males lack a forewing costal fold. FWL: 5.5-7.5 mm (male); 6.5-8.0 mm (female).

Adults can appear similar to other species of *Argyrotaenia*. In the Nearctic, this includes species such as *A. floridana*, *A. kimballi*, *A. niscana*, *A. pinatubana*, and *A. tabulana*. In the Palearctic, *A. ljungiana* may appear similar. A genitalic dissection can be used to confirm identity. Male *A. velutinana* have a distal, pointed projection from the median sclerotized portion of the valva that is absent in *A. ljungiana*.

Larval Morphology

Late instar larvae are 13-18 mm in length with a green to yellowish green abdomen. The head, prothoracic shield, and thoracic legs are yellowish green and unmarked (Chapman and Lienk 1971). Larvae can be confused with those of many other tortricids, including other species of *Argyrotaenia*, *Epiphyas postvittana*, and *Choristoneura rosaceana*.

Biology

The following life history information is summarized from Chapman and Lienk (1971) and Summerland and Hamilton (1955). *Argyrotaenia velutinana* completes 2-3 full generations over much of its range. Because this species undergoes facultative diapause, the number of generations can vary depending on latitude. In the North, only two generations are completed, with a partial third possible. In the South, a possible fourth generation is present. Overwintering occurs in the pupal stage. In New York, adults of the overwintering (second) generation are present in April and May. Those of the first generation are present in late June to July. In southern Indiana and Virginia, adults of the overwintering (fourth) generation are present in March and April. Those of the first generation are present in late May to June, those of the second generation are present in July, and those of the third generation present in August and September.

In the spring, females lay eggs in masses on smooth bark of the trunk and lower limbs of host trees. During the summer, females lay egg masses on the upper surface of leaves. Each egg mass contains approximately 40-45 individual eggs. Egg development time ranges from 7-12 days in the South to 14-21 days in the North. First instar larvae crawl up limbs in search of food or disperse on silk threads to other parts of the host or to other plants. Early instars skeletonize the upper surface of a leaf along the midrib, concealed by a patch of silk. They remain under the silk patch until the penultimate instar, at which point they move to feed on other leaves or fruit. Late instar larvae of the summer generations will often construct a shelter by webbing a leaf to fruit, and feeding underneath directly on the fruit. Larval feeding damage causes fruit rot and early drop in hosts such as apple. Larvae will continue to feed on fallen fruit and may be dispersed in this manner if fallen fruit is moved to a different location. Larvae complete development in approximately 30 days and move to the ground to pupate in a folded leaf under other leaves and debris. Adults of the first two generations eclose in 7-13 days; those of the last generation eclose the following spring.

Argyrotaenia velutinana was once considered one of the most important tortricid pests on apple in the eastern United States (Chapman and Lienk 1971). Its status as a major pest peaked after the widespread use of DDT in the late 1940's presumably destroyed many of its natural enemies. It is currently controlled under most IPM programs and is only considered a minor pest.

Larvae of *Argyrotaenia velutinana* are highly polyphagous and have been described by Freeman (1958) as feeding "on almost any plant." This includes several conifers, as reported by Prentice (1966). Chapman and Lienk (1971) speculate that primary hosts may be limited to members of the Rosaceae, as apple appears to be a preferred host in many regions. A list of documented host plants is provided in Table 39.

Distribution

Argyrotaenia velutinana is widely distributed in eastern North America (Chapman and Lienk 1971, Freeman 1958).

Table 39: *Argyrotaenia velutinana* host plants

Family	**Genus/species**	**Common name**
Aceraceae	*Acer* L.	maple
Apocynaceae	*Apocynum* L.	dogbane
Aquifoliaceae	*Ilex decidua* Walter	possumhaw
Asteraceae	[unspecified]	
Asteraceae	*Ambrosia trifida* L.	great ragweed
Asteraceae	*Chrysanthemum* L.	daisy
Asteraceae	*Zinnia violacea* Cav.	elegant zinnia
Betulaceae	*Alnus* Mill.	alder
Betulaceae	*Betula papyrifera* Marshall	paper birch
Campanulaceae	*Lobelia* L.	lobelia
Caprifoliaceae	*Lonicera* L.	honeysuckle
Ericaceae	*Vaccinium* L.	blueberry
Fagaceae	*Quercus* L.	oak
Geraniaceae	*Geranium* L.	geranium
Malvaceae	*Alcea rosea* L.	hollyhock
Myricaceae	*Myrica gale* L.	sweetgale
Orchidaceae	*Platanthera cristata* (Michx.) Lindl.	crested yellow orchid

Table 39: *Argyrotaenia velutinana* host plants

Family	**Genus/species**	**Common name**
Pinaceae	*Abies balsamea* (L.) Mill.	balsam fir
Pinaceae	*Larix* Mill.	larch
Pinaceae	*Picea glauca* (Moench) Voss	white spruce
Pinaceae	*Picea mariana* (Mill.) Britton, Sterns & Poggenb.	black spruce
Pinaceae	*Picea rubens* Sarg.	red spruce
Pinaceae	*Pinus sylvestris* L.	Scots pine
Pinaceae	*Tsuga canadensis* (L.) Carriere	eastern hemlock
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus domestica* L.	European plum
Rosaceae	*Prunus pensylvanica* L. f.	pin cherry
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Prunus serotina* Ehrh.	black cherry
Rosaceae	*Rosa* L.	rose
Salicaceae	*Populus tremuloides* Michx.	quaking aspen
Salicaceae	*Salix* L.	willow
Tiliaceae	*Tilia americana* L.	American basswood
Ulmaceae	*Ulmus americana* L.	American elm
Violaceae	*Viola* L.	violet
Vitaceae	*Vitis vinifera* L.	wine grape

Cacoecimorpha pronubana (Hübner)

Nomenclature

Cacoecimorpha pronubana (Hübner) (Tortricidae: Tortricinae: Archipini)

Common names: carnation tortrix, European carnation moth

Synonyms: *ambustana* (*Tortrix*), *hermineana* (*Tortrix*), *insolatana* (*Tortrix*), *perochreana* (*Tortrix*)

Adult Recognition

Forewings are orangish brown to dark brown. A dark-brown median fascia is usually expressed in males while females are lighter with dark reticulations. The hindwings in both sexes are a distinctive bright orange and black, although many females lack black scaling on the hindwings. Males lack a forewing costal fold. FWL: 6.5-8.5 mm (male); 7.5-11.5 mm (female).

Males fly during the day or early evening and have been observed “swarming” around sources of the female pheromone, sometimes in large numbers. Males have been captured in pheromone traps using pheromones from several other species, including *Clepsis spectrana*, *Pandemis heparana*, and *Adoxophyes orana* (E. LaGasa, pers. comm.). Individuals of *C. pronubana* can be separated from most other tortricids by their bright orange and black hindwings.

Larval Morphology

Late instar larvae are green to dark green with conspicuous light green pinacula, white seate, and dark brown to black spiracles. The head is green to yellowish brown with a dark brown to black posterolateral dash and variable dark brown to black markings on the posterior margin. The prothoracic shield is brownish green to yellowish brown, usually with distinctive dark brown to black markings on the posterolateral corners and other small markings near the mid-dorsal line. Thoracic legs are brown. The anal shield is variably mottled with brown or black, and an anal comb is present with 6 teeth in most individuals (Bradley et al. 1973, Wyoski and Izhar 1976).

Biology

The following life history information is summarized from Chapman and Lienk (1971), Van de Vrie (1991), and Wyoski and Izhar (1976). Number of generations per year varies from two generations in northern regions to 4-6 continuously overlapping generations in southern regions, where adults may be present year round. In Washington, adults are present in May through the end of September.

Females deposit eggs in small groups. First instar larvae mine leaves or buds, while later instars roll or web leaves and terminals. Larvae may cause damage to fruit by webbing leaves to fruit or feeding between adjacent fruits. In areas where there are not continuous generations, overwintering occurs in the larval stage.

Larvae of *C. pronubana* are highly polyphagous and have been recorded from more than 160 species of plants in 42 families (Table 40). As its common name would suggest, this species is most often encountered as a pest of flowers in greenhouses.

Distribution

Cacoecimorpha pronubana is native to Northern Africa and it is widely distributed across Western Europe to Asia Minor (Razowski 2002). It has been introduced into South Africa and North America, where it is present in the Pacific Northwest (Oregon and Washington). In early 2011 this species was discovered in California feeding on *Daphne odora* (winter daphne) (M. Epstein, pers. comm.).

Table 40: *Cacoecimorpha pronubana* host plants

Family	**Genus/species**	**Common name**
Apiaceae	*Aegopodium podagraria* L.	bishop's goutweed
Apocynaceae	*Neruim* L.	oleander
Apocynaceae	*Vinca* L.	periwinkle
Asteraceae	*Aster* L.	aster
Asteraceae	*Hieracium* L.	hawkweed
Brassicaceae	*Brassica oleracea* L.	cabbage
Brassicaceae	*Brassica* L.	mustard
Caryophyllaceae	*Dianthus* L.	pink
Celastraceae	*Euonymus japonicus* Thunb.	Japanese spindletree
Celastraceae	*Euonymus* L.	spindletree
Cupressaceae	*Chamaecyparis pistifera* (Siebold & Zucc.) Endl.	
Cupressaceae	*Cupressocyparis leylandii* (Dallim. & A.B. Jacks.) Dallim.	
Cupressaceae	*Juniperus scopulorum* Sarg.	Rocky Mountain juniper
Elaeagnaceae	*Hippophae rhamnoides* L.	seaberry
Ericaceae	*Arbutus* L.	madrone
Ericaceae	*Rhododendron* L.	rhododendron
Ericaceae	*Vaccinium corymbosum* L.	highbush blueberry
Ericaceae	*Vaccinium* L.	blueberry
Euphorbiaceae	*Euphorbia amygdaloides* L.	wood spurge
Fabaceae	*Robinia pseudoacacia* L.	black locust
Fabaceae	*Trifolium* L.	clover
Fabaceae	*Vicia faba* L.	horsebean
Geraniaceae	*Pelargonium* L'Her. ex Aiton	geranium

Table 40: *Cacoecimorpha pronubana* host plants

Family	**Genus/species**	**Common name**
Lauraceae	*Laurus nobilis* L.	sweet bay
Lauraceae	*Persea americana* Mill.	avocado
Liliaceae	*Narcissus* L.	daffodil
Oleaceae	*Ligustrum* L.	privet
Oleaceae	*Olea europaea* L.	olive
Onagraceae	*Fuchsia* L.	fuchsia
Pinaceae	*Pinus halepensis* Mill.	aleppo pine
Polygonaceae	*Rumex crispus* L.	curly dock
Punicaceae	*Punica granatum* L.	pomegranate
Rosaceae	*Fragaria* L.	strawberry
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus* L.	plum
Rosaceae	*Prunus* L.	cherry
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Pyrus* L.	pear
Rosaceae	*Rosa* L.	rose
Rutaceae	*Citrus reticulata* Blanco	tangerine
Rutaceae	*Citrus* L.	citrus
Solanaceae	*Capsicum* L.	pepper
Solanaceae	*Solanum lycopersicum* L. var. *lycopersicum*	garden tomato
Tamaricaceae	*Tamarix* L.	tamarisk
Thymelaeceae	*Daphne* L.	daphne
Vitaceae	*Vitis* L.	grape

***Choristoneura occidentalis* (Walsingham)**

Nomenclature

Choristoneura occidentalis (Walsingham) (Tortricidae: Tortricinae: Archipini)

This species is not to be confused with the Nearctic western spruce budworm (formally *C. occidentalis* Freeman). Razowski (2008) transferred the former *Archips occidentalis* into *Choristoneura* and proposed *C. freemani* as a replacement name for the western spruce budworm.

Commons names: citrus leafroller

Adult Recognition

Forewings are a mix of purplish brown and yellowish brown, with a dark reddish-brown median fascia and costal spot. Hindwings and abdomen are orange. Males are more heavily marked than females and dark forms exist where the hindwings and abdomen are grayish brown. Females are larger than males, and the costal margin of the female forewing is sinuate. Males have a short forewing costal fold. FWL: 9.0-13.0 mm.

Male genitalia are characterized by a broad uncus, large gnathos, and membranous valvae. Female genitalia are characterized by a twisted ductus bursae and single signum in the corpus bursae.

Choristoneura heliaspis (= *Cacoecia heliaspis*) was synonymized under *Archips occidentalis* by Brown (2005). Razowski (2008) resurrected the name based on minor differences in the male sacculus and placed it in the genus *Choristoneura*.

Biology

Females lay eggs in masses on the upper surface of leaves and cover them with a white protective secretion. On citrus, larvae prefer to feed on young fruit under the calyx, causing damage similar to that of citrus thrips. Larvae will also feed on leaves, young growth, and web leaves to fruit. Pupation occurs in rolled or webbed leaves. In South Africa, most damage to oranges occurs from October to November (Jeppson 1989, Pinhey 1975).

In South Africa, *C. occidentalis* is a pest of citrus, avocado, coffee, and various ornamentals (Table 41). Larvae of the apple leafroller (*Lozotaenia capensana*) cause similar damage to crops such as avocado (Erichsen and Schoeman 1994).

Distribution

A native of South Africa, *C. occidentalis* is widely distribued throughout Sub-Saharan Africa as evident by records from Gambia, Kenya, Mozambique, Rhodesia, Sierra Leone, Tanzania, and Zambia (Pinhey 1975, Razowski 2008).

Table 41: *Choristoneura occidentalis* host plants

Family	Genus/species	Common name
Fabaceae	*Acacia mearnsii* De Wild.	black wattle
Fabaceae	*Cajanus* Adans.	cajanus
Fabaceae	*Glycine max* (L.) Merr.	soybean
Fabaceae	*Vigna unguiculata* (L.) Walp.	blackeyed pea
Liliaceae	*Allium* L.	onion
Malvaceae	*Gossypium* L.	cotton
Ochnaceae	*Ochna thomasiana* Engl. & Gilg	Thomas' bird's-eye bush
Pinaceae	*Pinus patula* Schiede ex Schltdl. & Cham.	Mexican weeping pine
Rosaceae	*Rosa* L.	rose
Rubiaceae	*Coffea* L.	coffee
Rubiaceae	*Coffea arabica* L.	Arabian coffee
Rutaceae	*Citrus* L.	citrus
Rutaceae	*Citrus* X *aurantium* L. ssp. *aurantium*	sour orange
Stericulaceae	*Theobroma cacao* L.	cacao
Tiliaceae	*Corchorus* L.	corchorus

***Choristoneura rosaceana* (Harris)**

Nomenclature

Choristoneura rosaceana (Harris) (Tortricidae: Tortricinae: Archipini)

Common names: oblique banded leaf roller, rosaceous leaf roller

Synonyms: *gossypiana* (*Lozotaenia*), *vicariana* (*Teras*)

Adult Recognition

Forewing pattern is variable, but the majority of individuals have three fasciae that are variably expressed from costa to dorsum. The costal margin is sinuate, giving resting individuals a classic "bell-shaped" appearance. The distal half of the hindwing is white in males and yellowish in females. Males have a forewing costal fold. FWL: 7.5-11 mm (male); 11.5-14 mm (female).

Adults are similar to other species of *Choristoneura*, especially *C. fractivittana* and *C. parallela*, although these species are not nearly as common and males do not have a forewing costal fold. Many species of *Pandemis* have a similar wing pattern but male *Pandemis* do not have a forewing costal fold.

Larval Morphology

Last instar larvae are green to yellowish green and 25-30 mm long. The head is black to light brown. The prothoracic shield varies from completely shaded (black or brown), to partially shaded, to having no shading (body color). The thoracic legs are dark brown to black (Chapman and Lienk 1971).

Choristoneura rosaceana larvae are variable and can appear similar to larvae of many other tortricid species. This is one of the most common tortricid larvae encountered in North America.

Biology

The following life history information is summarized from Chapman and Lienk (1971), Chapman et al. (1968), and Powell (1964). *Choristoneura rosaceana* completes two annual generations over the majority of its range; however, only one generation is completed in northern areas and at higher elevations. Adults are present in late June though July and again in late August through September.

Eggs are laid in flat masses on the upper side of leaves. Egg masses may contain 200-900 individual eggs and are covered by the female with a waxy substance. First instar larvae hatch in 5-12 days and crawl to a protected location on the underside of leaves, in the buds, or under the calyx of a fruit. Young larvae may also disperse to other hosts by ballooning in the wind on a silk thread. Later instar larvae construct tubular shelters by webbing leaves. Larvae typically complete six instars, and second or third instars may overwinter in the fall and complete development in late May to mid-June. Pupation occurs in the final feeding shelter and lasts approximately 10-12 days.

Larvae of *C. rosaceana* are highly polyphagous and feed on the leaves or fruits of a wide variety of plants (Table 42). Preferred hosts appear to be woody plants and members of the family Rosaceae. Although this species is not currently considered a serious economic threat in North America, there have been populations of *C. rosaceana* reported as being resistant to insecticides (Carriere et al. 1996).

Table 42: *Choristoneura rosaceana* host plants

Family	**Genus/species**	**Common name**
Aceraceae	*Acer negundo* L.	boxelder
Aceraceae	*Acer rubrum* L.	red maple
Aceraceae	*Acer saccharinum* L.	silver maple
Aceraceae	*Acer* L.	maple
Anacardiaceae	*Pistacia* L.	pistache
Anacardiaceae	*Rhus coriaria* L.	Sicilian sumac
Asteraceae	*Ambrosia* L.	ragweed
Asteraceae	*Aster* L.	aster
Asteraceae	*Helianthus annuus* L.	common sunflower
Asteraceae	*Solidago* L.	goldenrod
Betulaceae	*Alnus incana* (L.) Moench	gray alder
Betulaceae	*Betula alleghaniensis* Britton	yellow birch
Betulaceae	*Betula papyrifera* Marshall	paper birch
Betulaceae	*Betula populifolia* Marsh.	gray birch
Betulaceae	*Betula* L.	birch
Betulaceae	*Corylus avellana* L.	common filbert
Betulaceae	*Corylus* L.	hazelnut
Betulaceae	*Ostrya virginiana* (Mill.) K. Koch	hophornbeam
Calycanthaceae	*Calycanthus occidentalis* Hook. & Arn.	western sweetshrub
Caprifoliaceae	*Lonicera periclymenum* L.	European honeysuckle
Caprifoliaceae	*Lonicera* L.	honeysuckle
Caprifoliaceae	*Symphoricarpos oreophilus* A. Gray var. *utahensis* (Rydb.) A. Nelson	Utah snowberry
Caprifoliaceae	*Viburnum lentago* L.	nannyberry
Caprifoliaceae	*Viburnum* L.	viburnum
Caryophyllaceae	*Dianthus caryophyllus* L.	carnation
Clusiaceae	*Hypericum* L.	St. Johnswort
Cornaceae	*Cornus florida* L.	flowering dogwood
Cornaceae	*Cornus racemosa* Lam.	gray dogwood
Cornaceae	*Cornus* L.	dogwood
Ericaceae	*Rhododendron* L.	rhododendron
Ericaceae	*Vaccinium corymbosum* L.	highbush blueberry
Ericaceae	*Vaccinium* L.	blueberry
Fabaceae	*Amorpha fruticosa* L.	desert false indigo

Table 42: *Choristoneura rosaceana* host plants

Family	**Genus/species**	**Common name**
Fabaceae	*Cercis canadensis* L.	eastern redbud
Fabaceae	*Phaseolus vulgaris* L.	kidney bean
Fabaceae	*Trifolium pratense* L.	red clover
Fagaceae	*Quercus agrifolia* Nee	California live oak
Fagaceae	*Quercus alba* L.	white oak
Fagaceae	*Quercus macrocarpa* Michx.	bur oak
Fagaceae	*Quercus rubra* L.	red oak
Fagaceae	*Quercus* L.	oak
Geraniaceae	*Geranium* L.	geranium
Hippocastanaceae	*Aesculus californica* (Spach) Nutt.	California buckeye
Oleaceae	*Fraxinus* L.	ash
Oleaceae	*Syringa vulgaris* L	common lilac
Oleaceae	*Syringa* L.	lilac
Pinaceae	*Abies balsamea* (L.) Mill.	balsam fir
Pinaceae	*Picea glauca* (Moench) Voss	white spruce
Pinaceae	*Pseudotsuga menziesii* (Mirb.) Franco	Douglas-fir
Pinaceae	*Tsuga heterophylla* (Raf.) Sarg.	western hemlock
Rhamnaceae	*Ceanothus integerrimus* Hook. & Arn.	deerbrush
Rhamnaceae	*Rhamnus* L.	buckthorn
Rosaceae	*Amelanchier stolonifera* Wiegand	running serviceberry
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Malus domestica* Borkh.	apple
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus avium* (L.) L.	sweet cherry
Rosaceae	*Prunus ilicifolia* (Nutt. ex Hook. & Arn.) D. Dietr.	hollyleaf cherry
Rosaceae	*Prunus pensylvanica* L. f.	pin cherry
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Prunus virginiana* L.	chokecherry
Rosaceae	*Prunus* L.	
Rosaceae	*Pyrus communis* L.	common pear
Rosaceae	*Pyrus* L.	pear
Rosaceae	*Rosa* L.	rose
Rosaceae	*Rubus flagellaris* Willd.	northern dewberry
Rosaceae	*Rubus idaeus* L.	American red raspberry
Rosaceae	*Rubus* L.	blackberry
Rosaceae	*Spiraea* L.	meadowsweet
Salicaceae	*Populus balsamifera* L.	balsam poplar
Salicaceae	*Populus* L.	cottonwood
Salicaceae	*Populus tremuloides* Michx.	quaking aspen
Salicaceae	*Salix lasiolepis* Benth.	arroyo willow

Table 42: *Choristoneura rosaceana* host plants

Family	**Genus/species**	**Common name**
Salicaceae	*Salix* L.	willow
Santalaceae	*Comandra umbellata* (L.) Nutt.	bastard toadflax
Tiliaceae	*Tilia americana* L.	American basswood
Tiliaceae	*Tilia* L.	basswood
Typhaceae	*Typha latifolia* L.	broadleaf cattail
Ulmaceae	*Celtis occidentalis* L.	common hackberry
Ulmaceae	*Ulmus americana* L.	American elm
Ulmaceae	*Ulmus* L.	elm
Verbenaceae	*Verbena* L.	vervain

Distribution

Choristoneura rosaceana is widely distributed throughout the continental United States and southern Canada.

***Clepsis* Guenée**

Overview

The genus *Clepsis* contains approximately 150 described species that are distributed throughout the Holarctic, Oriental, and Neotropical regions (Razowski 1979). Forewing patterns vary within the genus, but many species are brown and marked with a well-defined median fascia and dark costal spot. Male genitalia are characterized by a spatulate uncus and valvae that may be membranous at the apex or have a membranous apical lobe. Female genitalia are characterized by a cestum in the ductus bursae and a signum in the corpus bursae may be present or absent.

Species of economic importance include the Palearctic *C. spectrana*. In Europe, larvae of this species cause considerable damage to flowing and ornamental plants in greenhouses. The other six species treated here are common non-targets encountered during tortricid surveys. *Clepsis peritana* is one of the most commonly collected tortricids in the U.S.; it is similar to a number of other *Clepsis* species, but most can be separated by a combination of male and female morphological characters.

It is difficult to distinguish between different species of *Clepsis* using larval morphology. As such, most *Clepsis* larvae intercepted at U.S. ports of entry are not identified beyond the subfamily or genus level. One exception is larvae of *C. spectrana*, which are commonly intercepted at U.S. ports of entry on peppers (*Capsicum*) and cut flowers arriving from the Netherlands.

***Clepsis clemensiana* (Fernald)**

Nomenclature

Clepsis clemensiana (Fernald) (Tortricidae: Tortricinae: Archipini)

Synonyms: *nervosana* (*Tortrix*)

Adult Recognition

Forewings are straw yellow, narrow, and unmarked, although some females may have a faint reticulate pattern. The termen is steeply angled creating an apex that is nearly falcate in some individuals. Hindwings are primarily white. Males have a forewing costal fold. FWL: 9.5-11.5 mm.

Xenotemna pallorana is similar but can be distinguished from *C. clemensiana* by the lack of a forewing costal fold, a rounded termen on the forewing, and brownish or shaded hindwings. Genitalia of these two species are different and a dissection can be used to confirm identity.

Larval Morphology

Late instar larvae are approximately 10 mm in length. The head and prothoracic shield are yellowish brown. Pinacula are large and conspicuous. An anal comb is present (MacKay 1962).

Biology

Clepsis clemensiana completes one or two annual generations. Adults are present in June and July and again in September for the second generation. Larvae feed on leaves of the host plants. Pupation occurs in a silk tube. Grasses (Poaceae) are considered to be the primary host for Clepsis clemensiana, although the larvae have been reported feeding on plants in several other families (Table 43) (Powell 1964, Powell and Opler 2009).

Table 43: *Clepsis clemensiana* host plants

Family	**Genus/species**	**Common name**
Apocynaceae	*Apocynum* L.	dogbane
Asteraceae	*Aster* L.	aster
Asteraceae	*Solidago* L.	goldenrod
Asteraceae	*Symphyotrichum novae-angliae* (L.) G. L. Nesom	New England aster
Poaceae	*Phleum pratense* L.	timothy
Poaceae	*Triticum aestivum* L.	common wheat
Poaceae	[unspecified]	
Rosaceae	*Malus* Mill.	apple

Distribution

Clepsis clemensiana is widely distributed from northeastern North America west to British Columbia, south to northern Utah, Missouri, and Kentucky (Freeman 1958, Powell and Opler 2009).

Clepsis fucana **(Walsingham)**

Nomenclature

Clepsis fucana (Walsingham) (Tortricidae: Tortricinae: Archipini)

Synonyms: *busckana* (*Clepsis*), *victoriana* (*Cacoecia*)

Adult Recognition

Forewing color varies from dark brown to white. Most individuals have a well-defined median fascia and costal spot, although some may be nearly unmarked. Hindwing color is primarily white. Males lack a forewing costal fold. Males have a small membranous lobe on the apex of the valva. Females lack a signum in the corpus bursae. FWL: 6.0-10.5 mm.

Clepsis fucana, *C. penetralis*, *C. peritana*, and *C. virescana* are all similar in appearance. *Clepsis fucana* is generally larger than the other three species and is found only on the West Coast. *Clepsis penetralis* has only been confirmed from Colorado, Utah, and Vermont, although it may be misidentified in collections making its true distribution unknown. *Clepsis peritana* is the most commonly collected *Clepsis*, and it can be found throughout the United States and southern Canada. *Clepsis virescana* is larger and more boreal than *C. peritana* and it is not present in the Southeast. Table 44 lists diagnostic features that can be used to separate these four *Clepsis* species.

Table 44: Comparison of *Clepsis* species.

***Clepsis* species**	**FWL (mm)**	**Male FW costal fold**	**Male valva membranous lobe**	**Female ductus bursae**	**Female signum**	**Distribution**
fucana	6.5-10.5	absent	present as small lobe	straight	absent	West Coast
penetralis	6.0-7.5	absent	present as moderate lobe	weakly twisted	absent	Unknown; recorded from Colorado, Utah, and Vermont
peritana	4.5-7.5	absent	absent; entire apex is membranous	tightly coiled	absent	United States and southern Canada
virescana	6.0-9.0	present	present as small lobe	straight	present	United States and southern Canada; not present in the Southeast

Larval Morphology

According to MacKay (1962; as *C. busckana*), late instar larvae are approximately 13 mm long and are pale with large pinacula and long setae. The head is yellow. An anal comb is present.

Biology

This species completes two generations per year. Adults are most common in April to June and again in September and October. Larvae hollow out terminals of the host and feed on leaves that are webbed to the larval shelter (Powell 1964, Powell and Opler 2009). A list of documented host plants is provided in Table 45.

Table 45: *Clepsis fucana* host plants

Family	**Genus/species**	**Common name**
Asteraceae	*Anaphalis margaritacea* (L.) Benth.	western pearly everlasting
Asteraceae	*Anaphalis* DC.	pearly everlasting
Asteraceae	*Gnaphalium* L.	cudweed
Asteraceae	*Senecio jacobaea* L.	stinking willie
Boraginaceae	[unspecified]	
Cucurbitaceae	*Marah oreganus* (Torr. ex S. Watson) Howell	coastal manroot
Fagaceae	*Quercus agrifolia* Nee	California live oak
Hydrophyllaceae	*Phacelia californica* Cham.	California phacelia
Lamiaceae	*Stachys bullata* Benth.	California hedgenettle
Lamiaceae	*Stachys* L.	hedgenettle
Primulaceae	*Cyclamen* L.	cyclamen
Rhamnaceae	*Frangula californica* (Eschsch.) A. Gray	California buckthorn
Rosaceae	*Fragaria chiloensis* (L.) Mill.	beach strawberry
Rosaceae	*Horkelia californica* Cham. & Schltdl.	California horkelia
Rosaceae	*Rubus parviflorus* Nutt.	thimbleberry
Rosaceae	*Rubus* L.	blackberry
Scrophulariaceae	*Scrophularia californica* Cham. & Schltdl.	California figwort
Scrophulariaceae	*Scrophularia* L.	figwort
Urticaceae	*Urtica dioica* L. ssp. *holosericea* (Nutt.) Thorne	stinging nettle

Distribution

Clepsis fucana occurs along the West Coast of North America from British Columbia south to Monterey County, California (Powell and Opler 2009).

Clepsis melaleucanus **(Walker)**

Nomenclature

Clepsis melaleucanus (Walker) (Tortricidae: Tortricinae: Archipini)

Synonyms: *biustulana* (*Lozotaenia*), *invexana* (*Conchylis*), *semifuscana* (*Ptycholoma*)

Adult Recognition

Forewings are pale yellow with variable brown to yellowish-brown markings that may be suffused with bluish-gray scales. Most individuals have a large dark-brown patch in the area of the median fascia that is not continuous to the costa. Males have a forewing costal fold. FWL: 8.5-11.0 mm. The male uncus is short, broad at the base, and is not spatulate like that of many other *Clepsis* species. Females have a long cestum and a signum is present in the corpus bursae.

Well marked individuals are unlikely to be confused with any other Nearctic *Clepsis*. A genitalic dissection can be used to confirm identity.

Larval Morphology

Last instar larvae are 14-19 mm in length with a pale green abdomen. Pinacula are lighter than body color with dark setae. The head is yellowish brown, the prothoracic shield is pale green with dark shading on the posterolateral corners, and the thoracic legs are pale (Chapman and Lienk 1971, MacKay 1962).

Biology

Clepsis melaleucanus completes a single generation per year. Adults are present in June and July. Chapman and Lienk (1971) observed last instar larvae feeding on low growth of various plants in May and June. They speculate that overwintering occurs as a late instar larva in the ground cover. A list of documented host plants is provided in Table 46.

Table 46: *Clepsis melaleucanus* host plants

Family	**Genus/species**	**Common name**
Berberidaceae	*Caulophyllum* Michx.	cohosh
Betulaceae	*Alnus viridis* (Chaix) DC.	green alder
Betulaceae	*Alnus viridis* (Chaix) DC. ssp. *crispa* (Aiton) Turrill	mountain alder
Betulaceae	*Betula* L.	birch
Ericaceae	*Pyrola* L.	wintergreen
Hamamelidaceae	*Hamamelis* L.	witchhazel
Liliaceae	*Polygonatum* Mill.	Solomon's seal
Liliaceae	*Trillium* L.	trillium
Rosaceae	*Spiraea* L.	meadowsweet

Distribution

Clepsis melaleucanus is distributed from the northeastern United States and southern Canada west to Minnesota and Manitoba and south to North Carolina. It has been documented from England but is currently not established outside of North America (Bradley et al. 1973, Freeman 1958).

Clepsis peritana **(Clemens)**

Nomenclature

Clepsis peritana (Clemens) (Tortricidae: Tortricinae: Archipini)

Common names: garden tortrix, strawberry garden tortrix

Synonyms: *inconclusana* (*Dichelia*)

Adult Recognition

Forewing color is tan to brown. Males have a brown to dark-brown well-defined median fascia that is continuous from costa to dorsum and a dark-brown costal spot. Females have similar markings but the median fascia and costal spot are usually less distinct. Males lack a forewing costal fold. FWL: 4.5-7.5 mm.

Male genitalia are characterized by a single short, thin cornutus in the aedeagus and a membranous apex on the valva. Females have a ductus bursae that is spiraled or tightly coiled and lack a signum in the corpus bursae. Table 44 lists characters that can be used to separate *C. fucana*, *C. penetralis*, *C. peritana*, and *C. virescana*.

Larval Morphology

Last instar larvae are approximately 13-14 mm in length with a light green abdomen, but body color can vary depending on the host plant. The head and prothoracic shield are yellowish brown. An anal comb is present (MacKay 1962).

Biology

The following life history information is compiled from Freeman (1958), Powell (1964), and Powell and Opler (2009). *Clepsis peritana* completes several generations per year. The exact number of generations varies from 2-4 in the North to 6-7 in the South, and in some locations adults are present most of the year.

Eggs are deposited in small masses of approximately 10-20 individual eggs. Larvae live in silk tubes built on the surface of leaves and feed on dead or decaying leaf litter. Larvae will occasionally feed in the buds or fruits of living plants. On strawberry, larvae may cause damage to fruit in contact with the ground by webbing leaves to the fruit and chewing holes in the berries. In citrus groves, larvae feed on decaying leaves until population levels are high, at which point they may switch to feeding on fruit. Significant damage to citrus is caused only when fruit is close to the ground or has dropped from the tree.

Clepsis peritana is known in the economic literature as a pest of strawberry (*Fragaria* sp.) and citrus. Larvae prefer to feed on dead or decaying leaves and have also been reared from fungus. As its wide distribution would suggest, this species is probably a generalist feeder on a large number of plants. A list of documented host plants is provided in Table 47.

Table 47: *Clepsis peritana* host plants

Family	**Genus/species**	**Common name**
Asteraceae	*Chrysanthemum* L.	daisy
Asteraceae	*Cynara cardunculus* L.	cardoon
Asteraceae	*Senecio jacobaea* L.	stinking willie
Lamiaceae	*Stachys* L.	hedgenettle
Rosaceae	*Fragaria* L.	strawberry
Rutaceae	*Citrus* L.	citrus
Scrophulariaceae	*Scrophularia californica* Cham. & Schltdl.	California figwort
Solanaceae	*Solanum torvum* Sw.	turkey berry

Distribution

Clepsis peritana is widely distributed in southern Canada and the continental United States. This is one of the most common and widespread tortricid species in North America. It has been introduced into Denmark where it is reported to be established (Powell and Opler 2009, Razowski 2002).

***Clepsis persicana* (Fitch)**

Nomenclature

Clepsis persicana (Fitch) (Tortricidae: Tortricinae: Archipini)

Common names: white triangle tortrix, green needleworm

Synonyms: *blandana* (*Ditula*), *conigerana* (*Tortrix*), *fragariana* (*Lozotaenia*)

Subspecies: *C. persicana forbesi* (British Columbia)

Adult Recognition

Forewings are orange basally, becoming darker and purplish towards the pale termen. Most individuals have a well-defined white costal triangle, although this marking is reduced in some western phenotypes. Males have a long forewing costal fold that extends to nearly half the length of the costa. FWL: 8.5-10.5 mm (male); 10.0-11.0 mm (female). Forewing pattern is sufficient to distinguish adults of this species from other Nearctic Tortricidae.

Larval Morphology

Last instar larvae are approximately 14-18 mm long with a pale green abdomen. The head and prothoracic shield are yellowish brown and unmarked. An anal comb is present (MacKay 1962). Larvae of *Clepsis persicana* may be confused with a number of other tortricid larvae, including those of *Clepsis*, *Argyrotaenia*, *Choristoneura*, and *Epiphyas postvittana*.

Biology

The following life history information is compiled from Chapman and Lienk (1971), Freeman (1958), Powell (1964), and Powell and Opler (2009). *Clepsis persicana* completes a single generation per year. Adults are present from June to August.

Eggs are laid in overlapping patches on the upper surface of leaves or on smooth bark. Newly hatched larvae are incapable of constructing their own shelters and lower themselves on a silk thread searching for abandonded shelters of other tortricid species, such as *Spilonota ocellana*. If a suitable shelter is not found, larvae will descend to the ground and feed on cover plants for the remainder of the summer and autumn. Mid-instar larvae overwinter and complete development in the spring, where they may ascend trees or other woody plants. Larvae can cause damage to fruit by webbing leaves to the fruit or feeding in the calyx. Pupation occurs under bark or in fallen leaves near the base of a tree.

Larvae are general feeders and have been recorded from over 40 species of deciduous and coniferous trees (Table 48). Although Fitch described *C. persicana* from larvae he reared on peach (*Prunus persica*), this plant is not thought to be a primary host (Chapman and Lienk 1971).

Table 48: *Clepsis persicana* host plants

Family	Genus/species	Common name
Aceraceae	*Acer negundo* L.	boxelder
Aceraceae	*Acer* L.	maple
Apiaceae	*Osmorhiza berteroi* DC.	sweetcicely

Table 48: *Clepsis persicana* host plants

Family	**Genus/species**	**Common name**
Asteraceae	*Solidago* L.	goldenrod
Betulaceae	*Alnus incana* (L.) Moench	gray alder
Betulaceae	*Alnus viridis* (Chaix) DC.	green alder
Betulaceae	*Alnus viridis* (Chaix) DC. ssp. *crispa* (Aiton) Turrill	mountain alder
Betulaceae	*Alnus* Mill.	alder
Betulaceae	*Betula nana* L.	dwarf birch
Betulaceae	*Betula papyrifera* Marshall	paper birch
Betulaceae	*Betula* L.	birch
Betulaceae	*Corylus* L.	hazelnut
Cornaceae	*Cornus canadensis* L.	bunchberry dogwood
Ericaceae	*Rhododendron canadense* (L.) Torr.	rhodora
Ericaceae	*Vaccinium* L.	blueberry
Gentianaceae	*Frasera fastigiata* (Pursh) A. Heller	clustered green gentian
Gentianaceae	*Frasera* Walter	green gentian
Grossulariaceae	*Ribes* L.	currant
Liliaceae	*Maianthemum canadense* Desf.	Canada mayflower
Myrtaceae	*Comptonia peregrina* (L.) J. M. Coult.	sweet fern
Oleaceae	*Fraxinus* L.	ash
Pinaceae	*Abies balsamea* (L.) Mill.	balsam fir
Pinaceae	*Abies concolor* (Gord. & Glend.) Lindl. ex Hildebr.	white fir
Pinaceae	*Abies lasiocarpa* (Hook.) Nutt.	subalpine fir
Pinaceae	*Abies* Mill.	fir
Pinaceae	*Larix occidentalis* Nutt.	western larch
Pinaceae	*Larix* Mill.	larch
Pinaceae	*Picea engelmannii* Parry ex Engelm.	Engelmann spruce
Pinaceae	*Picea glauca* (Moench) Voss	white spruce
Pinaceae	*Picea* A. Dietr.	spruce
Pinaceae	*Pinus banksiana* Lamb.	jack pine
Pinaceae	*Pinus* L.	pine
Pinaceae	*Pseudotsuga menziesii* (Mirb.) Franco	Douglas-fir
Pinaceae	*Pseudostuga* Carriere	Douglas-fir
Rhamnaceae	*Ceanothus* L.	ceanothus
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Prunus virginiana* L.	chokecherry
Rosaceae	*Prunus* L.	
Rosaceae	*Rosa* L.	rose
Rosaceae	*Rubus* L.	blackberry
Salicaceae	*Populus balsamifera* L.	balsam poplar
Salicaceae	*Populus balsamifera* L. ssp. *trichocarpa* (Torr. & A. Gray ex Hook.) Brayshaw	black cottonwood

Table 48: *Clepsis persicana* host plants

Family	**Genus/species**	**Common name**
Salicaceae	*Populus tremuloides* Michx.	quaking aspen
Salicaceae	*Populus* L.	cottonwood
Salicaceae	*Salix* L.	willow
Ulmaceae	*Ulmus* L.	elm

Distribution

Clepsis persicana is distributed from the northeastern United States west across southern Canada and the northern United States to British Columbia, and south to California and the Rocky Mountains (Powell 1964, Powell and Opler 2009).

Clepsis spectrana **(Treitschke)**

Nomenclature

Clepsis spectrana (Treitschke) (Tortricidae: Tortricinae: Archipini)

Common names: cyclamen tortrix, straw-colored tortrix

Synonyms: *fuliginosana* (*Cacoecia costana* ab.), *intermedia* (*Tortrix costana* var.), *larseni* (*Cacoecia costana* ab.), *latiorana* (*Tortrix*), *liverana* (*Tortrix costana* var.), *vinculana* (Tortrix)

Adult Recognition

Forewings are pale yellow to tan with brown to dark-brown markings. Most individuals have a costal spot, a median fascia that is well defined from the costa to 1/3 the distance to the dorsum, and dark-brown to black irrorations. Forewing coloration and pattern can vary extensively, and immaculate and melanic forms have been described. Males have a forewing costal fold. FWL: 7.0-12.0 mm.

In the Nearctic, *C. spectrana* is most likely to be confused with *Clepsis fucana*. Males of the two species are easily separated by the costal fold that is present in *C. spectrana* and absent in *C. fucana*. Females can be separated by the presence of a signum in the corpus bursae of *C. spectrana*; *C. fucana* females lack a signum.

Males have been captured in pheromone traps using pheromones from several other species, including *Cacoecimorpha pronubana* and *Pandemis heparana* (E. LaGasa, pers. comm.).

Larval Morphology

Late instar larvae are 18-25 mm in length. The abdomen is brown to olive-green with conspicuous whitish pinacula and a pale subspiracular lateral line. The head and prothoracic shield are dark brown to black. An anal comb is present with 6-8 long teeth (Bradley et al. 1973, LaGasa 1999). Larvae of *C. spectrana* are commonly intercepted at U.S. ports of entry on peppers (*Capsicum*) and cut flowers arriving from the Netherlands.

Biology

The following life history information is summarized from Bradley et al. (1973), Dang et al. (1996), and LaGasa (1999). *Clepsis spectrana* completes 2-3 annual generations. Adults are present in May to July and again in August and September.

Females lay eggs in small masses on the host plant. Larvae feed in webbed leaves or flowers, and may cause considerable damage to foliage and developing fruits. Mid-instar larvae of the second or third generation overwinter until the following spring. Pupation occurs in the final larval shelter or in dead leaves.

Larvae are polyphagous and have been recorded feeding on plants in more than a dozen families. In Europe, *Clepsis spectrana* is an important pest of strawberry, blackberry, hops, and blackcurrant. It is also a serious pest of floriculture in greenhouses and has been recorded damaging a variety of flowering and ornamental plants (Razowski 1979, 2002).

Distribution

Clepsis spectrana is widely distributed across Europe, ranging as far east as Turkey and Kazakhstan. The earliest record in North America is a single specimen collected in British Columbia in 1950. It was "rediscovered" feeding on raspberry, currant, spruce, and cedar in the early 1990s in British Columbia, and the first U.S. record was collected in Washington in 1997. It is currently present in western Washington, although it has not yet reached pest status (Dang et al. 1996, LaGasa 1999, Razowski 2002).

Table 49: *Clepsis spectrana* host plants

Family	**Genus/species**	**Common name**
Aquifoliaceae	*Ilex* L.	holly
Asteraceae	*Artemisia maritima* L.	sea wormwood
Asteraceae	*Tripolium pannonicum* (Jacq.) Dobrocz.	sea aster
Begoniaceae	*Begonia* L.	begonia
Cannabaceae	*Humulus lupulus* L.	common hop
Caryophyllaceae	*Dianthus* L.	pink
Cyperaceae	*Scirpus* L.	bulrush
Cyperaceae	*Scirpus sylvanticus* L.	
Geraniaceae	*Pelargonium* L'Her. ex Aiton	geranium
Grossulariaceae	*Ribes* L.	currant
Iridaceae	*Iris* L.	iris
Iridaceae	*Iris pseudacorus* L.	paleyellow iris
Oleaceae	*Syringa* L.	lilac
Onagraceae	*Epilobium* L.	willowherb
Onagraceae	*Epilobium palustre* L.	marsh willowherb
Plumbaginaceae	*Limonium vulgare* Mill.	Mediterranean sea lavender
Poaceae	*Phragmites australis* (Cav.) Trin. ex Steud.	common reed
Primulaceae	*Cyclamen* L.	cyclamen
Rosaceae	*Filipendula ulmaria* (L.) Maxim.	queen of the meadow
Rosaceae	*Fragaria* L.	strawberry
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Potentilla* L.	cinquefoil
Rosaceae	*Pyrus* L.	pear
Rosaceae	*Rosa* L.	rose
Rosaceae	*Rubus* L.	blackberry
Rosaceae	*Spiraea* L.	meadowsweet
Solanaceae	*Capsicum annuum* L.	cayenne pepper
Solanaceae	*Capsicum* L.	pepper
Urticaceae	*Urtica dioica* L.	stinging nettle
Urticaceae	*Urtica* L.	nettle
Violaceae	*Viola* L.	violet
Vitaceae	*Vitis* L.	grape

***Clepsis virescana* (Clemens)**

Nomenclature

Clepsis virescana (Clemens) (Tortricidae: Tortricinae: Archipini)

Synonyms: *glaucana* (*Lozotaenia*), *sescuplana* (*Tortrix*)

Subspecies: *C. virescana salebrosa* (Mexico)

Adult Recognition

Forewing color varies from grayish brown to yellow brown. Wing pattern is uniform across most individuals, with a dark costal spot and poorly-defined median fascia. Males have a forewing costal fold. Male genitalia are distinctive with two long cornuti in the aedeagus and a small membranous lobe on the apex of the valva. Females have a long, thornlike signum in the corpus bursae. FWL: 6.0-9.0 mm. Table 44 lists characters that can be used to separate *C. fucana*, *C. penetralis*, *C. peritana*, and *C. virescana*.

Larval Morphology

Although the larva has not been described, it is assumed to be morphologically similar to larvae of other *Clepsis* species such as *C. peritana* and *C. fucana*.

Biology

Little has been reported on the life history of this species. Adult capture records indicate that *C. virescana* is bivoltine in central California. Powell (1964) reared larvae on fresh and decaying leaves of *Prunus* and *Rosa* and hypothesized that *C. virescana* may have feeding habits similar to those of *C. peritana*. In the laboratory, larvae completed development in September and October.

The only documented host is western chokecherry (Table 50), although Powell and Opler (2009) state that the larvae will feed on various soft-leaf plants and decaying leaves.

Table 50: *Clepsis virescana* host plants

Family	Genus/species	Common name
Rosaceae	*Prunus virginiana* L. var. *demissa* (Nutt.) Torr.	western chokecherry

Distribution

Clepsis virescana is widespread throughout the continental United States, southern Canada, and northern Mexico. It is generally found in more boreal conditions than *C. peritana* and is not present in the southeastern United States (Powell 1964, Powell and Opler 2009).

***Ctenopseustis obliquana* (Walker)**

Nomenclature

Ctenopseustis obliquana (Walker) (Tortricidae: Tortricinae: Archipini)

Common names: brown-headed leafroller

Synonyms: *charactana* (*Cacoecia*), *ropeana* (*Tortrix*), *spurcatana* (*Teras*), *transtrigana* (*Sciaphila*), *turbulentana* (*Sciaphila*)

Adult Recognition

Adults are brown to browish gray with a variable wing pattern. Most individuals have several dark markings along the costa, including a remnant of the median fascia. Males have a long forewing costal fold that extends to half the length of the wing. Hindwings are mottled in both males and females. FWL: 7.5-12.0 mm.

The brown-headed leafroller complex in New Zealand contains several species that are not easy to distinguish. Dugdale et al. (2005) recommend using DNA sequence data to separate members of this complex. Langhoff et al. (2009) provides DNA barcode data for distinguishing *Ctenopseustis* and *Planotortrix*.

Larval Morphology

Late instar larvae are approximately 20 mm in length with conspicuous pinacula. The head is dark brown to reddish brown and may be marked with faint red mottling. The prothoracic shield has dark shading on the lateral and posterior margins. Earlier instars have a black head and prothoracic shield (Dugdale et al. 2005, Kay 1979).

Biology

Ctenopseustis obliquana completes 4-6 overlapping generations per year. Adults are most common November-March. Females lay eggs in smooth masses composed of up to 150 individual eggs. Early instar larvae web together shoot tips or roll leaves. Later instars feed on leaves, buds, and fruit of the host plant. Larvae may cause economic damage by webbing leaves to fruit. Pupation occurs in the larval nest. Larval damage is nearly identical to that caused by *Epiphyas postvittana* and *Planotortrix excessana* (Dugdale et al. 2005).

The brown-headed leafroller complex is an important horticultural pest in New Zealand (Wearing et al. 1991). Larvae are highly polyphagous and have been recorded feeding on plants in more than 20 families (Table 51).

Table 51: *Ctenopseustis obliquana* host plants

Family	**Genus/species**	**Common name**
Actinidiaceae	*Actinidia arguta* (Siebold & Zucc.) Planch. ex Miq.	tara vine
Actinidiaceae	*Actinidia deliciosa* (A. Chev.) C. F. Liang & A. R. Ferguson	
Asteraceae	*Aster* L.	daisy
Verbenaceae	*Avicennia marina* (Forssk.) Vierh. var. *resinifera* (G. Forst.) Bakh.	gray mangrove
Berberidaceae	*Berberis* L.	barberry
Caprifoliaceae	*Lonicera* L.	honeysuckle
Cornocarpaceae	*Corynocarpus laevigata* Forst.	New Zealand laurel
Ebenaceae	*Diospyros kaki* L. f.	Japanese persimmon
Fabaceae	*Clianthus puniceus* (G. Don) Sol. ex Lindl.	Kaka beak
Salicaceae	*Dovyalis caffra* Warb.	kei apple
Escalloniaceae	*Escallonia* Mutis ex L.f.	
Grossulariaceae	*Ribes* L.	currant
Grossulariaceae	*Ribes nigrum* L.	European black currant
Myrtaceae	*Acca sellowiana* (O. Berg.) Burret	feijoa
Myrtaceae	*Syzygium smithii* (Poir.) Nied.	lilly pilly
Primulaceae	*Cyclamen* L.	cyclamen
Rosaceae	*Malus domestica* Borkh.	apple

Table 51: *Ctenopseustis obliquana* host plants

Family	**Genus/species**	**Common name**
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus armeniaca* L.	apricot
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Prunus* L.	cherry
Rosaceae	*Prunus* L.	plum
Rosaceae	*Rosa* L.	rose
Rosaceae	*Rubus* L.	cranberry
Rubiaceae	*Coprosma rotundifolia* A. Cunn.	
Rutaceae	*Citrus* L.	citrus
Scrophulariaceae	*Veronica* L.	speedwell
Theaceae	*Camellia japonica* L.	
Vitaceae	*Vitis vinifera* L.	wine grape

Distribution

Ctenopseustis obliquana is a native of New Zealand (Dugdale et al. 2005, Wearing et al. 1991). Reports of it being introduced into Hawaii could not be confirmed.

***Epichoristodes acerbella* (Walker)**

Nomenclature

Epichoristodes acerbella (Walker) (Tortricidae: Tortricinae: Archipini)

Common names: South African carnation tortrix, carnation worm, pear leafroller

Synonyms: *galeata* (*Epichorista*), *iocoma* (*Tortrix*), *ionephela* (*Proselena*)

Adult Recognition

Adults are yellow to reddish brown and generally unmarked. Some individuals may have a darker reddish-brown patch on the dorum near the tornus. Males lack a forewing costal fold. FWL: 6.5-10.0 mm.

Adults may appear similar to other yellow-brown, unmarked archipines. The genus *Epichoristodes* contains 15 described species (Baixeras et al. 2008); *E. acerbella* is the only species distributed outside of Africa.

Larval Morphology

Late instar larvae are green to yellowish green. The head and prothoracic shield are yellow brown and both may have extensive posterior shading or mottling. An anal comb is present with 6-9 teeth (Timm et al. 2008).

Other carnation/greenhouse pests present in Africa and Europe include *Cacoecimorpha proubana*. Larvae of *C. pronubana* have a set of distinctive dark-brown to black markings of the posterolateral corners of the prothoracic shield.

Biology

In southern Europe, *E. acerbella* completes four generations per year, with adults present May-September. More generations are possible in a greenhouse environment. Females lay eggs on leaves in elongate masses of approximately 25 individual eggs. Larvae feed in rolled leaves, in flower buds, and on flower petals. Larvae may also feed on the surface of fruit or tunnel into stems. Pupation occurs in the final larval feeding site (Glavendekic 2006, Van de Vrie 1991).

Larvae of *E. acerbella* are polyphagous and have been reported feeding on plants in more than a dozen families (Table 52). This species is recognized as an important pest of carnations and other floricultural crops (Van de Vrie 1991).

Table 52: *Epichoristodes acerbella* host plants

Family	**Genus/species**	**Common name**
Asteraceae	*Aster* L.	aster
Asteraceae	*Chrysanthemum* L.	daisy
Asteraceae	*Erigeron* L.	fleabane
Asteraceae	*Gerbera* J. F. Gmel.	Transvaal daisy
Asteraceae	*Sonchus* L.	sowthistle
Caryophyllaceae	[unspecified]	
Caryophyllaceae	*Dianthus caryophyllus* L.	carnation
Caryophyllaceae	*Dianthus* L.	pink
Cucurbitaceae	*Cucurbita* L.	gourd
Fabaceae	*Medicago* L.	alfalfa

Table 52: *Epichoristodes acerbella* host plants

Family	Genus/species	Common name
Liliaceae	*Ornithogalum* L.	star of Bethlehem
Liliaceae	*Ornithogalum thyrsoides* Jacq.	chinkerinchee
Myrtaceae	*Eucalyptus saligna* Sm.	Sydney bluegum
Polygonaceae	*Rumex* L.	dock
Rhamnaceae	*Rhamnus* L.	buckthorn
Rosaceae	*Fragaria* L.	strawberry
Rosaceae	*Prunus* L.	
Rosaceae	*Pyrus* L.	pear
Rosaceae	*Rosa* L.	rose
Rubiaceae	*Coffea* L.	coffee
Solanaceae	*Capsicum annuum* L.	cayenne pepper
Valerianaceae	*Centranthus ruber* (L.) DC.	red valerian

Distribution

A native of South Africa, *E. acerbella* is widely distributed in eastern and southern Africa. It was first reported in Europe (Italy) in the late 1960s, and it has been subsequently introduced to the following countries: Bulgaria, Croatia, France, Italy, Romania, Serbia, Slovenia, and Spain (Bradley et al. 1973, Glavendekic 2006, Timm et al. 2008).

***Epiphyas postvittana* (Walker)**

Refer to Chapter 3 for information on *Epiphyas postvittana*.

***Isotenes miserana* (Walker)**

Nomenclature

Isotenes miserana (Walker) (Tortricidae: Tortricinae: Archipini)

Common names: orange fruitborer

Synonyms: *absumptana* (*Teras*)

Adult Recognition

Forewings are light brownish gray with brown or gray fasciate markings. Males have a forewing costal fold. Male genitalia are characterized by valvae that appear to be twisted dorsally. Female genitalia are characterized by a twisted ductus bursae and a single daggerlike signum in the corpus bursae. FWL: 8.0-11.0 mm.

Adults may appear similar to other gray-brown archipines with a fasciate forewing pattern. A genitalic dissection can be used to confirm identity.

Larval Morphology

Late instar larvae are pale cream with a reddish-brown line running along the SD setae and faint reddish-brown markings on each segment dorsal to the pale D setal pinacula. The head and thoracic legs are dark brown. The prothoracic shield is dark brown posteriorly, becoming pale cream anteriorly (Dugdale et al. 2005).

Biology

Isotenes miserana completes several generations per year. Generations are overlapping and adults may be present year-round in some locations. Females lay eggs in masses and surround them with "fences" constructed from specialized scales on the abdomen. Larvae tunnel into fruit and feed just below the skin; they may also feed on buds and young leaves. Pupation occurs in the larval feeding shelter. Larvae of *I. miserana* are polyphagous and have been recorded feeding on citrus, camellia, macadamia, peach, rose, and a variety of other plants in at least 14 families (Table 53) (Dugdale et al. 2005).

Table 53: *Isotenes miserana* host plants

Family	**Genus/species**	**Common name**
Annonaceae	*Melodorum* Lour.	
Apocynaceae	*Nerium* L.	oleander
Asclepiadaceae	*Marsdenia* sp.	
Fabaceae	*Lupinus* L.	lupine
Fabaceae	*Pultenaea* Sm.	

Table 53: *Isotenes miserana* host plants

Family	**Genus/species**	**Common name**
Lamiaceae	*Premna* L.	premna
Lauraceae	*Cinnamomum* Schaeff.	cinnamon
Lauraceae	*Litsea* Lam.	
Meliaceae	*Toona* (Endl.) M. Roem.	
Moraceae	*Ficus benjamina* L.	weeping fig
Moraceae	*Ficus* L.	fig
Moraceae	*Morus* L.	mulberry
Myrsinaceae	*Aegiceras corniculatum* (L.) Blanco	black mangrove
Pinaceae	*Pinus merkusii* Jungh. & de Vriese	Sumatran pine
Proteaceae	*Macadamia* F. Muell.	macadamia
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus* L.	[unspecified]
Rosaceae	*Rosa* L.	rose
Rutaceae	*Citrus* L.	citrus
Theaceae	*Camellia* L.	camellia
Verbenaceae	*Lantana* L.	lantana
Verbenaceae	*Vitex* L.	vitex

Distribution

Isotenes miserana is a native of Australia (Northern Territory and Queensland) that has spread to New Zealand and South Asia (Diakonoff 1939, Dugdale et al. 2005).

***Pandemis cerasana* (Hübner)**

Nomenclature

Pandemis cerasana (Hübner) (Tortricidae: Tortricinae: Archipini)

Common names: barred fruit-tree tortrix

Synonyms: *balticola* (ab.), *grossulariana* (*Lozotaenia*), *obscura* (ab.), *ribeana* (*Tortrix*), *transiens* (var.)

Adult Recognition

Adults are straw to light brown with fasciate markings and grayish-brown hindwings. FWL: 8.0-12.0 mm. *Pandemis cerasana* can be separated from other *Pandemis* listed here by the grayish-brown hindwings and dark scales on the second abdominal sternite in the male.

Six species of *Pandemis* occur in the Nearctic. Four, *P. canadana*, *P. lamprosana*, *P. limitata*, and *P. pyrusana*, are native, while two, *P. cerasana* and *P. heparana*, have been introduced from the Palearctic. All species treated here have a straw (yellow) to brown forewing with brown to dark-brown markings consisting of a patch on the costa below the apex, a median band that extends from costa to dorsum, and a basal band. The two bands (or fasciae) may be lined with light or dark scales in some individuals, creating the appearance of three lines running vertically across the wing. Males have a distinctive notch at the base of the antennae and modified dark scales on the ventral surface of abdominal segments 2-3 (this character is absent in *P. lamprosana* and *P. heparana*). Males lack a forewing costal fold.

Species identification within the group is difficult. *Pandemis lamprosana*, *P. cerasana*, and *P. heparana*, can be identified by wing color and male genitalia. The other three species, *P. canadana*, *P. limitata*, and *P. pyrusana*, exhibit variable wing patterns, share identical genitalia, and cannot be reliably separated where their distributions overlap. Dombroskie (2011) provides a combination of characters and geographic distribution that can be used to identify many *Pandemis* individuals collected in the U.S.; this information is summarized in Table 54.

Larval Morphology

Late instar larvae are entirely green and unmarked with moderately large pinacula and long setae. Head and prothoracic shield are light green to yellowish green with black posterolateral markings. The spiracles on the prothorax and eighth abdominal segment are 2-3 times the diameter of other abdominal spiracles. A well developed anal comb is present with 6-8 teeth (MacKay 1962, Mutuura 1980).

Table 54: Comparison of *Pandemis* species.

***Pandemis* species**	**Forewing color**	**Hindwing color**	**Sex scales on male 2nd abd. segment**	**Distribution**
canadana	medium to dark brown	all gray	present	Maine, Colorado, Wyoming, Southern Canada
cerasana	straw to light brown	grayish brown	present	Pacific Northwest, British Columbia; Europe and Asia
heparana	medium brown	light to medium grayish brown	absent	Pacific Northwest, British Columbia; Europe and Asia
lamprosana	tan to light brown	white to light gray	absent	Northeastern U.S., southern Quebec and Ontario
limitata	straw to medium brown	gray and white	present	Eastern U.S. and southern Canada; generally absent in the U.S. west of the Rocky Mtns.
pyrusana	straw to medium brown	all white	present	Rocky Mtns. west to California, southern Alberta and British Columbia

MacKay (1962) examined several species of Nearctic *Pandemis* and could find no species-specific larval characters. Diagnostic characters for the genus include: SD2 on A1-8 on same pinaculum as SD1; L1 and L2 anterior to spiracle on A2-8; SV group on A1, 2, 7, 8, 9 usually 3:3:3:2:2; D2s on A8 as far apart as D1s; D1 on A9 on its own pinaculum; anal setae very long; anal comb with 6-8 teeth.

Biology

The following life history information is compiled from Bradley et al. (1973), Mutuura (1980), and Razowski (2002). In Europe, *P. cerasana* completes one or two generations per year. Adults are present June-July for the first generation and August-September for the second generation.

Females deposit eggs in masses on the upper surface of leaves or on branches. Some eggs hatch in late summer; others overwinter and larvae emerge the following spring. Larvae that emerge before winter construct a hibernaculum in the second or third instar. Larvae feed on leaves in the spring and pupation occurs in the final larval feeding site. Larvae of *P. cerasana* have been recorded feeding on plants in 15 families (Table 55). This species is an occasional orchard pest.

Distribution

Pandemis cerasana is widely distributed in the Palearctic from Western Europe to Asia. In North America it has been introduced to the Pacific Northwest. The first North American records are from British Columbia in 1965 (Mutuura 1980, Razowski 2002).

Table 55: *Pandemis cerasana* host plants

Family	**Genus/species**	**Common name**
Aceraceae	*Acer* L.	maple
Balsaminaceae	*Impatiens* L.	touch-me-not
Berberidaceae	*Berberis* L.	barberry
Betulaceae	*Alnus glutinosa* (L.) Gaertn.	European alder
Betulaceae	*Betula* L.	birch
Betulaceae	*Corylus avellana* L.	common filbert
Caprifoliaceae	*Lonicera periclymenum* L.	European honeysuckle
Ericaceae	*Vaccinium* L.	blueberry
Fagaceae	*Quercus* L.	oak
Fagaceae	*Quercus robur* L.	English oak
Grossulariaceae	*Ribes* L.	currant
Oleaceae	*Fraxinus* L.	ash
Primulaceae	*Lysimachia* L.	yellow loosestrife
Rhamnaceae	*Rhamnus* L.	buckthorn
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Geum* L.	avens
Rosaceae	*Malus domestica* Borkh.	apple
Rosaceae	*Malus sylvestris* (L.) Mill.	European crab apple
Rosaceae	*Prunus* L.	
Rosaceae	*Pyrus* L.	pear
Rosaceae	*Rosa* L.	rose
Rosaceae	*Sorbus* L.	mountain ash
Rosaceae	*Sorbus aucuparia* L.	European mountain ash
Salicaceae	*Salix cinerea* L.	large gray willow
Salicaceae	*Salix* L.	willow
Tiliaceae	*Tilia* L.	basswood
Ulmaceae	*Ulmus* L.	elm

***Pandemis heparana* (Denis & Schiffermüller)**

Nomenclature

Pandemis heparana (Denis & Schiffermüller) (Tortricidae: Tortricinae: Archipini)

Common names: dark fruit-tree tortrix

Synonyms: *cappana* (*Tortrix*), *cappata* (*Tortrix*), *carpiniana* (*Tortrix*), *fasciana* (*Pyralis*), *heperana* (*Tortrix*), *padana* (*Tortrix*), *pasquayana* (*Tortrix*), *rubrana* (*Tortrix*), *subclarana* (var.), *vulpisana* (*Lozotaenia*)

Adult Recognition

Adults are medium brown with fasciate markings and light to medium grayish-brown hindwings. *Pandemis heparana* can be separated from other *Pandemis* listed here by the grayish-brown hindwings and the lack of dark scales on the second abdominal sternite in the male. FWL: 8.0-12.0 mm.

Six species of *Pandemis* occur in the Nearctic. Four, *P. canadana*, *P. lamprosana*, *P. limitata*, and *P. pyrusana*, are native, while two, *P. cerasana* and *P. heparana*, have been introduced from the Palearctic. Characters for identifying these species are listed under the description of *P. cerasana* and in Table 54.

Larval Morphology

Late instar larvae are entirely green and unmarked with moderately large pinacula and long setae. The head is variably colored from light green to yellowish brown with black lateral markings. The prothoracic shield is green or yellowish brown with black posterolateral markings. A well developed anal comb is present with 6-8 teeth (MacKay 1962, Mutuura 1980).

MacKay (1962) examined several species of Nearctic *Pandemis* and could find no species-specific larval characters. Diagnostic characters for the genus are listed under the description of *P. cerasana*.

Biology

The following life history information is compiled from Bradley et al. (1973), Mutuura (1980), and Razowski (2002). In Europe, *P. heparana* completes one or two generations per year. Adults are present June-July for the first generation and August-September for the second generation.

Females deposit eggs in masses on the upper surface of leaves. Larvae feed on leaves and construct a hibernaculum in the second or third instar in which to overwinter. Larvae resume feeding in the spring. Pupation occurs in the final larval feeding site. Larvae of *P. heparana* are polyphagous and have been recorded feeding on plants in more than 20 families (Table 56). This species is considered an occasional orchard pest in the Palearctic.

Table 56: *Pandemis heparana* host plants

Family	**Genus/species**	**Common name**
Aceraceae	*Acer* L.	maple
Asteraceae	*Achillea* L.	yarrow
Asteraceae	*Arctium* L.	burdock
Asteraceae	*Arctium lappa* L.	greater burdock
Asteraceae	*Artemisia montana* Pamp.	
Asteraceae	*Erigeron annuus* (L.) Pers.	eastern daisy
Betulaceae	*Alnus glutinosa* (L.) Gaertn.	European alder
Betulaceae	*Alnus japonica* (Thunb.) Steud.	Japanese alder
Betulaceae	*Alnus* Mill.	alder
Betulaceae	*Betula* L.	birch
Betulaceae	*Betula platyphylla* Sukaczev	Asian white birch
Betulaceae	*Carpinus* L.	hornbeam
Betulaceae	*Corylus* L.	hazelnut
Boraginaceae	*Anchusa* L.	bugloss
Cannabaceae	*Humulus* L.	hop
Caprifoliaceae	*Lonicera periclymenum* L.	European honeysuckle
Chenopodiaceae	*Beta vulgaris* L.	common beet
Cornaceae	*Cornus controversa* Hemsl. ex Prain	giant dogwood
Cornaceae	*Cornus* L.	dogwood
Elaeagnaceae	*Elaeagnus* L.	oleaster
Ericaceae	*Rhododendron mucronulatum* Turcz.	
Ericaceae	*Vaccinium* L.	blueberry

Table 56: *Pandemis heparana* host plants

Family	**Genus/species**	**Common name**
Ericaceae	*Vaccinium uliginosum* L.	bog blueberry
Ericaceae	*Vaccinium vitis-idaea* L.	lingonberry
Fabaceae	*Phaseolus* L.	bean
Fabaceae	*Trifolium repens* L.	white clover
Fagaceae	*Castanea crenata* Siebold & Zucc.	Japanese chestnut
Fagaceae	*Castanea* Mill.	chestnut
Fagaceae	*Fagus* L.	beech
Fagaceae	*Quercus cerris* L.	European turkey oak
Fagaceae	*Quercus dentata* Thunb.	Daimyo oak
Fagaceae	*Quercus* L.	oak
Fagaceae	*Quercus robur* L.	English oak
Grossulariaceae	*Ribes* L.	currant
Juglandaceae	*Juglans* L.	walnut
Linaceae	*Linum usitatissimum* L.	common flax
Moraceae	*Morus* L.	mulberry
Myricaceae	*Myrica gale* L.	sweetgale
Oleaceae	*Fraxinus* L.	ash
Oleaceae	*Ligustrum* L.	privet
Oleaceae	*Ligustrum yesoense* Nakai	
Oleaceae	*Syringa* L.	lilac
Oleaceae	*Syringa vulgaris* L.	common lilac
Polygonaceae	*Rumex obtusifolius* L.	bitter dock
Primulaceae	*Lysimachia* L.	yellow loosestrife
Rhamnaceae	*Rhamnus* L.	buckthorn
Rosaceae	*Crataegus chlorosarca* Maxim.	
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Cydonia oblonga* Mill.	quince
Rosaceae	*Fragaria* L.	strawberry
Rosaceae	*Malus domestica* Borkh.	apple
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Malus sylvestris* (L.) Mill.	European crab apple
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus armeniaca* L.	apricot
Rosaceae	*Prunus avium* (L.) L.	sweet cherry
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Prunus salicina* Lindl.	Japanese plum
Rosaceae	*Prunus serrulata* Lindl.	Japanese flowering cherry
Rosaceae	*Prunus* X *yedoensis* Matsum. (pro sp.) [*subhirtella* X *speciosa*]	
Rosaceae	*Prunus* L.	

Table 56: *Pandemis heparana* host plants

Family	**Genus/species**	**Common name**
Rosaceae	*Pyrus* L.	pear
Rosaceae	*Pyrus ussuriensis* Maxim.	
Rosaceae	*Rosa* L.	rose
Rosaceae	*Rubus* L.	blackberry
Rosaceae	*Sorbus* L.	mountain ash
Rutaceae	*Phellodendron amurense* Rupr.	Amur corktree
Salicaceae	*Populus* L.	cottonwood
Salicaceae	*Salix caprea* L.	goat willow
Salicaceae	*Salix cinerea* L.	large gray willow
Salicaceae	*Salix* L.	willow
Tiliaceae	*Tilia* L.	basswood
Ulmaceae	*Ulmus davidiana* Planch.	Japanese elm
Ulmaceae	*Ulmus* L.	elm

Distribution

Pandemis heparana is widely distributed in the Palearctic from Western Europe to Asia. In North America it has been introduced to the Pacific Northwest. The first North American records are from British Columbia in 1978 (Mutuura 1980, Razowski 2002).

***Pandemis lamprosana* (Robinson)**

Nomenclature

Pandemis lamprosana (Robinson) (Tortricidae: Tortricinae: Archipini)

Adult Recognition

Adults are light brown with fasciate markings and white to light gray hindwings. FWL: 8.0-10.5 mm (male); 9.5-12.0 mm (female). *Pandemis lamprosana* can be separated from other *Pandemis* listed here by the white to light gray hindwings and the lack of dark scales on the second abdominal sternite in the male.

Six species of *Pandemis* occur in the Nearctic. Four, *P. canadana*, *P. lamprosana*, *P. limitata*, and *P. pyrusana*, are native, while two, *P. cerasana* and *P. heparana*, have been introduced from the Palearctic. Characters for identifying these species are listed under the description of *P. cerasana* and in Table 54.

Larval Morphology

Late instar larvae are approximately 20 mm in length and are entirely green and unmarked with moderately large pinacula and long setae. Earlier instars may have a dark lateral mark on each side of the prothoracic shield. A well developed anal comb is present with 6-8 teeth.

MacKay (1962) examined several species of Nearctic *Pandemis* and could find no species-specific larval characters. Diagnostic characters for the genus are listed under the description of *P. cerasana*.

Biology

Pandemis lamprosana completes a single generation per year. Adults are present in late June and July.

Females lay eggs on the upper surface of leaves in large masses. Larvae feed on leaves and have not been observed feeding on fruit. Third instar larvae construct a hibernaculum in a protected site and overwinter until the following spring. Feeding resumes in the spring, and larvae do not complete development until June. Pupation occurs in the final larval feeding site (Chapman and Lienk 1971).

Larvae have been recorded feeding on a variety of deciduous trees (Table 57). Reports of larvae feeding on Urticaceae in Illinois are questionable.

Distribution

Pandemis lamprosana is found in the northeastern U.S. and southern Canada (Quebec and Ontario) (Chapman and Lienk 1971).

Table 57: *Pandemis lamprosana* host plants

Family	**Genus/species**	**Common name**
Aceraceae	*Acer rubrum* L.	red maple
Aceraceae	*Acer saccharinum* L.	silver maple
Aceraceae	*Acer spicatum* Lam.	mountain maple
Betulaceae	*Betula alleghaniensis* Britton	yellow birch
Betulaceae	*Betula papyrifera* Marshall	paper birch
Betulaceae	*Ostrya virginiana* (Mill.) K. Koch	hophornbeam
Fabaceae	*Gleditsia triacanthos* L.	honeylocust
Fagaceae	*Fagus* L.	beech
Fagaceae	*Quercus* L.	oak
Fagaceae	*Quercus rubra* L.	red oak
Hamamelidaceae	*Hamamelis* L.	witchhazel
Lauraceae	*Sassafras* Nees & Eberm.	sassafras
Oleaceae	*Fraxinus americana* L.	white ash
Oleaceae	*Fraxinus* L.	ash
Platanaceae	*Platanus* L.	sycamore
Rosaceae	*Prunus virginiana* L.	chokecherry
Salicaceae	*Populus tremuloides* Michx.	quaking aspen
Tiliaceae	*Tilia americana* L.	American basswood
Ulmaceae	*Ulmus americana* L.	American elm
Ulmaceae	*Ulmus rubra* Muhl.	slippery elm

Pandemis limitata **(Robinson)**

Nomenclature

Pandemis limitata (Robinson) (Tortricidae: Tortricinae: Archipini)

Common names: three-lined leaf roller

Synonyms: *limitana* (*Pandemis*)

Adult Recognition

Adults are brown with fasciate markings and gray and white hindwings. FWL: 7.0-9.5 mm (male); 9.0-12.0 mm (female). *Pandemis limitata* is similar to *P. pyrusana* and *P. canadana*, and the three species are not easily separated. A combination of geographic distribution and wing color can assist in identification. In the Pacific Northwest and central Rocky Mountains all three species are present and reliable species-level identification is difficult or impossible.

Characters for identifying the various species of *Pandemis* treated here are listed under the description of *P. cerasana* and in Table 54.

Larval Morphology

Late instar larvae are approximately 20 mm in length and are entirely green and unmarked with moderately large pinacula and long setae. Earlier instars may have a dark lateral mark on each side of the prothoracic shield. A well developed anal comb is present with 6-8 teeth (MacKay 1962).

MacKay (1962) examined several species of Nearctic *Pandemis* and could find no species-specific larval characters. Diagnostic characters for the genus are listed under the description of *P. cerasana*.

Biology

Pandemis limitata completes one or two generations over most of its range. Adults are present June through August.

Females lay eggs on the upper surface of leaves in large masses that contain between 60-182 eggs. Larvae feed on terminal leaf growth and occasionally on fruit. Those of the first generation complete development in late July or early August. Pupation occurs in the final larval feeding site and adults emerge in approximately 10 days. Third instar larvae of the second generation construct a hibernaculum in a protected site on small diameter growth and overwinter until the following spring, when they resume feeding on leaves and young fruits (Freeman 1958, Mutuura 1980, Powell and Opler 2009).

Larvae of *P. limitata* have been recorded feeding on a variety of deciduous woody plants (Table 58). This species is considered a minor pest of apple in some regions.

Table 58: *Pandemis limitata* host plants

Family	**Genus/species**	**Common name**
Aceraceae	*Acer negundo* L.	boxelder
Aceraceae	*Acer saccharinum* L.	silver maple
Betulaceae	*Alnus incana* (L.) Moench	gray alder
Betulaceae	*Alnus* Mill.	alder
Betulaceae	*Alnus rubra* Bong.	red alder
Betulaceae	*Betula* L.	birch
Betulaceae	*Betula papyrifera* Marshall	paper birch
Betulaceae	*Corylus americana* Walter	American hazelnut
Betulaceae	*Corylus* L.	hazelnut
Caprifoliaceae	*Viburnum* L.	viburnum
Celastraceae	*Euonymus atropurpureus* Jacq.	burningbush
Cornaceae	*Cornus racemosa* Lam.	gray dogwood
Ericaceae	*Vaccinium* L.	blueberry
Fabaceae	*Amorpha fruticosa* L.	desert false indigo
Fabaceae	*Trifolium* L.	clover
Fagaceae	*Castanea* Mill.	chestnut
Fagaceae	*Quercus alba* L.	white oak
Fagaceae	*Quercus* L.	oak
Fagaceae	*Quercus macrocarpa* Michx.	bur oak
Myricaceae	*Myrica gale* L.	sweetgale
Osmundaceae	*Osmunda* L.	osmunda
Rosaceae	*Malus domestica* Borkh.	apple
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Malus sylvestris* (L.) Mill.	European crab apple
Rosaceae	*Prunus avium* (L.) L.	sweet cherry
Rosaceae	*Prunus* L.	
Rosaceae	*Prunus virginiana* L.	chokecherry
Rosaceae	*Sorbus* L.	mountain ash
Salicaceae	*Populus alba* L.	white poplar
Salicaceae	*Populus balsamifera* L.	balsam poplar
Salicaceae	*Populus* L.	cottonwood
Salicaceae	*Populus tremuloides* Michx.	quaking aspen
Salicaceae	*Salix* L.	willow
Tiliaceae	*Tilia americana* L.	American basswood
Ulmaceae	*Ulmus americana* L.	American elm
Ulmaceae	*Ulmus* L.	elm
Ulmaceae	*Ulmus rubra* Muhl.	slippery elm

Distribution

Pandemis limitata ranges in southern Canada from Nova Scotia to British Columbia and in the U.S. from the East Coast west to the Rocky Mountains and Arizona. It is generally absent in the U.S. west of the Rockies. There is a single record from Durango, Mexico (Dombroskie 2011, Powell and Opler 2009).

Pandemis pyrusana **(Kearfott)**

Nomenclature

Pandemis pyrusana (Kearfott) (Tortricidae: Tortricinae: Archipini)

Common names: apple pandemis

Synonyms: *pyrana* (*Pandemis*)

Adult Recognition

Adults are brown with fasciate markings and white hindwings. FWL: 8.0-12.5 mm (male); 9.5-14.0 mm (female). *Pandemis pyrusana* is similar to *P. limitata* and *P. canadana*, and the three species are not easily separated. A combination of geographic distribution and wing color can assist in identification (see below). In the Pacific Northwest and central Rocky Mountains all three species are present and reliable species-level identification is difficult or impossible. Characters for identifying the *Pandemis* species treated here are listed under the description of *P. cerasana* and in Table 54.

Larval Morphology

Late instar larvae are approximately 20 mm in length and are entirely green and unmarked with moderately large pinacula and long setae. Earlier instars may have a dark lateral mark on each side of the prothoracic shield. A well developed anal comb is present with 6-8 teeth.

MacKay (1962) examined several species of Nearctic *Pandemis* and could find no species-specific larval characters. Diagnostic characters for the genus are listed under the description of *P. cerasana*.

Biology

Pandemis pyrusana completes one or two generations per year. Adults are present in the costal areas of California from May to July and again in September to November. Females deposit eggs in masses on the upper surfaces of leaves and on fruit. Early instar larvae of the first (summer) generation feed under a shelter constructed along the mid-rib of a leaf. Later instars feed on foliage in various locations on the host and may cause economic damage by feeding between clusters of fruit. Larvae of the second generation overwinter in shelters at the base of trees or under bark. In the spring they begin feeding on terminal leaves and pupate in April or May inside a folded leaf (Freeman 1958, Newcomer and Carlson 1952). A list of documented host plants is presented in Table 59.

Table 59: *Pandemis pyrusana* host plants

Family	**Genus/species**	**Common name**
Betulaceae	*Alnus* Mill.	alder
Betulaceae	*Betula occidentalis* Hook.	water birch
Caprifoliaceae	*Lonicera* L.	honeysuckle
Cornaceae	*Cornus sericea* L.	redosier dogwood
Grossulariaceae	*Ribes* L.	currant
Rhamnaceae	*Ceanothus cuneatus* (Hook.) Nutt.	buckbrush
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Prunus avium* (L.) L.	sweet cherry
Rosaceae	*Prunus* L.	
Rosaceae	*Prunus virginiana* L.	chokecherry
Rosaceae	*Pyrus* L.	pear
Rosaceae	*Rosa* L.	rose
Salicaceae	*Populus tremuloides* Michx.	quaking aspen
Salicaceae	*Salix* L.	willow
Salicaceae	*Salix lasiolepis* Benth.	arroyo willow
Salicaceae	*Salix sessilifolia* Nutt.	northwest sandbar willow

Distribution

Pandemis pyrusana is distributed from Alberta west to British Columbia, south to Idaho, Utah, Colorado, and California (Dombroskie 2011, Powell and Opler 2009).

Planotortrix excessana **(Walker) [complex]**

Nomenclature

Planotortrix excessana (Walker) [complex] (Tortricidae: Tortricinae: Archipini)

Common names: greenheaded leafroller

Synonyms: *biguttana* (*Teras*)

Adult Recognition

Forewings are pale orange brown to dark reddish brown. Males are generally darker than females. Most individuals lack prominent wing markings except for a dark spot in the distal one-third of the forewing. Some individuals have a series of faint dark spots covering the wing and/or a white or pale spot in the basal one-third of the wing. Males have a forewing costal fold. FWL: 8.0-12.0 mm (male); 10.0-14.5 mm (female). There are at least two pheromonally distinct races assigned to the *P. excessana* complex which may represent distinct species (Dugdale et al. 2005, Langhoff 2009).

Adults are similar to other *Planotortrix* and the various species may be difficult to separate, even with a genitalic dissection. Dugdale et al. (2005) provide descriptions and illustrations for *P. notophaea* and *P. octo* in addition to *P. excessana*. Langhoff et al. (2009) used DNA barcodes to distinguish different species of *Ctenopseustis* and *Planotortrix*.

Larval Morphology

Last instar larvae are approximately 25 mm long and entirely green. The head is transparent light brown to green and may have faint brown mottling. The prothoracic shield is pale green with no lateral shading. An anal comb is present with 10-12 teeth (Dugdale et al.

2005, Thomas 1979). Larvae of *P. excessana* are occasionally intercepted at U.S. ports of entry on *Fragaria*, *Malus*, or *Prunus* originating in New Zealand.

Biology

The following life history information is summarized from Baker and Dick (1981), Dugdale et al. (2005), Thomas (1979), and Wearing et al. (1991). *Planotortrix excessana* completes 2-3 generations per year. On the South Island of New Zealand, adults are present February-March, April-May, and October-December.

Females lay eggs in masses that contain an average of 54 individual eggs. Egg masses of *P. excessana* have an opaque coating while those of *P. octo* are coated with white "particles." Early instars construct a silk shelter on the underside of leaves. Later instars web leaves together or web leaves to fruit and may cause economic damage by feeding directly on the surface of fruit. Pupation occurs in the larval shelter.

Larvae of *P. excessana* have been reported feeding on more than 100 species of plants (Table 60). This species is a pest of strawberries, stone fruits, and walnuts in New Zealand.

Table 60: *Planotortrix excessana* host plants

Family	**Genus/species**	**Common name**
Araliaceae	*Pseudopanax arboreus* K. Koch	
Asteraceae	*Chrysanthemum* L.	daisy
Cornocarpaceae	*Corynocarpus laevigata* Forst.	New Zealand laurel
Cupressaceae	*Cupressus sempervirens* L.	Italian cypress
Ebenaceae	*Diospyros kaki* L. f.	Japanese persimmon
Grossulariaceae	*Ribes nigrum* L.	European black currant
Onagraceae	*Fuchsia* L.	fuchsia
Pinaceae	*Cedrus deodara* (Roxb.) G. Don f.	Deodar cedar
Pittosporaceae	*Pittosporum crassifolium* Banks & Sol. ex A. Cunn.	stiffleaf cheesewood
Rosaceae	*Fragaria* L.	strawberry
Rosaceae	*Malus domestica* Borkh.	apple
Rosaceae	*Prunus armeniaca* L.	apricot
Rosaceae	*Prunus* L.	cherry
Rosaceae	*Prunus* L.	[unspecified]
Rosaceae	*Prunus persica* (L.) Batsch	peach

Table 60: *Planotortrix excessana* host plants

Family	Genus/species	Common name
Rosaceae	*Rosa* L.	rose
Rosaceae	*Rubus* L.	blackberry
Rutaceae	*Citrus* L.	citrus
Salicaceae	*Salix* L.	willow
Theaceae	*Camellia japonica* L.	camellia
Vitaceae	*Vitis vinifera* L.	wine grape

Distribution

Planotortrix excessana is native to New Zealand where it is present on both the North and South Islands. It has been artificially introduced to Hawaii (Dugdale et al. 2005).

***Xenotemna pallorana* (Robinson)**

Nomenclature

Xenotemna pallorana (Robinson) (Tortricidae: Tortricinae: Archipini)

Synonyms: *lata* (*Tortrix*)

Adult Recognition

Forewing color varies from pale yellow to cream to light brown. Many individuals are unmarked although some exhibit a faint reticulated forewing pattern. Hindwings are brownish gray and white or yellowish. Males lack a forewing costal fold. FWL: 8.5-14.0 mm.

Adults are similar to *Clepsis clemensiana*, and the two species are often mixed in collections. Males can be separated by the forewing costal fold, which is present in *C. clemensiana* and absent in *X. pallorana*. A genitalic dissection can be used to confirm identity.

Larval Morphology

Late instar larvae are approximately 16-28 mm long with a green abdomen. The head and prothoracic shield are green and unmarked. An anal comb is present with 7-9 teeth (MacKay 1962). The unmarked larvae of *X. pallorana* may appear similar to those of many other Archipini.

Biology

Xenotemna pallorana completes two annual generations over much of its range. Adults are present May to August. Females lay eggs in masses on the upper surface of leaves. Larvae feed in folded leaves and mid-instar larvae construct a hibernaculum in which overwintering occurs. Feeding resumes in the spring and pupation occurs in the final larval feeding site (Chapman and Lienk 1971).

Chapman and Lienk (1971) suggest that primary larval hosts are limited to herbaceous legumes such as alfalfa and white sweet clover. Other larval hosts, such as apple, are considered secondary hosts. This species has also been reported to cause economic injury to young pines in plantations. A list of documented host plants is presented in Table 61.

Table 61: *Xenotemna pallorana* host plants

Family	**Genus/species**	**Common name**
Asteraceae	*Aster* L.	aster
Asteraceae	*Erigeron annuus* (L.) Pers.	eastern daisy
Asteraceae	*Silphium* L.	rosinweed
Asteraceae	*Solidago* L.	goldenrod
Asteraceae	*Symphyotrichum novae-angliae* (L.) G. L. Nesom	New England aster
Clusiaceae	*Hypericum perforatum* L.	common St. Johnswort
Fabaceae	*Medicago sativa* L.	alfalfa
Fabaceae	*Melilotus officinalis* (L.) Lam.	yellow sweetclover
Fabaceae	*Melilotus* Mill.	sweetclover
Fabaceae	*Trifolium* L.	clover
Lamiaceae	*Monarda fistulosa* L.	wild bergamot
Pinaceae	*Picea glauca* (Moench) Voss	white spruce
Pinaceae	*Pinus banksiana* Lamb.	jack pine
Pinaceae	*Pinus resinosa* Aiton	red pine
Pinaceae	*Pinus strobus* L.	eastern white pine
Pinaceae	*Pinus sylvestris* L.	Scots pine
Pinaceae	*Pinus* L.	pine
Rosaceae	*Fragaria* L.	strawberry
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus pumila* L.	sandcherry
Rosaceae	*Prunus serotina* Ehrh.	black cherry
Rosaceae	*Prunus virginiana* L.	chokecherry

Table 61: *Xenotemna pallorana* host plants

Family	**Genus/species**	**Common name**
Rosaceae	*Prunus* L.	
Rosaceae	*Rosa* L.	rose
Santalaceae	*Comandra umbellata* (L.) Nutt.	bastard toadflax
Ulmaceae	*Ulmus* L.	elm
Verbenaceae	*Verbena* L.	vervain

Distribution

Xenotemna pallorana is widespread in the continental United States and southern Canada (Chapman and Lienk 1971, Powell and Opler 2009).

Tortricidae: Tortricinae: Sparganothini

Amorbia Clemens

Overview

The genus *Amorbia* contains approximately 40 species (Baixeras et al. 2008). Most are distributed in the Neotropics, although a few are present in the Nearctic. Adults are characterized by the absence of ocelli, a dark line of scales on the apical area of the hindwing, and several genitalic characters, including: gnathos absent; aedeagus lacking external armature; corpus bursae sac-shaped; signum large and band-shaped; and sterigma developed into a pair of lateral pockets (Phillips-Rodriguez and Powell 2007).

The majority of *Amorbia* larvae arriving at U.S. ports are from Central and South America. Larvae have been intercepted on *Crataegus* (hawthorn), *Limonium* (lavender), *Ocimum* (basil), *Persea* (avocado), *Rubus* (blackberry, raspberry, etc.), and cut flowers. Interceptions are very common although most specimens are not identified to species.

Late instar larvae can be distinguished by the dark brown to black lateral bands on the head and prothoracic shield. Identification of early instars is difficult as the lateral bands are often absent and molecular diagnostics may be necessary to confirm identity.

The two species treated here, *A. cuneana* and *A. emigratella*, are occasional pests of various crops in western North America. Adults and larvae are most often encountered as non-target captures during surveys for exotic pests such as *Epiphyas postvittana*.

Amorbia cuneana **(Walsingham)**

Nomenclature

Amorbia cuneana (Walsingham) (Tortricidae: Tortricinae: Sparganothini)

Common names: western avocado leafroller

Synonyms: *adumbrana* (*Amorbia*), *essigana* (*Amorbia*), *synneurana* (*Amorbia*)

Adult Recognition

Male forewing color varies from pale tan to rust to dark gray brown. Many individuals have a dark costal spot that is a remnant of the median fascia or more well-defined transverse markings. Females have a more uniform rust to tan forewing with obscured marking. Males lack a forewing costal fold. FWL: 10.0-14.5 mm (male); 13.5-17.5 mm (female). A dorsal pit is present on the second abdominal segment and ocelli are absent (Phillips-Rodriguez and Powell 2007).

Few other North American tortricids are similar in size or appearance to *Amorbia*, and this is one of the few tortricid genera in which the adults lack ocelli. The two *Amorbia* species treated here can be separated by the number of dorsal pits on the abdomen: dorsal pits on segments 2-6 for *A. emigratella* versus a single dorsal pit on segment 2 for *A. cuneana*.

Larval Morphology

Last instar larvae are approximately 25mm long. The head and prothoracic shield are tan and marked with dark brown to black lateral bands; these lateral bands can be used to distinguish late instar *Amorbia* larvae from those of many other tortricids. Early instars lack the distinctive dark bands, and morphological identification of early instar larvae may be difficult or impossible.

Biology

The following life history information is compiled from Faber et al. (2010), Hoffman et al. (1983), and Powell and Opler (2009). *Amorbia cuneana* completes two generations per year in northern California, where adults are present in May through June and again in October. In southern California, *A. cuneana* is multivoltine and adults have been recorded every month of the year.

Larvae feed on leaves, skeletonizing them in early instars and consuming the entire leaf in later instars. Larvae also web leaves to fruit and feed on the skin of fruit in a cluster, sometimes causing economic damage. A list of documented host plants is presented in Table 62.

Amorbia cuneana can be a significant pest of avocado in California. Its putative synonym, *A. essigana* Busck, was described as an avocado pest in southern California in 1929, and it is possible that avocado-feeding populations may represent this "pheromone race."

Distribution

Amorbia cuneana is found along the Pacific Coast from southwestern Canada south to Baja California and east to Arizona and Idaho (Powell and Opler 2009).

Table 62: *Amorbia cuneana* host plants

Family	**Genus/species**	**Common name**
Caprifoliaceae	*Lonicera japonica* Thunb.	Japanese honeysuckle
Ericaceae	*Arbutus menziesii* Pursh	Pacific madrone
Ericaceae	*Arctostaphylos glauca* Lindl.	bigberry manzanita
Ericaceae	*Arctostaphylos insularis* Greene ex Parry	island manzanita
Ericaceae	*Arctostaphylos patula* Greene	greenleaf manzanita
Ericaceae	*Arctostaphylos* Adans.	manzanita
Grossulariaceae	*Ribes sanguineum* Pursh	redflower currant
Lamiaceae	*Trichostema lanceolatum* Benth.	vinegarweed
Lauraceae	*Cinnamomum camphora* (L.) J. Presl	camphortree
Lauraceae	*Laurus nobilis* L.	sweet bay
Lauraceae	*Persea americana* Mill.	avocado
Lauraceae	*Persea* Mill.	bay

Table 62: *Amorbia cuneana* host plants

Family	**Genus/species**	**Common name**
Lauraceae	*Umbellularia californica* (Hook. & Arn.) Nutt.	California laurel
Pinaceae	*Abies concolor* (Gord. & Glend.) Lindl. ex Hildebr.	white fir
Pinaceae	*Pseudotsuga menziesii* (Mirb.) Franco	Douglas-fir
Rhamnaceae	*Ceanothus arboreus* Greene	feltleaf ceanothus
Rhamnaceae	*Ceanothus leucodermis* Greene	chaparral whitethorn
Rhamnaceae	*Ceanothus sorediatus* Hook. & Arn.	jimbrush
Rosaceae	*Heteromeles arbutifolia* (Lindl.) M. Roem. var. *arbutifolia*	toyon
Rosaceae	*Lyonothamnus floribundus* A. Gray	Catalina ironwood
Rosaceae	P*runus ilicifolia* (Nutt. ex Hook. & Arn.) D. Dietr.	hollyleaf cherry
Rosaceae	*Prunus ilicifolia* (Nutt. ex Hook. & Arn.) D. Dietr. ssp. *lyonii* (Eastw.) P.H. Raven	hollyleaf cherry
Rosaceae	*Pyracantha* M. Roem.	firethorn
Rutaceae	*Citrus* L.	citrus
Ulmaceae	*Ulmus americana* L.	American elm
Vitaceae	*Vitis* L.	grape

***Amorbia emigratella* Busck**

Nomenclature

Amorbia emigratella Busck (Tortricidae: Tortricinae: Sparganothini)

Common names: Mexican leafroller

Adult Recognition

Adults are pale yellow to brown and may be variably mottled with dark brown. Most individuals have a well defined costal spot that is a remnant of the median fascia. Abdominal segments 2-6 have a median dorsal pit and ocelli are absent (Phillips-Rodriguez and Powell 2007). Males lack a forewing costal fold. FWL: 8.0-11.0 mm (M); 11.5-12.0 mm (F).

Few other North American tortricids are similar in size or appearance to *Amorbia*, and this is one of the few tortricid genera in which the adults lack ocelli. The two *Amorbia* species treated here can be separated by the number of dorsal pits on the abdomen: dorsal pits on segments 2-6 for *A. emigratella* versus a single dorsal pit on segment 2 for *A. cuneana*.

Larval Morphology

Last instar larvae are yellowish green and may have dark lateral lines. The prothoracic shield and head is tan to brown with dark-brown to black lateral bands.

Late instar *Amorbia* larvae can be distinguished from most other tortricids by the dark-brown to black lateral bands on the head and prothoracic shield. These characters can be used to separate late instar larvae of *A. emigratella* and *Epiphyas postvittana* from Hawaii, as both occur on similar host plants (Zimmerman 1978). Early instars lack the distinctive dark bands, and morphological identification of early instar larvae may be difficult or impossible.

Biology

Eggs are laid in masses of 65-120 eggs on the upper surface of leaves. Females cover the egg mass with a whitish coating. Larvae feed within a shelter constructed of webbed leaves or leaves webbed to fruit; they may also feed within flower heads or buds. Larvae complete 4-5 instars and pupation occurs in the larval shelter. Adults eclose in approximately 10 days. *Amorbia emigratella* has been recorded from plants in more than 25 families (Table 63), many of which include economically important crops (Fullaway and Krauss 1945, Zimmerman 1978).

Table 63: *Amorbia emigratella* host plants

Family	**Genus/species**	**Common name**
Apiaceae	*Daucus carota* L.	Queen Anne's lace
Apocynaceae	*Nerium oleander* L.	oleander
Araliaceae	*Schefflera actinophylla* (Endl.) Harms	octopus tree
Asteraceae	*Parthenium hysterophorus* L.	Santa Maria feverfew
Brassicaceae	*Brassica oleracea* L.	cabbage
Cannabaceae	*Cannabis sativa* L.	marijuana
Caricaceae	*Carica papaya* L.	papaya
Convolvulaceae	*Ipomoea batatas* (L.) Lam.	sweetpotato
Euphorbiaceae	*Jatropha gossypifolia* L.	bellyache bush
Fabaceae	*Acacia koaia* Hillebr.	koaoha
Fabaceae	*Arachis hypogaea* L.	peanut
Fabaceae	*Chamaecrista nictitans* (L.) Moench	sensitive partridge pea
Fabaceae	*Gliricidia sepium* (Jacq.) Kunth ex Walp.	quickstick
Fabaceae	*Phaseolus* L.	bean

Table 63: *Amorbia emigratella* host plants

Family	**Genus/species**	**Common name**
Fabaceae	*Sophora* L.	necklacepod
Fabaceae	*Ulex europaeus* L.	common gorse
Lauraceae	*Persea americana* Mill.	avocado
Malvaceae	*Gossypium hirsutum* L. var. *hirsutum*	upland cotton
Malvaceae	*Gossypium* L.	cotton
Myrtaceae	*Psidium guajava* L.	guava
Orchidaceae	[unspecified]	
Orchidaceae	*Phaius* Lour.	nun's-hood orchid
Passifloraceae	*Passiflora* L.	passionflower
Poaceae	*Zea mays* L.	corn
Proteaceae	*Macadamia* F. Muell.	macadamia
Rosaceae	*Rosa* L.	rose
Rosaceae	*Rubus hawaiensis* A. Gray	Hawaii blackberry
Rosaceae	*Rubus* L.	blackberry
Rubiaceae	*Gardenia* Ellis	gardenia
Ruscaceae	*Dracaena* L.	dracaena
Rutaceae	*Citrus* X *sinensis* (L.) Osbeck (pro sp.) [*maxima* X *reticulata*]	sweet orange
Sapindaceae	*Dodonaea viscosa* (L.) Jacq.	Florida hopbush
Solanaceae	*Solanum lycopersicum* L. var. *lycopersicum*	garden tomato
Solanaceae	*Solanum melongena* L.	eggplant
Solanaceae	*Solanum tuberosum* L.	Irish potato
Stericulaceae	*Theobroma cacao* L.	cacao
Thymelaeaceae	*Wikstroemia foetida* var. *oahuensis* Gray	
Urticaceae	*Pipturus* Weddell	pipturus
Verbenaceae	*Lippia alba* (Mill.) N. E. Br. ex Britton & P. Wilson	bushy lippia

Distribution

Amorbia emigratella is distributed across the southern United States and Central America. It has been introduced to all major Hawaiian Islands except Lanai (Phillips-Rodriguez and Powell 2007, Zimmerman 1978).

Platynota **Clemens**

Overview

The genus *Platynota* contains approximately 30 species (Baixeras et al. 2008). The highest species diversity is in the Neotropics, although a few species are described from the northern U.S. and Canada. Adults are sexually dimorphic and wing pattern varies throughout the genus. The forewings of many males are dark brown basally and golden or yellowish brown apically. Female forewings are usually uniformly brown or yellowish brown with dark brown markings. Males have a forewing costal fold. Male genitalia are characterized by a long thin uncus, long, fingerlike socii, and elongate subrectangular valvae. Female genitalia are characterized by a bandlike sterigma and a variably-shaped signum in some species. Labial palpi are elongate in most species.

The three species treated here, *P. flavedana*, *P. idaeusalis*, and *P. stultana*, are pests of various crops, including apple, citrus, grape, peach, pepper, rose, and strawberry. Larvae are highly polyphagous and some species, such as *P. stultana*, have been recorded feeding on plants in more than 20 families. As these three species are likely the most common *Platynota* in the continental U.S., they are presented here as non-targets potentially encountered during domestic surveys.

Larvae of *Platynota* are commonly intercepted at U.S. ports of entry on peppers (*Capsicum*) from the Caribbean and Mexico, and apple (*Malus*), basil (*Ocimum*), rose (*Rosa*), and *Rubus* from Cental and South America. Larvae are difficult to identity using only morphology, and most identifications are based on a host/origin association. Larvae of *Platynota* may be confused with those of *Argyrotaenia*, other *Archipini*, or other *Sparganothini*.

***Platynota flavedana* Clemens**

Nomenclature

Platynota flavedana Clemens (Tortricidae: Tortricinae: Sparganothini)

Synonyms: *concursana* (*Teras*), *iridana* (*Platynota*), *laterana* (*Teras*), *tinctana* (*Teras*)

Adult Recognition

Platynota flavedana is sexually dimorphic. The male forewing is dark purplish brown basally and yellowish to orangish brown apically. The female forewing is brown to orangish brown with dark-brown to purplish-brown markings. Hindwings of both sexes are brown to orangish brown; males tend to exhibit more orange than females. Males have a long forewing costal fold that extends to mid-costa. FWL: 5.0-6.5 mm (male); 6.0-8.5 mm (female).

Adults may appear similar to other species of *Platynota*, but can be separated from most other species by their orangish appearance. A genitalic dissection can be used to confirm identity.

Larval Morphology

Late instar larvae are approximately 13-21 mm in length with a green to pale green abdomen. The head and prothoracic shield are brownish yellow. An anal comb is present with 5-8 teeth (MacKay 1962).

Larvae appear similar to those of *Sparganothis sulfureana*, and the two species are often found in similar habitats in the eastern United States. MacKay (1962) stated that larvae of *Platynota* could be separated from similar species of *Sparganothis* by the small dorsal pinacula on A1-8, which are slightly elongate and cream colored in living individuals.

Biology

The following life history information is compiled from Chapman and Lienk (1971), Sandberg and Passoa (1989), and Wilde and Semel (1966). *Platynota flavedana* completes two annual generations over much of its range; a partial third generation may be present in the South. Adults are present May-June and July-September.

Table 64: *Platynota flavedana* host plants

Family	**Genus/species**	**Common name**
Aceraceae	*Acer* L.	maple
Asteraceae	*Eupatorium* L.	thoroughwort
Asteraceae	*Helianthus* L.	sunflower
Caryophyllaceae	*Dianthus caryophyllus* L.	carnation
Cistaceae	*Helianthemum* Mill.	frostweed
Clusiaceae	*Hypericum perforatum* L.	common St. Johnswort
Ericaceae	*Rhododendron* L.	rhododendron
Fabaceae	*Trifolium* L.	clover
Lauraceae	*Sassafras* Nees & Eberm.	sassafras
Malvaceae	*Gossypium hirsutum* L. var. *hirsutum*	upland cotton
Malvaceae	*Gossypium thurberi* Todaro	Thurber's cotton
Rosaceae	*Fragaria* L.	strawberry
Rosaceae	*Prunus persica (L.) Batsch* L.	peach
Rosaceae	*Rosa* L.	rose
Rutaceae	*Citrus* L.	citrus
Rutaceae	*Citrus* X *sinensis* (L.) Osbeck (pro sp.) [*maxima* X *reticulata*]	sweet orange

Females lay eggs in masses that contain approximately 50 individual eggs on the upper surface of leaves. Larvae feed within a shelter constructed of tied or folded leaves. Larvae may cause economic damage by feeding on blossoms or fruit, and will often web leaves together with blossoms and immature fruit. Mid-instar larvae of the last generation overwinter and resume feeding the following spring. Pupation occurs in webbed leaves. Larvae of *P. flavedana* have been reported as pests of strawberry, peach, and rose. Other important hosts include cotton and citrus. A list of documented hosts is presented in Table 64.

Distribution

Platynota flavedana is widely distributed in the eastern United States. Records from the western U.S. are questionable.

***Platynota idaeusalis* (Walker)**

Nomenclature

Platynota idaeusalis (Walker) (Tortricidae: Tortricinae: Sparganothini)

Common names: tufted apple bud moth

Synonyms: *dioptrica* (*Phylacteritis*), *sentana* (*Platynota*)

Adult Recognition

Forewings are gray with reddish brown to dark brown markings. Hindwings are grayish brown to dark brown. This species' common name is derived from the tufts of dark raised scales on the forewings. Labial palpi are elongate in both sexes. Males have a forewing costal fold. FWL: 6.0-12.5 mm.

The combination of elongate labial palpi and grayish forewings with reddish-brown markings is sufficient to separate this species from most other North American Tortricidae. A genitalic dissection can be used to confirm identity.

Larval Morphology

Late instar larvae are approximately 13-18 mm in length with a brownish-green abdomen. The head and prothoracic shield are brown with dark-brown mottling on the head and lateral shading on the shield. An anal comb is present with 5-8 teeth (MacKay 1962).

MacKay (1962) stated that larvae of *Platynota* could be separated from similar species of *Sparganothis* by the small dorsal pinacula on A1-8, which are slightly elongate and cream colored in living individuals.

Biology

The following life history information is summarized from Boyne et al. (1985), Chapman and Lienk (1971), and Hogmire and Howitt (1979). *Platynota idaeusalis* completes two generations per year. Adults are present May-July and again in August-September.

Females deposit eggs in large masses that contain approximately 100 individual eggs on the upper surface of leaves. Early instars construct a silk web on the underside of a leaf along the midrib and feed inside. Later instars feed within a shelter constructed of rolled, folded, or tied leaves. Larvae can cause fruit damage by webbing leaves to fruit and feeding on the surface of the fruit. Larval feeding can lead to early fruit drop as well as cosmetic damage. Pupation occurs in a folded or rolled leaf. Platynota idaeusalis is a pest of apple in the eastern U.S., although the diverse list of host plants (Table 65) suggests that the larvae are general feeders.

Distribution

Platynota idaeusalis is widely distributed in eastern North America and is also found in the Pacific Northwest (Chapman and Lienk 1971). Although not documented, it likely occurs throughout all of northern United States and southern Canada.

Table 65: *Platynota idaeusalis* host plants

Family	**Genus/species**	**Common name**
Asteraceae	*Vernonia noveboracensis* (L.) Michx.	New York ironweed
Betulaceae	*Betula* L.	birch
Caprifoliaceae	*Viburnum prunifolium* L.	blackhaw
Cornaceae	*Cornus drummondii* C. A. Mey.	roughleaf dogwood
Ericaceae	*Vaccinium* L.	blueberry
Moraceae	*Maclura pomifera* (Raf.) C. K. Schneid.	osage orange
Oleaceae	*Fraxinus nigra* Marsh.	black ash
Pinaceae	*Pinus banksiana* Lamb.	jack pine
Ranunculaceae	*Clematis* L.	leather flower
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Rubus idaeus* L.	American red raspberry
Rosaceae	*Rubus* L.	blackberry
Salicaceae	*Salix* L.	willow
Solanaceae	*Solanum* L.	nightshade
Vitaceae	*Vitis* L.	grape

***Platynota stultana* Walsingham**

Nomenclature

Platynota stultana Walsingham (Tortricidae: Tortricinae: Sparganothini)

Common names: omnivorous leafroller

Synonyms: *chiquitana* (*Platynota*)

Adult Recognition

Male forewings are usually dark brown on the basal half and golden brown on the distal half. Female forewings are more uniform golden brown to dark brown and markings are usually less distinct. Labial palpi are extremely elongate in both sexes. Males have a forewing costal fold. FWL: 4.5-7.0 mm (male); 6.5-9.0 mm (female).

Platynota stultana is similar to other *Platynota* species such as *P. flavedana* and *P. rostrana*. The forewing costal fold is generally smaller in *P. stultana* than in these other species. A genitalic dissection can be used to confirm identity.

Larval Morphology

Late instar larvae are approximately 12-15 mm long with a cream-colored, translucent abdomen. The head and prothoracic shield are yellowish brown. The posterolateral margins on the prothoracic shield are shaded with dark brown in some individuals. An anal comb is present with 5-6 teeth (MacKay 1962).

MacKay (1962) stated that larvae of *Platynota* could be separated from similar species of *Sparganothis* by the small dorsal pinacula on A1-8, which are slightly elongate and cream colored in living individuals.

Biology

The following life history information is summarized from AliNiazee and Stafford (1972) and Atkins et al. (1957). *Platynota stultana* completes 4-6 generations per year. Adults may be present year round over much of its range.

Eggs are laid in masses containing an average of 97 individual eggs per mass. Newly hatched larvae move towards the top of the plant and feed within a bud or between two leaves. Young larvae may also disperse to other hosts by ballooning in the wind on a silk thread. Later instars feed within a shelter constructed of rolled or folded leaves. Larvae complete 5-6 instars in a period of 20-30 days (in greenhouse conditions). Third through fifth instar lavae of the last generation overwinter in webbed nests. Pupation takes place in a rolled leaf.

Larvae of *P. stultana* are highly polyphagous and have been recorded feeding on plants in more than 20 families (Table 66). This species can be a serious pest in greenhouses and vineyards and economically important hosts include alfalfa, citrus, corn, cotton, grape, peach, pear, and pepper. As *P. stultana* expanded its range into northern California it appears to also have greatly expanded its host range onto a wide variety of non-native plants. It is recorded from only a few native plants in California (Powell 1983).

Table 66: *Platynota stultana* host plants

Family	**Genus/species**	**Common name**
Amaranthaceae	*Amaranthus* L.	pigweed
Apiaceae	*Apium graveolens* L.	wild celery
Apiaceae	*Conium maculatum* L.	poison hemlock
Asteraceae	*Ambrosia dumosa* (A. Gray) Payne	burrobush
Asteraceae	*Ambrosia psilostachya* DC.	Cuman ragweed
Asteraceae	*Aster* L.	aster
Asteraceae	*Baccharis pilularis* DC.	coyotebrush
Asteraceae	*Bidens laevis* (L.) Britton et al.	smooth beggartick
Asteraceae	*Conyza bilbaoana* J. Remy	
Asteraceae	*Grindelia camporum* Greene	Great Valley gumweed
Asteraceae	*Grindelia hirsutula* Hook. & Arn.	hairy gumweed
Asteraceae	*Parthenium hysterophorus* L.	Santa Maria feverfew
Asteraceae	*Senecio jacobaea* L.	stinking willie
Asteraceae	*Solidago californica* Nutt.	California goldenrod
Asteraceae	*Wyethia angustifolia* (DC.) Nutt.	California compassplant
Caryophyllaceae	*Dianthus caryophyllus* L.	carnation
Caryophyllaceae	*Dianthus* L.	pink
Chenopodiaceae	*Atriplex calotheca* (Rafn) Fr.	halberdleaf orach
Chenopodiaceae	*Beta* L.	beet

Table 66: *Platynota stultana* host plants

Family	**Genus/species**	**Common name**
Chenopodiaceae	*Chenopodium* L.	goosefoot
Chenopodiaceae	*Salsola kali* L.	Russian thistle
Convolvulaceae	*Convolvulus* L.	bindweed
Crassulaceae	*Dudleya virens* (Rose) Moran	bright green dudleya
Cupressaceae	*Juniperus* L.	juniper
Fabaceae	*Albizzia* Durazz.	albizia
Fabaceae	*Arachis* L.	peanut
Fabaceae	*Delonix regia* (Bojer ex Hook.) Raf.	royal poinciana
Fabaceae	*Glycine max* (L.) Merr.	soybean
Fabaceae	*Leucaena lanceolata* S. Watson	
Fabaceae	*Leucaena leucocephala* (Lam.) de Wit	white leadtree
Fabaceae	*Lotus scoparius* (Nutt.) Ottley	common deerweed
Fabaceae	*Medicago sativa* L.	alfalfa
Fabaceae	*Melilotus officinalis* (L.) Lam.	yellow sweetclover
Fabaceae	*Mimosa asperata* L.	Puerto Rico sensitive-briar
Fabaceae	*Parkinsonia aculeata* L.	Jerusalem thorn
Fabaceae	*Phaseolus* L.	bean
Fabaceae	*Tamarindus indica* L.	tamarind
Fabaceae	*Trifolium* L.	clover
Ginkgoaceae	*Ginkgo* L.	ginkgo
Juglandaceae	*Juglans* L.	walnut
Lauraceae	*Persea americana* Mill.	avocado
Malvaceae	*Gossypium hirsutum* L. var. *hirsutum*	upland cotton
Malvaceae	*Malva* L.	mallow
Onagraceae	*Epilobium brachycarpum* C. Presl	tall annual willowherb
Pinaceae	*Pinus* L.	pine
Poaceae	*Sorghum* Moench	sorghum
Poaceae	*Zea mays* L.	corn
Polygonaceae	*Eriogonum grande* Greene var. *grande*	redflower buckwheat
Polygonaceae	*Eriogonum latifolium* Sm.	seaside buckwheat
Polygonaceae	*Rumex crispus* L.	curly dock
Portulaceae	*Portulaca* L.	purslane
Portulaceae	*Portulaca oleracea* L.	little hogweed
Primulaceae	*Cyclamen* L.	cyclamen
Punicaceae	*Punica granatum* L.	pomegranate
Rosaceae	*Rosa* L.	rose
Rosaceae	*Rubus* L.	blackberry
Rutaceae	*Citrus* L.	citrus
Rutaceae	*Citrus* X *limon* (L.) Burm. f. (pro sp.) [*medica* X *aurantifolia*]	lemon

Table 66: *Platynota stultana* host plants

Family	**Genus/species**	**Common name**
Rutaceae	*Citrus* X *sinensis* (L.) Osbeck (pro sp.) [*maxima* X *reticulata*]	sweet orange
Salicaceae	*Salix* L.	willow
Solanaceae	*Capsicum* L.	pepper
Solanaceae	*Solanum lycopersicum* L. var. *lycopersicum*	garden tomato
Taxaceae	*Taxus* L.	yew
Verbenaceae	*Citharexylum spinosum* L.	spiny fiddlewood
Vitaceae	*Vitis* L.	grape
Vitaceae	*Vitis vinifera* L.	wine grape

Distribution

Platynota stultana has been recorded from California, Arizona, Hawaii, Texas, Florida, and Mexico. Records from other locations in the eastern United States are likely a result of artificial introduction by movement of infested host plants (J. Brown, pers. comm.). This species was recently discovered in pepper (*Capsicum*) shipments originating from Spain, suggesting that it has been introduced into that country.

***Sparganothis senecionana* (Walsingham)**

Nomenclature

Sparganothis senecionana (Walsingham) (Tortricidae: Tortricinae: Sparganothini)

Synonyms: *rudana* (*Oenectra*)

Adult Recognition

Forewings are pale yellow with brown to purplish-brown markings. Wing pattern varies greatly, but most individuals are marked with an incomplete median fascia and conspicuous costal spot. The distal half of the wing may be lightly shaded or solid brown. Labial palpi are long. Males lack a forewing costal fold. FWL: 7.5-12.5 mm.

Adults may be confused with other *Sparganothis*. *S. tunicana* is similar but can be distinguished from *S. senecionana* by its smaller size and shorter labial palpi.

Biology

Sparganothis senecionana completes a single annual generation over much of its range. Adults are present April to August depending on location. Larvae are highly polyphagous (Table 67) and generally feed on flowers of the host. In California, larvae hatch from the egg, construct a silk hibernaculum, and enter an obligate diapause that lasts through the dry season. Activity is resumed again at the onset of winter rains (Lambert 1950, Powell and Opler 2009).

Table 67: *Sparganothis senecionana* host plants

Family	**Genus/species**	**Common name**
Amaryllidaceae	*Brodiaea* Sm.	brodiaea
Apiaceae	*Lomatium californicum* (Nutt.) Mathias & Constance	California lomatium
Asteraceae	*Achillea* L.	yarrow
Asteraceae	*Achillea milleflorum* L.	
Asteraceae	*Anaphalis margaritacea* (L.) Benth.	western pearly everlasting
Asteraceae	*Artemisia douglasiana* Besser	Douglas' sagewort
Asteraceae	*Balsamorhiza sagittata* (Pursh) Nutt.	arrowleaf balsamroot
Asteraceae	*Gnaphalium* L.	cudweed
Asteraceae	*Hieracium scouleri* Hook. var. *albertinum* (Farr) G. W. Douglas & G. A. Allen	
Asteraceae	*Senecio integerrimus* Nutt.	lambstongue ragwort
Asteraceae	*Senecio* L.	ragwort
Asteraceae	*Wyethia* Nutt.	mule-ears
Boraginaceae	*Cynoglossum grande* Dougl. ex Lehm.	Pacific hound's tongue
Boraginaceae	*Horkelia californica* Cham. & Schltdl.	California horkelia
Boraginaceae	*Horkelia* Cham. & Schltdl.	horkelia
Brassicaceae	*Barbarea orthoceras* Ledeb.	American yellowrocket
Clusiaceae	*Hypericum perforatum* L.	common St. Johnswort
Cornaceae	*Cornus* L.	dogwood
Fabaceae	*Lupinus albifrons* Benth.	silver lupine
Fabaceae	*Thermopsis macrophylla* Hook. & Arn.	Santa Inez goldenbanner
Hydrophyllaceae	*Eriodictyon californicum* (Hook. & Arn.) Torr.	California yerba santa
Hydrophyllaceae	*Phacelia californica* Cham.	California phacelia
Hydrophyllaceae	*Phacelia hastata* Douglas ex Lehm.	silverleaf phacelia
Iridaceae	*Iris* L.	iris
Lamiaceae	*Stachys rigida* Nutt. ex Benth.	rough hedgenettle
Lilaceae	*Chlorogalum pomeridianum* (DC.) Kunth	wavyleaf soap plant
Lilaceae	*Zigadenus paniculatus* (Nutt.) S. Watson	foothill deathcamas

Table 67: *Sparganothis senecionana* host plants

Family	**Genus/species**	**Common name**
Liliaceae	*Chlorogalum* Kunth	soapplant
Onagraceae	*Clarkia* Pursh	clarkia
Pinaceae	*Abies concolor* (Gord. & Glend.) Lindl. ex Hildebr.	white fir
Pinaceae	*Abies magnifica* A. Murray	California red fir
Pinaceae	*Abies* Mill.	fir
Pinaceae	*Pseudotsuga menziesii* (Mirb.) Franco	Douglas-fir
Primulaceae	*Dodecatheon hendersonii* A. Gray	mosquito bills
Rhamnaceae	*Ceanothus papillosus* Torr. & A. Gray	wartleaf ceanothus
Rosaceae	*Cercocarpus montanus* Raf.	alderleaf mountain mahogany
Rosaceae	*Horkelia fusca* Lindl.	pinewoods horkelia
Rosaceae	*Prunus* L.	[unspecified]
Rosaceae	*Prunus virginiana* L.	chokecherry
Scrophulariaceae	*Castilleja applegatei* Fernald ssp. *pinetorum* (Fernald) T.I. Chuang & Heckard	wavyleaf Indian paintbrush
Scrophulariaceae	*Penstemon deustus* Douglas ex Lindl.	scabland penstemon
Scrophulariaceae	*Penstemon newberryi* A. Gray	mountain pride
Scrophulariaceae	*Penstemon* Schmidel	beardtongue
Scrophulariaceae	*Penstemon speciosus* Douglas ex Lindl.	royal penstemon

Distribution

Sparganothis senecionana is distributed throughout western North America from British Columbia to Mexico and east to Colorado. It is absent in desert locations (Lambert 1950, Powell and Opler 2009).

Sparganothis sulfureana **(Clemens)**

Nomenclature

Sparganothis sulfureana (Clemens) (Tortricidae: Tortricinae: Sparganothini)

Common names: Sparganothis fruitworm

Synonyms: *belfrageana* (var.), *euphronopa* (*Sparganothis*), *fulvoroseana* (*Croesia*?), *gallivorana* (*Croesia*?), *gratana* (*Conchylis*), *virgineana* (*Croesia*), *virginiana* (*Croesia*?)

Adult Recognition

Forewings are bright yellow with orange to purplish markings. Wing pattern is variable, but most individuals have two marks along the costa and a “V” shaped mark extending from the costa to dorsum. This pattern creates the appearance of an “X” when the moth is resting with its wings folded. Other individuals may be nearly patternless or have extensive reticulations. Hindwings are light gray to grayish brown. FWL: 6.5-10.0 mm. Some adult forms are similar to other species of *Sparganothis*, including *S. lycopodiana* and *S. unifasciana*.

Larval Morphology

Late instar larvae are approximately 13-17 mm long. The abdomen is yellowish to grayish green with lighter, conspicuous pinacula. The head is yellowish to reddish brown with antennae that are white basally and black distally. The prothoracic shield is concolorous with the abdomen and is edged laterally (and possibly posterolaterally) with black. An anal comb is present with 6-9 teeth (Landry et al. 2002, MacKay 1962).

Biology

The following life history information is summarized from Beckwith (1938) and Landry et al. (2002). *Sparganothis sulfureana* completes two generations per year. Adults are present from mid-June to July and August to September. Females lay eggs in masses that contain 20-50 individual eggs on the upper surface of leaves. Larvae of the first (spring) generation feed on flower buds and leaves; those of the second (summer) generation feed on leaves and fruit. Overwintering occurs as an early instar larva in leaf debris on the ground.

Larvae of *S. sulfureana* are polyphagous and have been recorded feeding on plants in nearly 20 families (Table 68). This species is best known as a serious pest of cranberry, where it causes damage to both leaves and fruit.

Distribution

Sparganothis sulfureana is widespread in eastern North America. There are also records from the Pacific Northwest (E. LaGasa pers. comm.).

Table 68: *Sparganothis sulfureana* host plants

Family	**Genus/species**	**Common name**
Apiaceae	*Apium graveolens* L.	wild celery
Apiaceae	*Hydrocotyle* L.	hydrocotyle
Asteraceae	*Arctium* L.	burdock
Asteraceae	*Aster* L.	aster
Asteraceae	*Erigeron annuus* (L.) Pers.	eastern daisy
Asteraceae	*Helianthus* L.	sunflower
Asteraceae	*Solidago* L.	goldenrod
Asteraceae	*Symphyotrichum novae-angliae* (L.) G.L. Nesom	New England aster
Clusiaceae	*Hypericum* L.	St. Johnswort
Clusiaceae	*Hypericum perforatum* L.	common St. Johnswort
Cupressaceae	*Thuja occidentalis* L.	arborvitae
Ericaceae	*Vaccinium* L.	blueberry
Ericaceae	*Vaccinium* L.	cranberry
Ericaceae	*Vaccinium macrocarpon* Aiton	cranberry
Fabaceae	*Gleditsia* L.	locust
Fabaceae	*Medicago sativa* L.	alfalfa
Fabaceae	*Phaseolus lunatus* L.	sieva bean
Fabaceae	*Robinia* L.	locust
Fabaceae	*Trifolium* L.	clover
Fabaceae	*Trifolium pratense* L.	red clover
Lamiaceae	*Mentha* L.	mint
Lamiaceae	*Monarda fistulosa* L.	wild bergamot
Liliaceae	*Lilium* L.	lily
Onagraceae	*Oenothera* L.	evening primrose
Pinaceae	*Abies balsamea* (L.) Mill.	balsam fir
Pinaceae	*Larix* Mill.	larch
Pinaceae	*Picea glauca* (Moench) Voss	white spruce
Pinaceae	*Pinus banksiana* Lamb.	jack pine
Pinaceae	*Pinus resinosa* Aiton	red pine
Pinaceae	*Pinus rigida* Mill.	pitch pine
Pinaceae	*Pinus strobus* L.	eastern white pine
Pinaceae	*Pinus sylvestris* L.	Scots pine
Pinaceae	*Pinus* L.	pine
Poaceae	*Zea mays* L.	corn
Ranunculaceae	*Ranunculus* L.	buttercup
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Fragaria* L.	strawberry
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Prunus virginiana* L.	chokecherry
Rutaceae	*Citrus* L.	citrus
Salicaceae	*Salix* L.	willow

Table 68: *Sparganothis sulfureana* host plants

Family	**Genus/species**	**Common name**
Santalaceae	*Comandra umbellata* (L.) Nutt.	bastard toadflax
Scrophulariaceae	*Penstemon* Schmidel	beardtongue
Ulmaceae	*Ulmus americana* L.	American elm
Verbenaceae	*Verbena* L.	vervain
Vitaceae	*Vitis* L.	grape
Vitaceae	*Vitis vinifera* L.	wine grape

Sparganothis tunicana **(Walsingham)**

Nomenclature

Sparganothis tunicana (Walsingham) (Tortricidae: Tortricinae: Sparganothini)

Synonyms: *californiana* (*Dichelia*)

Adult Recognition

Forewings are yellow with brown to purplish-brown markings. The median fascia is complete and the distal half of the wing is solid brown in most individuals. Labial palpi are short in comparison to other Sparganothini. Males lack a forewing costal fold. FWL: 5.5-9.5 mm.

Sparganothis tunicana may be confused with other species of *Sparganothis*. *Sparganothis senecionana* is similar but can be distinguished from *S. tunicana* by its larger size and longer labial palpi.

Biology

Sparganothis tunicana completes a single annual generation over much of its range. Adults are present April to August depending on location.

Life history information for *S. tunicana* is assumed to be similar to that of *S. senecionana*. In California, larvae hatch from the egg, construct a silk hibernaculum, and enter an obligate diapause that lasts through the dry season. Activity is resumed again at the onset of winter rains. Larvae feed on flowers (Powell and Opler 2009).

Table 69: *Sparganothis tunicana* host plants

Family	**Genus/species**	**Common name**
Apiaceae	*Lomatium nudicaule* (Pursh) J.M. Coult. & Rose	barestem biscuitroot
Apiaceae	*Lomatium utriculatum* (Nutt. ex Torr. & A. Gray) J.M. Coult. & Rose	common lomatium
Asteraceae	*Artemisia douglasiana* Besser	Douglas' sagewort
Asteraceae	*Baccharis* L.	baccharis
Asteraceae	*Balsamorhiza sagittata* (Pursh) Nutt.	arrowleaf balsamroot
Asteraceae	*Crepis intermedia* A. Gray	limestone hawksbeard
Asteraceae	*Eriophyllum lanatum* (Pursh) Forbes	common woolly sunflower
Asteraceae	*Microseris nutans* (Hook.) Sch. Bip.	nodding microseris
Asteraceae	*Senecio aronicoides* DC.	rayless ragwort
Asteraceae	*Senecio integerrimus* Nutt.	lambstongue ragwort
Asteraceae	*Tragopogon lamottei* Rouy	Jack-go-to-bed-at-noon
Berberidaceae	*Mahonia repens* (Lindl.) G. Don	creeping barberry
Boraginaceae	*Lithospermum ruderale* Douglas ex Lehm.	western stoneseed
Fabaceae	*Trifolium hybridum* L.	alsike clover
Fabaceae	*Trifolium pratense* L.	red clover
Fagaceae	*Quercus durata* Jeps.	leather oak
Hydrophyllaceae	*Phacelia heterophylla* Pursh	varileaf phacelia
Lamiaceae	*Salvia dorrii* (Kellogg) Abrams	purple sage
Pinaceae	*Pinus ponderosa* C. Lawson	ponderosa pine
Rosaceae	*Prunus* L.	[unspecified]
Rosaceae	*Rosa* L.	rose
Salicaceae	*Populus* L.	cottonwood
Scrophulariaceae	*Castilleja* Mutis ex L. f.	Indian paintbrush
Scrophulariaceae	*Penstemon deustus* Douglas ex Lindl.	scabland penstemon
Scrophulariaceae	*Penstemon laetus* A. Gray	mountain blue penstemon

Host plants

Larvae of *S. tunicana* are polyphagous have been recorded feeding on plants in more than 10 families (Table 69) (Lambert 1950, Powell and Opler 2009).

Distribution

Sparganothis tunicana is recorded from California, Washington, Oregon, Utah, Idaho, and British Columbia (Lambert 1950, Powell and Opler 2009). A single record from North Carolina is questionable.

Tortricidae: Tortricinae: Euliini

Proeulia and related species

Nomenclature

Accuminulia Brown (Tortricidae: Tortricinae: Euliini)

  A. buscki Brown

Chileulia Powell (Tortricidae: Tortricinae: Euliini)

  C. stalactitis (Meyrick)

Proeulia Clarke (Tortricidae: Tortricinae: Euliini)

  P. apospasta Obraztsov

  P. auraria (Clarke)

  P. chrysopteris (Butler)

  P. triquetra Obraztsov

Overview

The Chilean Tortricidae fauna is unique in that the majority of taxa are endemic. Members of the tribe Euliini are the most abundant, comprising nearly 90% of the described tortricid fauna (Razowski and Pelz 2010). Included in this tribe are several genera containing species of economic concern: *Proeulia*, with 38 described species; *Accuminulia*, with two described species; and *Chileulia* with two described species (Razowski and Pelz 2010). Species in these genera have been reported feeding on *Citrus* (orange, tangerine), *Malus* (apple), *Prunus* (apricot, cherry, peach, plum), *Pyrus* (pear), and *Vitis* (grape), and some are considered serious pests in orchards and vineyards (Biosecurity Australia 2003, Brown and Passoa 1998, Epeda and Cubillos 2011). Razowski and Pelz (2010) provide a complete list of tortricids recorded from Chile and describe numerous new species.

Chilean tortricids of greatest concern to the U.S. are those that are pests of table grapes. The U.S. imports nearly twice as many grapes as it exports; imported grapes totaled approximately $730 million in 2004. A majority of the fresh grapes imported to the U.S. are used as table grapes, and imports are necessary to maintain a supply of table grapes during the months of January through June. Chile is the largest exporter of grapes to the U.S., accounting for 70% of total fresh grape imports ($511 million in 2004); Mexico accounts for 28% of fresh grape imports (Boriss et al. 2006).

It is not known how many Chilean tortricid species are intercepted in products destined for the U.S. The USDA interception database has less than 10 records of "*Proeulia* sp." and no records of *Accuminulia* or *Chileulia*; however, there are more than 1000 entries of larvae identified as "Tortricinae" and close to 3000 identified as "Tortricidae" from locations where *Proeulia* and related genera occur (USDA PestID database; Query performed 10/13/2011). As the larvae of these genera are not well-studied or described, it is assumed that interceptions of these larvae have been largely unrecognized.

Because of the difficulties in identification and the lack of species-level larval characters, *Proeulia* and related pest species are treated here as a single entity. This group includes: *Accuminulia buscki*, *Chileulia stalactitis*, *Proeulia apospasta*, *Proeulia auraria*, *Proeulia chrysopteris*, and *Proeulia triquetra*.

Adult Recognition

Accuminulia

Adults are white, pale tan, or gray with dark gray, brown, and black markings. Males lack a forewing costal fold. FWL: 6.0-7.0 mm. Male genitalia are characterized by the following features: aedeagus with distal thornlike projection; transtilla densely spined; gnathos with distal portion enlarged, triangular, and ventrally spined; and valvae with constant width (parallel-sided). Female genitalia are characterized by a partially twisted ductus bursae and a lack of signum in the corpus bursae (Brown 1999).

Chileulia

Forewings are gray or brown with a dark gray to black costal triangle. FWL: 6.5-8.0 mm. Male genitalia are characterized by a short thin uncus, small broad socii, long narrow valvae, and a well-developed sacculus. Female genitalia are characterized by a bowl-shaped sterigma and a long thin ductus bursae (Powell 1986).

Proeulia

Forewing color is variable within the genus and ranges from pale tan to orange to dark brown to white. Forewing markings are generally sparse to moderate, and several species are nearly unmarked. Males lack a forewing costal fold. FWL: 7.0-12.0 mm. Male genitalia are characterized by the following features: uncus slender, varying from long to short; socii well-developed, varying from long to short, densely setose in some species; valva with well-developed sacculus, extending beyond neck of valva in some species; vesica with few stout cornuti. Female genitalia are characterized by a short, wide ductus bursae and a sclerotized process projecting from the ventral surface of the corpus bursae (Razowski and Pelz 2010).

Larval Morphology

Mature larvae of this group are similar to those of other Euliini and few species-specific characters have been identified. Horak and Brown (1991) list the following diagnostic characters for Euliini larvae: D1 and SD1 setae on separate pinacula on A9; SV setal counts on A1, 2, 7, 8, 9 as 3:3:3:2:2; and V1 setae on A9 not further apart than those on A8. These characters apply to the species treated here with some exceptions: the D1 and SD1 setae on A9 are located on the same pinaculum in some (but not all) *Proeulia*; and SV setal counts on A1, 2, 7, 8, 9 are usually 3:3:2:2:2 (but occasionally 3:3:3:2:2) in *Chileulia*. Other diagnostic features of this group include: SD2 seta small and on different pinaculum than SD1 seta on A1-7; SD2 seta on anteroventral margin of SD1 pinaculum on A8; SD1 pinaculum directly anterior of spiracle on A8; D2 setae on A9 on large shared (saddle) pinaculum; L pinaculum on A9 trisetose; and anal comb present (Brown 1999, Cepeda and Cubillos 2011). General descriptions for *Chileulia* and *Proeulia* larvae are presented below; the larva of *Accuminulia* is undescribed.

Chileulia stalactitis

Late instar larvae are approximately 16 mm long with a light green abdomen. The head is light brown and the prothoracic shield is light green with brown mottling. An anal comb is present with 4-6 teeth. Abdominal spiracles are closely approximate to the SD1 pinacula and are circular, surrounded by a conspicuous smooth ring (Cepeda and Cubillos 2011).

Proeulia

Late instar larvae are approximately 22-26 mm long with a light-green abdomen. The head is light brown and may be shaded laterally or posterolaterally in some species. The prothoracic shield is light brown to light green with brown mottling in some species. An anal comb is present with 6-9 (usually 7) teeth (Cepeda and Cubillos 2011).

Biology

Species in this group all share relatively similar life histories. Larvae are highly polyphagous (Table 70) and feed on leaves as well as on fruit, sometimes causing serious economic damage to crops such as grape and citrus. The life histories of *Chileulia* and *Proeulia* are outlined below; the biology of *Accuminulia* is unknown. Life history information is summarized from Biosecurity Australia (2003), Brown and Passoa (1998), Cepeda and Cubillos (2011), and Razowski and Pelz (2010).

Larvae of this group are highly polyphagous and have been reported feeding on plants in more than 30 families. All species listed here are primary or secondary pests of grape. *Chileulia* stalactitis is an important pest of *Prunus* while *Proeulia* are reported as pests of apple, apricot, blueberry, cherry, citrus, peach, plum, and various other species.

Chileulia stalactitis

Females lay eggs in masses on leaves. Larvae feed on leaves, flowers, and fruit, causing direct economic damage by tunneling inside fruit. Overwintering occurs as a larva inside dried fruit. Adults are present in August in Chile.

Proeulia

Females lay eggs in masses on leaves. Larvae feed on leaves, flowers, buds, and on the surface of fruit. Some species have continuous generations and are present throughout the year while others overwinter as a first instar larva. Adults are present September through April for most species.

Table 70: *Proeulia* and related species host plants

Family	**Genus/species**	**Common name**	**Tortricid species**
Aceraceae	*Acer buergerianum* Miq.	trident maple	*P. chrysopteris*
Aceraceae	*Acer pseudoplatanus* L.	sycamore maple	*P. chrysopteris*
Actinidiaceae	*Actinidia deliciosa* (A. Chev.) C. F. Liang & A. R. Ferguson		*P. auraria, P. chrysopteris*
Aristolochiaceae	*Aristolochia chilensis* Bridges ex Lindl.		*P. auraria, P. chrysopteris*
Berberidaceae	*Berberis* L.	barberry	*P. chrysopteris*
Betulaceae	*Corylus avellana* L.	common filbert	*P. chrysopteris*
Betulaceae	*Nothofagus obliqua* (Birb.) Blume	roble beech	*P. auraria*
Buddlejaceae	*Buddleja davidii* Franch.	orange eye butterflybush	*P. auraria, P. triquetra*
Buddlejaceae	*Buddleja globosa* Hope	orange-ball-tree	*P. chrysopteris*
Caprifoliaceae	*Lonicera japonica* Thunb.	Japanese honeysuckle	*P. chrysopteris, P. triquetra*
Caprifoliaceae	*Viburnum* L.	viburnum	*P. chrysopteris*
Cardiopteridaceae	*Citronella mucronata* (Ruiz et Pavon) D. Don.	Chilean citronella tree	*P. chrysopteris*
Celastraceae	*Euonymus* L.	spindletree	*P. chrysopteris*
Celastraceae	*Maytenus boaria* Molina	mayten	*P. triquetra*
Convolvulaceae	*Convolvulus arvensis* L.	field bindweed	*P. triquetra*
Cupressaceae	*Austrocedrus chilensis* (D. Don) Pic. Serm. & Bizzarri	Chilean cedar	*C. stalactitis*
Ebenaceae	*Diospyros* L.	diospyros	*P. chrysopteris*
Ericaceae	*Vaccinium* L.	blueberry	*P. chrysopteris, P. triquetra*
Fabaceae	*Caesalpinia paraguariensis* (D. Parodi) Burkart	guayacan	*P. chrysopteris*
Fabaceae	*Cercis siliquastrum* L.	Judas-tree	*P. auraria*
Fabaceae	*Prosopis tamarugo* F. Philippi	tamarugo	*C. stalactitis*
Fabaceae	*Robinia pseudoacacia* L.	black locust	*P. auraria*
Juglandaceae	*Juglans regia* L.	English walnut	*P. auraria, P. chrysopteris*
Lauraceae	*Cryptocarya* R. Br.	cryptocarya	*P. auraria, P. chrysopteris*
Myrtaceae	[unspecified]		*P. auraria*

Table 70: *Proeulia* and related species host plants

Family	**Genus/species**	**Common name**	**Tortricid species**
Myrtaceae	*Eugenia apiculata* DC.	shortleaf stopper	*P. chrysopteris*
Oleaceae	*Ligustrum* L.	privet	*P. chrysopteris*
Onagraceae	*Fuschia magellanica* Lam.	fuchsia	*P. auraria,* *P. chrysopteris,* *P. triquetra*
Pinaceae	*Pinus* L.	pine	*P. chrysopteris*
Pinaceae	*Pinus radiata* D. Don	Monterey pine	*P. chrysopteris*
Pittosporaceae	*Pittosporum tobira* (Thunb.) W. T. Aiton	Japanese cheesewood	*P. auraria*
Platanaceae	*Platanus orientalis* L.	Oriental planetree	*P. auraria, P. chrysopteris*
Punicaceae	*Punica granatum* L.	pomegranate	*P. auraria*
Rosaceae	*Cotoneaster* Medik.	cotoneaster	*C. stalactitis, P. auraria,* *P. chrysopteris*
Rosaceae	*Eriobotrya japonica* (Thunb.) Lindl.	loquat	*P. chrysopteris*
Rosaceae	*Malus domestica* Borkh.	apple	*P. auraria,* *P. chrysopteris,* *P. triquetra*
Rosaceae	*Prunus armeniaca* L.	apricot	*A. buscki, P. auraria,* *P. chrysopteris*
Rosaceae	*Prunus avium* (L.) L.	sweet cherry	*P. auraria*
Rosaceae	*Prunus cerasifera* Ehrh.	cherry plum	*P. chrysopteris,* *P. triquetra*
Rosaceae	*Prunus cerasus* L.	sour cherry	*C. stalactitis*
Rosaceae	*Prunus domestica* L.	European plum	*A. buscki, C. stalactitis,* *P. auraria, P. chrysopteris*
Rosaceae	*Prunus persica* (L.) Batsch	peach	*A. buscki, P. chrysopteris*
Rosaceae	*Pyrus communis* L.	common pear	*P. auraria, P. chrysopteris*
Rosaceae	*Quillaja saponaria* Molina	soapbark	*P. auraria*
Rosaceae	*Rosa* L.	rose	*P. auraria, P. chrysopteris*
Rosaceae	*Rubus idaeus* L.	American red raspberry	*Proeulia* sp.
Rosaceae	*Rubus* L.	blackberry	*Proeulia* sp.
Rosaceae	*Rubus occidentalis* L.	black raspberry	*P. triquetra*
Rosaceae	*Rubus ulmifolius* Schott	elmleaf blackberry	*Proeulia* sp.
Rutaceae	*Citrus reticulata* Blanco	tangerine	*P. triquetra*
Rutaceae	*Citrus* X *sinensis* (L.) Osbeck (pro sp.) [*maxima* X *reticulata*]	sweet orange	*C. stalactitis, P. auraria,* *P. chrysopteris*
Salicaceae	*Salix* X *sepulcralis* Simonkai [*alba* X ?*pendulina*]	weeping willow	*P. chrysopteris*
Santalaceae	*Myoschilos oblonga* Ruiz & Pav.		*P. chrysopteris,* *P. triquetra*
Scrophulariaceae	*Hebe* Comm. ex Juss.	hebe	*P. triquetra*
Simmondsiaceae	*Simmondsia chinensis* (Link) K. C. Schneid.	jojoba	*P. auraria, P. chrysopteris*

Table 70: *Proeulia* and related species host plants

Family	**Genus/species**	**Common name**	**Tortricid species**
Ulmaceae	*Ulmus* L.	elm	*P. chrysopteris*
Vitaceae	*Vitis* L.	grape	*A. buscki, P. chrysopteris*
Vitaceae	*Vitis vinifera* L.	wine grape	*C. stalactitis,* *P. apospasta, P. auraria,* *P. triquetra*

Distribution

Accuminulia and *Chileulia* are known exclusively from Chile. The majority of *Proeulia* are also recorded only from Chile, with the exception of one species that is found in Bolivia Razowski and Pelz (2010).

Seticosta rubicola Brown & Nishida

Nomenclature

Seticosta rubicola Brown & Nishida (Tortricidae: Tortricinae: Euliini)

Adult Recognition

Adults are tan, brown, or reddish brown with white and yellow-green markings. Labial palpi are extremely elongate in both sexes. Males have long cilia on the antennae, a hairpencil on the foreleg, and lack a forewing costal fold. FWL: 5.0-8.5 mm.

Seticosta rubicola is similar in forewing pattern to other species of *Seticosta*, including *S. aeolozona*, *S. arachnogramma*, *S. tridens*, and *S. tambomachaya*. Male *S. rubicola* can be distinguished from these other species by a pair of lateral processes near the distal end of the uncus and the lack of a dense patch of setae on the costa of the valva (Brown and Nishida 2003).

Larval Morphology

Last instar larvae are approximately 12-13 mm in length. The abdomen is brownish purple with dark, large, conspicuous pinacula. The head is black to orange with a dark lateral band and the prothoracic shield is brownish yellow to reddish brown with pale-brown mottling.

Larvae are distinguished by a combination of tortricine and olethreutine characters: SD1 and D1 on a shared pinaculum on A9; bisetose SV-group on A7; SD2 and SD1 on separate pinacula on A1-8; V setae on A9 widely separated; meso- and metathorax with extra SD, L, SV, and V setae; A1-8 with extra D and V setae; D pinacula on A2-5 notched; and L pinaculum on prothorax ventrad of the spiracle (Brown and Nishida 2003).

Biology

Adults of *Seticosta rubicola* have been collected in nearly every month in central Costa Rica, suggesting continuous generations throughout the year.

Larvae bore into stems and induce gall formation. Galls are fusiform, approximately 4-6 mm wide, 12-40 mm long, and are located between nodes on the terminal (young) portions of the stem. One gall contains a single larva. An opening at the base of the gall is often covered in frass and debris; this entrance hole can be detected before the stem begins to swell. Pupation most likely occurs outside of the gall (Brown and Nishida 2003).

Seticosta rubicola feeds on *Rubus* (Table 71) and is considered a serious pest of blackberry in parts of Costa Rica. Larvae have been intercepted on *Rubus* arriving at U.S. ports-of-entry from Guatemala (Brown and Nishida 2003).

Table 71: *Seticosta rubicola* host plants

Family	**Genus/species**	**Common name**
Rosaceae	*Rubus eriocarpus* Liebm.	mora (blackberry)
Rosaceae	*Rubus vulcanicola* (Donn. Sm.) Rydb.	
Rosaceae	*Rubus* L.	[various]

Distribution

Seticosta rubicola has been reported from Costa Rica and Guatemala (Brown and Nishida 2003).

Tortricidae: Olethreutinae: Microcorsini

Cryptaspasma **Walsingham**

Overview

The genus *Cryptaspasma* is pan-tropical and comprised of 34 described species. It has been recorded from Central America, South America, southern North America, Africa, Madagascar, Australia, New Zealand, New Caledonia, the Orient, and the eastern Palearctic (Brown and Brown 2004). The genus has traditionally been considered the sole representative of the tribe Microcorsini; however, the Australian genus *Collogenes* was recently transferred to the Microcorsini by Horak (2006). Microcorsini are hypothesized to be the most basal group in Olethreutinae (Diakonoff 1959). The genus is currently divided into seven subgenera based on geographic distribution and structures of the male genitalia; Aarvik (2005) provided a key to the subgenera.

Adults are dark brown with subdued patterns that are relatively uniform across the entire genus. Males have an array of various sex scales on the legs, hindwing, and end of the abdomen; the presence and form of the sex scales is sometimes helpful in separating species. Male and female genitalia are also uniform within most subgenera, making species recognition difficult. Several undescribed species are present in Central America, South America, and Australasia.

The two species are treated here feed on avocado and other Lauraceae. *Cryptaspasma bipenicilla* is a native of the Caribbean, Central America, and the southeastern U.S.; it has not been reported causing damage to commercially grown avocado (Brown and Brown 2004). *Cryptaspasma perseana* is a native of Central America that is a pest of avocado in Mexico and

Guatemala (Gilligan et al. 2011). Larvae tunnel into the hard seed, damaging the pulp and potentially inducing early fruit drop.

***Cryptaspasma* (*Cryptaspasma*) *bipenicilla* Brown and Brown**

Nomenclature

Cryptaspasma (*Cryptaspasma*) *bipenicilla* Brown and Brown (Tortricidae: Olethreutinae: Microcorsini)

Adult Recognition

Males are grayish brown with faint black reticulated markings. Females are uniformly darker with fewer markings. Both males and females have a conspicuous white dot on the distal one-third of the forewing. Hindwings are pale grayish brown. FWL: 6.5-11.5 mm.

Male genitalia are characterized by an elongate tapering uncus, densely setose socii, and subtriangular valvae. Female genitalia are characterized by broad papillae anales and two large signa in the corpus bursae. Males have two large patches of coremata on the posterior margin of segment VIII that are paired in two lengths (Brown and Brown 2004).

The subgenus *Cryptaspasma* contains a total of six species, including *C. bipenicilla*, all of which are difficult to separate. *Cryptaspasma bipenicilla* can be separated from most described Neotropical *Cryptaspasma* (subgenus *Cryptaspasma*) by the two lengths of coremata on the male abdomen. Other undescribed species from Central and South America have two lengths of coremata but may be separated using a combination of other characters.

Larval Morphology

The larva of *C. bipenicilla* has not been described. Larvae of other species of *Cryptaspasma* are orange to pink with a yellowish brown head and prothoracic shield. Pinacula are weakly sclerotized. Diagnostic characters include: D2 setae on A8 on the same "saddle" pinaculum; D1, D2, and SD1 setae on A9 all on the same large "saddle" pinaculum; L pinaculum on A9 bisetose; and anal comb absent (Gilligan et al. 2011).

Biology

Life history information for *C. bipenicilla* is undocumented, although it is assumed to be similar to that of other avocado-feeding *Cryptaspasma*, such as *C. perseana*.

Host plants

Cryptaspasma bipenicilla has been recorded feeding on seeds of avocado and slugwood (Lauraceae) in Puerto Rico and Cuba (Table 72). Potential lauraceous hosts in the southeastern U.S. include *Persea borbonia* (L.) (redbay) and *Persea palustris* Sarg. (swamp bay).

Table 72: *Cryptaspasma bipenicilla* host plants

Family	**Genus/species**	**Common name**
Lauraceae	*Beilschmiedia pendula* (Sw.) Hemsl.	slugwood
Lauraceae	*Persea americana* Mill.	avocado

Distribution

Cryptaspasma bipenicilla has been reported from the southeastern United States (North Carolina to Texas), the Caribbean, and Central America (Brown and Brown 2004).

***Cryptaspasma* (*Cryptaspasma*) *perseana* Gilligan and Brown**

Refer to Chapter 6 for information on *Cryptaspasma perseana*.

Tortricidae: Olethreutinae: Bactrini

***Endothenia hebesana* (Walker)**

Nomenclature

Endothenia hebesana (Walker) (Tortricidae: Olethreutinae: Bactrini)

Common names: verbena bud moth

Synonyms: *adjustana* (*Endothenia*), *daeckeana* (*Olethreutes*), *fillerea* (*Endothenia*), *fullerea* (*Penthina*), *inexpectana* (*Endothenia*), *inexpertana* (*Carpocapsa*), *kiyosatoensi* (*Endothenia*), *torquata* (*Argyroploce*), *toteniana* (*Endothenia*)

Adult Recognition

Fresh specimens have a bluish-gray cast to the forewings with varying orange-brown scaling. Three gray spots edged with black are present near the termen. Worn specimens may be difficult to distinguish from similar species without a genitalic dissection. Males lack a forewing costal fold. FWL: 4.5-8.5 mm.

Some species of *Paralobesia* species resemble *E. hebesana* in wing coloration and pattern. Worn specimens of *E. hebesana* may resemble a variety of North American olethreutines, and a genitalic dissection may be necessary to confirm identity.

Larval Morphology

Late instar larvae are approximately 10 mm in length. The abdomen varies in color, but many individuals are yellowish or reddish with inconspicuous pinacula. The head and prothoracic shield are black. An anal comb is present with 4-6 teeth (MacKay 1959).

Larvae are morphologically similar to those of *Lobesia* and *Paralobesia*, including *L. botrana* and *P. viteana*. The D2 setae on A9 are on a joined "saddle" pinaculum, the D1 and SD1 setae on A9 are on a single pinaculum, and the SV group on A7 is trisetose (SV counts 3:3:3:2:2) (Gilligan et al. 2008). However, it unlikely that larvae of *E. hebesana* would be encountered on the same hosts as *L. botrana* or *P. viteana*.

Table 73: *Endothenia hebesana* host plants

Family	**Genus/species**	**Common name**
Anacardiaceae	*Toxicodendron diversilobum* (Torr. & A. Gray) Greene	Pacific poison oak
Asteraceae	*Solidago* L.	goldenrod
Betulaceae	*Ostrya* Scop.	hophornbeam
Gentianaceae	*Frasera caroliniensis* Walter	American columbo
Gentianaceae	*Gentiana saponaria* L.	harvestbells

Table 73: *Endothenia hebesana* host plants

Family	**Genus/species**	**Common name**
Gentianaceae	*Gentianopsis crinita* (Froel.) Ma	greater fringed gentian
Iridaceae	*Iris* L.	iris
Iridaceae	*Tigridia* Juss.	peacock flower
Lamiaceae	*Physostegia* Benth.	lionsheart
Lamiaceae	*Physostegia virginiana* (L.) Benth.	obedient plant
Lamiaceae	*Scutellaria* L.	skullcap
Lamiaceae	*Scutellaria lateriflora* L.	blue skullcap
Lamiaceae	*Stachys* L.	hedgenettle
Lamiaceae	*Teucrium canadense* L.	Canada germander
Ranunculaceae	*Delphinium* L.	larkspur
Sarraceniaceae	*Sarracenia* L.	pitcherplant
Sarraceniaceae	*Sarracenia purpurea* L.	purple pitcherplant
Scrophulariaceae	*Antirrhinum* L.	snapdragon
Scrophulariaceae	*Aureolaria flava* (L.) Farw.	smooth yellow false foxglove
Scrophulariaceae	*Aureolaria* Raf.	false foxglove
Scrophulariaceae	*Castilleja affinis* Hook. & Arn.	coast Indian paintbrush
Scrophulariaceae	*Castilleja affinis* Hook. & Arn. ssp. *affinis*	coast Indian paintbrush
Scrophulariaceae	*Castilleja latifolia* Hook. & Arn.	Monterey Indian paintbrush
Scrophulariaceae	*Castilleja* Mutis ex L. f.	Indian paintbrush
Scrophulariaceae	*Orthocarpus* Nutt.	owl's-clover
Scrophulariaceae	*Pedicularis canadensis* L.	Canadian lousewort
Scrophulariaceae	*Penstemon* Schmidel	beardtongue
Scrophulariaceae	*Verbascum* L.	mullein
Verbenaceae	*Verbena* L.	vervain

Larvae develop in the seeds of their host plants. Adults are present in mid-April through September, many times in large numbers. Multiple generations are produced in southern locations. Larvae of this species have been recorded feeding on members of at least eight plant families (Table 73). In some areas it is considered a minor pest of ornamentals (Gilligan et al. 2008).

Distribution

Endothenia hebesana is present in eastern North America, Texas, and the Pacific coast. It is also recorded from China, Finland, Japan, Norway, and Russia (Gilligan et al. 2008, Razowski 2003).

Tortricidae: Olethreutinae: Olethreutini

Lobesia botrana ([Denis & Schiffermüller])

Refer to Chapter 4 for information on *Lobesia botrana*.

Paralobesia Obraztsov

Overview

The genus *Paralobesia* contains 18 described, and numerous undescribed species (Gilligan et al. 2008). All but one are Nearctic; Razowski (2003) placed the Palearctic *P. andereggiana* in *Lobesia* but it appears that this species should remain in *Paralobesia* based on his drawing of the male genitalia. Moths in this genus are difficult to identify because of the various undescribed species and the lack of understanding of described species. Gilligan et al. (2008) detail the taxonomic history of this group and provide descriptions for many of the eastern North American species.

Adults are small (FWL 4.5-8.0 mm) and many species are leaden-gray with reddish brown fasciae. Wing pattern is not diagnostic for many species, and a genitalic dissection is necessary to confirm identity. Male genitalia are characterized by a cluster of long setae on the ventral margin of the sacculus, referred to as the saccular tuft, and a U-shaped indentation of the ventral margin of the valval neck, often with facing padlike projections. Female genitalia are characterized by a cone-shaped sterigma and two accessory sacs on the anterior end of the corpus bursae in some species.

The most familiar member of this genus is the grape berry moth, *P. viteana*. This species is a pest of grape in eastern North America, and larvae cause direct and indirect damage by feeding on grape berries. Adults can be confused with those of *L. botrana*, which was discovered in California in 2008-2009. Adults of *P. viteana* and *L. botrana* cannot be separated by wing pattern; a genitalic dissection is necessary to confirm identity. The two species are easily separated using male or female genitalia.

While most Paralobesia species appear to be restricted to eastern North America, at least three are present on the West Coast. *Paralobesia palliolana* and two undescribed species have been recorded from Washington, Oregon, and California; Gilligan et al. (2011b) detail these discoveries.

***Paralobesia viteana* (Clemens)**

Nomenclature

Paralobesia viteana (Clemens) (Tortricidae: Olethreutinae: Olethreutini)

Common names: grape berry moth

Synonyms: *vitivorana* (*Penthina*)

Adult Recognition

Forewing pattern is similar to other Nearctic species of *Paralobesia* and *Lobesia botrana*. *Paralobesia viteana* can be separated from most other tortricids by the sclerotized lobe projecting off the base of the cucullus in the male genitalia. Females lack signa in the corpus bursae and males lack a forewing costal fold. FWL: 4.5-6.0 mm.

Adults can be confused with those of *Lobesia botrana*, which was discovered in California in 2008-2009. Adults of *P. viteana* and *L. botrana* cannot be separated by wing pattern; a genitalic dissection is necessary to confirm identity (Gilligan et al. 2011b).

Larval Morphology

Late instar larvae are approximately 10-15 mm long with a yellowish-green to pale-brown abdomen. The head and prothoracic shield are yellowish brown and the shield is variably shaded with dark brown to black on the posterior and lateral margins. An anal comb is present with 5-8 teeth (MacKay 1959). Chaetotaxy is the same as that of *Lobesia botrana*.

No morphological characters have been identified to reliably separate the larvae of *Paralobesia* and *Lobesia*. Should *P. viteana* be introduced to the West Coast, or *L. botrana* expand out of California, molecular diagnostics may be required to identify larvae of *Paralobesia*/*Lobesia* found on grape (Gilligan et al. 2011b).

Biology

The following life history information is summarized from Botero-Garces and Isaacs (2003) and Isaacs et al. (2005). *Paralobesia viteana* completes 2-4 generations per year, depending on location. Adults of the various generations are present March to August.

Females lay eggs on individual grapes, blossoms, and stems. Larvae of the first generation feed on blossoms or small berries, often webbing together entire clusters. Larvae of subsequent generations tunnel into the berries and feed inside, also webbing together clusters. Mature larvae exit the clusters and pupate in a cresent-shaped fold cut into a leaf. Larvae of the last generation may also drop to the ground and pupate in leaf litter. Overwintering occurs in the pupal stage.

Grape (*Vitis* sp.) is the preferred larval host, although several secondary hosts have been documented (Table 74). This species is the primary lepidopteran pest of grapes in eastern North America (Botero-Garces and Isaacs 2003, Witzgall et al. 2000)

Distribution

Paralobesia viteana is widespread in grape-growing regions of eastern North America. It has also been recently recorded from vineyards in western Colorado. This species has not been reported from California or the grape-growing regions of the Pacific Northwest.

Table 74: *Paralobesia viteana* host plants

Family	Genus/species	Common name
Fabaceae	*Amorpha* L.	false indigo
Lauraceae	*Sassafras* Nees & Eberm.	sassafras
Rosaceae	*Rubus* L.	blackberry
Vitaceae	*Vitis* L.	grape
Vitaceae	*Vitis riparia* Michx.	riverbank grape

Tortricidae: Olethreutinae: Eucosmini

Crocidosema Zeller

Overview

The genus *Crocidosema* contains approximately 30 described species (Baixeras et al. 2008). Most species diversity occurs in the Neotropics. Adults are brown or gray and wing patterns are typical of many Eucosmini, with numerous white costal strigulae and a well-defined ocellus. Male genitalia are characterized by large socii, a short uncus, and variably shaped valvae. Female genitalia are characterized by a pair of pockets or lobes on sternum VII and two signa in the corpus bursae.

One species, *C. plebejana*, is cosmopolitan and occurs in Europe, northern Africa, Asia, Australia, North America, Central America, and South America (Razowski 2003). Larvae feed primarily on plants in the Malvaceae, and in some areas are pests of cotton. *Crocidosema aporema* is distributed from south Texas through Central and South America south to Argentina and Chile. It is a major pest of beans (*Glycine max* and *Phaseolus*) in Central and South America (Arneodo et al. 2010), but has not been reported to cause economic damage in the U.S. Larvae of *C. aporema* are frequently intercepted at U.S. ports of entry on beans originating from Central America, South America, or the Carribean.

Crocidosema aporema **(Walsingham)**

Nomenclature

Crocidosema aporema (Walsingham) (Tortricidae: Olethreutinae: Eucosmini)

Common names: bean shoot moth

Synonyms: *opposita* (*Epinotia*)

Adult Recognition

Adults are sexually dimorphic. The male forewing is mostly brown to reddish brown with a pale dorsum and large costal patch. The female forewing is mostly pale with a dark patch on the dorsum. Males have several secondary sexual structures including a forewing costal fold and black scaling on the hindwing. FWL: 6.5-8.0 mm.

Male genitalia are characterized by a short triangular uncus, large triangular socii, and a parallel-sided cucullus. Female genitalia are characterized by a pair of rounded pockets on sternum VII and a large rounded corpus bursae with two signa.

Other species of *Crocidosema*, including the cosmopolitan *C. plebejana*, may appear similar. Most individuals of *C. plebejana* have a distinctive white ocellus and white patch on the dorsum of the forewing. The male cucullus is enlarged and subtriangular, and the pockets on the female sternum VII are developed into two lobes or "flaps" in *C. plebejana*.

Larval Morphology

Late instar larvae are approximately 10 mm in length with a green to yellow abdomen. The head is black in the first four instars, becoming light brown to reddish brown in the final instar (Arneodo et al. 2010).

Larval damage on beans is similar to that caused by *Cydia fabivora*; however, larvae of *C. aporema* feed primarily on young leaflets while those of *C. fabivora* feed in the stems and pods (Pereyra and Sanchez 1998). An anal comb is present in *C. aporema* but is absent in *C. fabivora*. Other bean-feeding tortricids in South and Central America include *Amorbia* spp., *Cydia torostoma*, *Lusterala phaseolana*, and *Platynota* spp.

Biology

The following life history information is summarized from Altesor et al. (2010), Arneodo et al. (2010), and Pereyra and Sanchez (1998). *Crocidosema aporema* completes 4-6 generations per year, with at least two generations occuring on soybean. Adults can be present year-round and are most common between April and November in South America.

Females lay eggs on glabrous nodes of the soybean plant or other leguminous host. Early instar larvae feed primarily on terminal buds, folding or rolling the young leaflets. Later instars may tunnel into stems, floral buds, and pods. Pupation occurs in the soil or in rolled leaves. Larval feeding leads to reduced plant height, drying of terminal shoots, a decrease in lower pod insertion, and damage to flowers, all of which can result in reduced crop yields.

Crocidosema aporema is a major pest of soybean (*Glycine max*) and cultivated beans (*Phaseolus* spp.) in Central and South America. While larvae feed primarily on Fabaceae, they have also been recorded on cotton and corn (Table 75).

Table 75: *Crocidosema aporema* host plants

Family	**Genus/species**	**Common name**
Fabaceae	*Cicer arietinum* L.	chick pea
Fabaceae	*Erythrina crista-galli* L.	crybabytree
Fabaceae	*Glycine max* (L.) Merr.	soybean
Fabaceae	*Glycyrrhiza glabra* L.	cultivated licorice
Fabaceae	*Lens culinaris* Medik.	lentil
Fabaceae	*Medicago* L.	alfalfa
Fabaceae	*Medicago sativa* L.	alfalfa
Fabaceae	*Melilotus* Mill.	sweetclover
Fabaceae	*Phaseolus coccineus* L.	scarlet runner
Fabaceae	*Phaseolus vulgaris* L.	kidney bean
Fabaceae	*Vigna luteola* (Jacq.) Benth.	hairypod cowpea
Fabaceae	*Vigna* Savi	cowpea
Fabaceae	*Vigna unguiculata* (L.) Walp.	blackeyed pea
Fabaceae	*Vigna unguiculata* (L.) Walp. ssp. *unguiculata*	southern pea
Malvaceae	*Gossypium hirsutum* L.	upland cotton
Poaceae	*Zea mays* L.	corn

Distribution

Crocidosema aporema is distributed from south Texas through Central and South America south to Argentina and Chile (Altesor et al. 2010, Pereyra and Sanchez 1998). It has not been reported causing economic damage in the U.S.

Crocidosema plebejana **Zeller**

Nomenclature

Crocidosema plebejana Zeller (Tortricidae: Olethreutinae: Eucosmini)

Common names: cotton tipworm

Synonyms: *altheana* (*Penthina*), *blackburnii* (*Proteopteryx*), *bostrychodes* (*Crocidosema*), *charmera* (*Eucosma*), *excitana* (*Grapholitha*), *insulana* (*Crocidosema*), *iris* (*Crocidosema*), *lavaterana* (*Paedisca*), *obscura* (*Steganoptycha*), *peregrinana* (*Grapholitha*), *ptiladelpha* (*Crocidosema*), *synneurota* (*Crocidosema*), *tornocycla* (*Eucosma*)

Adult Recognition

Males are dark brown to black with a conspicuous white ocellus and dorsal patch. Females are pale brown to tan with a dark basal patch that does not extend to the costa. Males lack a forewing costal fold. FWL: 5.0-8.0 mm. Male genitalia are characterized by valvae with an enlarged, subtriangular cucullus. Female genitalia are characterized by a pair of lobes or "flaps" projecting from sternum VII and two signa in the corpus bursae.

In the Nearctic, *C. plebejana* may appear similar to other Eucosmini, including other species of *Crocidosema* or *Epinotia*. A genitalic dissection can be used to confirm identity. The bean shoot moth, *Crocidosema aporema*, can be separated from *C. plebejana* by a parallel-sided cucullus in the male and a pair of shallow, rounded pockets on sternum VII in the female.

Larval Morphology

Late instar larvae are approximately 8-10 mm in length with an abdomen that varies in color from whitish to yellowish brown to dark reddish brown. The head is yellowish brown to dark brown with black posterolateral margins. The prothoracic shield is dark yellowish brown. Pinacula are small and body colored. An anal comb is present with 4-6 teeth (MacKay 1959).

Larvae feeding on cotton can cause damage similar to that caused by the pink bollworm, *Pectinophora gossypiella* (Gelechiidae). Pink bollworm larvae can be separated from *C. plebejana* larvae by their pink coloration.

Biology

The following life history information is summarized from Bishop and Blood (1978), Bradley et al. (1979), and Powell and Opler (2009). *Crocidosema plebejana* is not known to diapause; although Bradley et al. (1979) report only 1-2 generations per year in southern England. In temperate regions, continuous generations occur and adults are present year-round.

Larvae are reported as a pest of cotton in Australia, but this species has not reached pest status in the cotton-growing regions of North America. In areas where cotton is a preferred host, other malvaceous plants are utilized during times of the year when cotton is unsuitable or unavailable. The life history information provided here is applicable to cotton as the larval host.

Females lay eggs singly on the upper and lower surfaces of leaves in or close to the terminals and lateral buds. Early instars feed on leaves under loose silk webbing. Later instars move to the terminals and tunnel into stems; they may also feed on leaf petioles and young bolls. Pupation occurs in terminal regions in webbed or tied leaves.

Although members of the Malvaceae are preferred hosts, larvae have also been recorded feeding on plants in the Anacardiaceae, Chenopodiaceae, Fabaceae, Myrtaceae, Poaceae, Rosaceae, Salicaceae, and Turneraceae (Table 76).

Table 76: *Crocidosema plebejana* host plants

Family	**Genus/species**	**Common name**
Anacardiaceae	*Rhus pyroides* Burch.	
Chenopodiaceae	*Atriplex limbata* Benth.	
Fabaceae	*Phaseolus lunatus* L.	sieva bean
Fabaceae	*Senna occidentalis* (L.) Link	septicweed
Malvaceae	[unspecified]	
Malvaceae	*Abelmoschus esculentus* (L.) Moench	okra
Malvaceae	*Abutilon eremitopetalum* Caum	hiddenpetal Indian mallow
Malvaceae	*Abutilon grandifolium* (Willd.) Sweet	hairy Indian mallow
Malvaceae	*Abutilon indicum* (L.) Sweet	monkeybush
Malvaceae	*Abutilon menziesii* Seem.	ko'oloa 'ula
Malvaceae	*Abutilon* Mill.	Indian mallow
Malvaceae	*Abutilon sandwicense* (O. Deg.) Christoph.	greenflower Indian mallow
Malvaceae	*Alcea rosea* L.	hollyhock
Malvaceae	*Althaea* L.	marshmallow
Malvaceae	*Anoda cristata* (L.) Schltdl.	crested anoda
Malvaceae	*Gossypium hirsutum* L.	upland cotton
Malvaceae	*Gossypium* L.	cotton
Malvaceae	*Hibiscus* L.	rosemallow
Malvaceae	*Hibiscus laevis* All.	halberdleaf rosemallow
Malvaceae	*Hibiscus rosa-sinensis* L.	shoeblackplant
Malvaceae	*Kosteletzkya* C. Presl	kosteletzkya
Malvaceae	*Lavatera arborea* L.	tree mallow
Malvaceae	*Lavatera* L.	tree mallow
Malvaceae	*Malacothamnus clementinus* (Munz & I.M. Johnst.) Kearney	San Clemente Island bushmallow
Malvaceae	*Malva* L.	mallow
Malvaceae	*Malva moschata* L.	musk mallow
Malvaceae	*Malva parviflora* L.	cheeseweed mallow
Malvaceae	*Malva sylvestris* L.	high mallow
Malvaceae	*Malvastrum americanum* (L.) Torr.	Indian Valley false mallow
Malvaceae	*Malvaviscus arboreus* Dill. ex Cav. var. *drummondii* (Torr. & A. Gray) Schery	wax mallow
Malvaceae	*Malvaviscus* Fabr.	wax mallow
Malvaceae	*Sida fallax* Walp.	yellow 'ilima
Malvaceae	*Sida* L.	fanpetals
Malvaceae	*Sida rhombifolia* L.	Cuban jute
Myrtaceae	*Eucalyptus* L'Her.	gum
Poaceae	*Triticum aestivum* L.	common wheat
Poaceae	*Zea mays* L.	corn
Rosaceae	*Crataegus* L.	hawthorn

Table 76: *Crocidosema plebejana* host plants

Family	Genus/species	Common name
Salicaceae	*Salix subserrata* Willd.	
Turneraceae	*Turnera ulmifolia* L.	ramgoat dashalong

Distribution

Crocidosema plebejana is a cosmopolitan species that is distributed across southern Europe, northern Africa, Asia, Australia, North America, Central America, and South America (Razowski 2003). In the U.S., it is found primarily in the Southeast, Texas, New Mexico, and California (Powell and Opler 2009).

Spilonota ocellana **([Denis & Schiffermüller])**

Nomenclature

Spilonota ocellana ([Denis & Schiffermüller]) (Tortricidae: Olethreutinae: Eucosmini)

Common names: eye-spotted bud moth

Synonyms: *centralasiae* (ssp.), *comitana* (*Tortrix*), *luscana* (*Pyralis*), *occulana* (*Penthina*), *pyrifoliana* (*Hedya*), *zellerana* (*Tmetocera*)

Adult Recognition

Adults are grayish brown and are found in both light and dark forms. Forewing markings include a wide median fasica which ranges in color from white to cream to gray, a dark-brown mark on the dorsum proximal to the tornus, and a series of small black dashes in the ocellus. In many individuals the median fascia is confluent with the postmedian and preterminal fasciae, creating an overall gray or whitish appearance. Males have a notch at the base of the antenna and lack a forewing costal fold. FWL: 5.5-8.0 mm.

In North America, *S. ocellana* is unlikely to be confused with any other species of tortricid. A genitalic dissection can be used to confirm identity of worn specimens. Males have a distinctively shaped cucullus and females have two thornlike signa.

Larval Morphology

Late instar larvae are approximately 9-14 mm long with a gray to dull reddish-brown abdomen. The head and prothoracic shield are reddish-brown to black, sometimes with dark mottling. Prothoracic legs are dark brown. Other distinguishing features include: abdominal pinacula large and raised; SV counts on A1,2,7,8,9 usually 3:3:2:2:2; L pinaculum on A9 trisetose; and anal comb present with 3-8 teeth (MacKay 1959).

Biology

Spilonota ocellana completes a single generation per year. Adults are present June to August. Females lay eggs singly on leaves. Newly hatched larvae feed primarily on leaves. Third instar larvae construct a hibernaculum, often in a spur crotch, where they overwinter. Feeding resumes in early spring on fruiting buds, leaves, and blossoms, with larvae forming a tubular chamber between leaves or in a rolled leaf. Pupation occurs in a leaf nest near the feeding site. *Spilonota ocellana* is a pest in orchards, with apple (*Malus*) and cherry (*Prunus*) being preferred hosts (Chapman and Lienk 1971). A list of documented host plants is provided in Table 77.

Distribution

Spilonota ocellana is present in all apple growing regions of the northern hemisphere. It was introduced into North America from Europe sometime before 1840 and now occurs across southern Canada and the northern United States, ranging as far south as North Carolina, Ohio, and California (Gilligan et al. 2008).

Table 77: *Spilonota ocellana* host plants

Family	**Genus/species**	**Common name**
Anacardiaceae	*Rhus* L.	sumac
Betulaceae	*Alnus glutinosa* (L.) Gaertn.	European alder
Betulaceae	*Alnus japonica* (Thunb.) Steud.	Japanese alder
Betulaceae	*Alnus* Mill.	alder
Betulaceae	*Betula* L.	birch

Table 77: *Spilonota ocellana* host plants

Family	**Genus/species**	**Common name**
Betulaceae	*Betula pubescens* Ehrh.	downy birch
Betulaceae	*Carpinus betulus* L.	European hornbeam
Elaeagnaceae	*Hippophae rhamnoides* L.	seaberry
Ericaceae	*Kalmia* L.	laurel
Euphorbiaceae	*Euphorbia paralias* L.	sea spurge
Fagaceae	*Quercus garryana* Douglas ex Hook.	Oregon white oak
Fagaceae	*Quercus robur* L.	English oak
Fagaceae	*Quercus* L.	oak
Juglandaceae	*Juglans* L.	walnut
Myricaceae	*Myrica gale* L.	sweetgale
Pinaceae	*Larix kaempferi* (Lamb.) Carriere	Japanese larch
Pinaceae	*Larix* Mill.	larch
Polygonaceae	*Rumex obtusifolius* L.	bitter dock
Rosaceae	*Chaenomeles speciosa* (Sweet) Nakai	flowering quince
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Crataegus rhipidophylla* Gand.	
Rosaceae	*Cydonia* Mill.	cydonia
Rosaceae	*Malus domestica* Borkh.	apple
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Malus sylvestris* (L.) Mill.	European crab apple
Rosaceae	*Prunus* L.	
Rosaceae	*Prunus mume* Siebold & Zucc.	Japanese apricot
Rosaceae	*Prunus pensylvanica* L. f.	pin cherry
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Prunus salicina* Lindl.	Japanese plum
Rosaceae	*Prunus serrulata* var. *spontanea* (Maxim.) E. H. Wilson	
Rosaceae	*Pyracantha* M. Roem.	firethorn
Rosaceae	*Pyrus communis* L.	common pear
Rosaceae	*Pyrus* L.	pear
Rosaceae	*Rubus* L.	blackberry
Rosaceae	*Sorbus aucuparia* L.	European mountain ash
Rosaceae	*Sorbus* L.	mountain ash
Salicaceae	*Salix cinerea* L.	large gray willow
Salicaceae	*Salix* L.	willow

Tortricidae: Olethreutinae: Enarmoniini

***Enarmonia formosana* (Scopoli)**

Nomenclature

Enarmonia formosana (Scopoli) (Tortricidae: Olethreutinae: Enarmoniini)

Common names: cherry bark tortrix

Synonyms: *ornatana* (*Tortrix*), *scriptana* (*Tortrix*), *woeberana* (*Pyralis*), *woeberiana* (*Tortrix*)

Adult Recognition

Forewings are black with distinctive yellow, orange, and silvery markings and a conspicuous ocellus with alternating black and yellow bands. Hindwings are dark grayish brown with white fringe. FWL: 7.0-9.0 mm.

Adults of *E. formosana* have forewing coloration similar to some *Olethreutes* and related genera (*Syricoris*, *Pristerognatha*, etc.), although most of these species lack a defined ocellus. In North America, *Eucosmomorpha nearctica* may appear similar, but adults lack the black forewing ground color. A genitalic dissection can be used to easily separate the various genera listed here.

Enarmonia formosana pheromone traps in Washington have captured a wide variety of tortricines and olethreutines as well as moths in other families (E. LaGasa, pers. comm.). Wing pattern is usually sufficient to separate *E. formosana* from non-target species.

Larval Morphology

Last instar larvae are approximately 8-11 mm in length. The abdomen is whitish to gray to pinkish with darker gray pinacula. The head and prothoracic shield are brown; sometimes the shield is marked with black on the posterolateral margin (Bradley et al. 1979).

Biology

The following life history information is compiled from Bradley et al. (1979), Dang and Parker (1990), Dickler (1991), and Tanigoshi and Stary (2003). *Enarmonia formosana* completes two annual generations. Adults are present from May to September.

Females lay eggs singly or in small clusters on tree bark. Larvae tunnel into the bark and feed under the surface. Older trees are usually more heavily infested; wounded tissue and the base of the tree are favored feeding sites. Larval damage is characterized by "frass tubes" consisting of fecal pellets, silk, and tree sap, which protrude from larval feeding sites. Overwintering occurs as a larva. Pupation occurs in the larval feeding tunnels close to the surface of the bark or within the frass tubes.

Larvae of *E. formosana* are a pest of fruit trees in the family Rosaceae (Table 78). Larval damage results in swellings and cankers, and branches or entire trees may be killed. Larvae have also been recorded on beech (Fagaceae).

Table 78: *Enarmonia formosana* host plants

Family	**Genus/species**	**Common name**
Fagaceae	*Fagus sylvatica* L.	European beech
Rosaceae	*Cydonia* Mill.	cydonia
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Malus sylvestris* (L.) Mill.	European crab apple
Rosaceae	*Prunus armeniaca* L.	apricot
Rosaceae	*Prunus avium* (L.) L.	sweet cherry
Rosaceae	*Prunus domestica* L.	European plum
Rosaceae	*Prunus dulcis* (Mill.) D. A. Webb	sweet almond
Rosaceae	*Prunus laurocerasus* L.	cherry laurel
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Pyrus communis* L.	common pear
Rosaceae	*Sorbus* L.	mountain ash

Distribution

Enarmonia formosana is widely distributed from Western Europe and northern Africa to Asia Minor, Russia, and Siberia (Tanigoshi and Stary 2003). The first North American records are from British Columbia in 1989; it was subsequently found in western Washington in 1991 and has spread to Oregon (Dang and Parker 1990, E. LaGasa, pers. comm.).

***Tetramoera schistaceana* (Snellen)**

Nomenclature

Tetramoera schistaceana (Snellen) (Tortricidae: Olethreutinae: Enarmoniini)

Common names: sugarcane gray borer, sugarcane shoot borer

Adult Recognition

Forewings are grayish brown with numerous longitudinal tan, gray, and brown lines. In fresh individuals the ocellus is outlined faintly in black and has numerous strong black dashes. Males lack a forewing costal fold. FWL: 4.5-7.0 mm.

Seven *Tetramoera* species are present in Asia and Australasia, all with similar wing patterns (Diakonoff 1967, Horak 2006). A genitalic dissection can be used to confirm identity.

Larval Morphology

Last instar larvae are approximately 25 mm long with a pale whitish abdomen. The head is dark brown. Distinguishing features include: pinacula small and pigmented; abdominal SD1 and SD2 setae on same pinacula; L1 seta longer than L3 on abdomen; SV pinacula trisetose on A7 and bisetose on A9; anal comb absent (Williams 1953).

Biology

Females lay eggs singly or in small batches on leaves and leaf sheaths. Larvae tunnel into stems of young plants and create irregular tunnels near the surface. Larvae may also feed on leaves. Pupation occurs in a cocoon spun in a hole in the leaf sheath. Larval damage is characterized by distorted shoots, death of the meristem, and broken stems (Diakonoff 1967, van der Geest 1991, Williams 1953).

Larvae of *T. schistaceana* feed primarily on sugarcane (Table 79) and are considered a major pest of sugarcane in Asia. This species is occasionally intercepted at U.S. ports of entry on sugarcane originating from Asia.

Table 79: *Tetramoera schistaceana* host plants

Family	Genus/species	Common name
Poaceae	*Saccharum officinarum* L.	sugarcane
Poaceae	*Saccharum spontaneum* L.	wild sugarcane
Poaceae	*Miscanthus* Andersson	silvergrass

Distribution

Tetramoera schistaceana is distributed throughout Asia (China, Indonesia, Philippines, Sri Lanka, Taiwan, Vietnam, and several smaller islands). Diakonoff (1967) lists *T. schistaceana* as present in Madagascar and Hawaii but these records could not be verified.

Tortricidae: Olethreutinae: Grapholitini

Cryptophlebia **Walsingham**

Overview

The genus *Cryptophlebia* contains approximately 45 species and is distributed in all regions except the Nearctic. Adults are moderately sized with brown forewings and variable, subdued patterns. Most species have a darker brown pretornal patch that is more pronounced in females. Males have a variety of sex scales on the legs, abdomen, and in special pockets on the hindwing. The male genitalia are characterized by swollen valvae with multiple long spines on the inner surface of the cucullus. The female genitalia are characterized by an ovate or V-shaped sterigma, an anterior widening of the ductus bursae, and two signa in the corpus bursae (Komai 1999).

Cryptophlebia larvae are borers in seeds, nuts, fruits, pods, and stems. Larvae are whitish or pinkish with dark pinacula and a brown head and prothoracic shield. An anal comb is absent in most species. The large prespiracular pinaculum on T1 extends under the spiracle and the spiracle on A8 is located on the posterodorsal margin of the segment; these two characters group *Cryptophlebia* larvae with other closely related genera such as *Gymnandrosoma* and *Thaumatotibia* (Adamski and Brown 2001, Komai 1999).

This genus contains several pest species. The three treated here, the koa seedworm (*C. illepida*), macadamia nut borer (*C. ombrodelta*), and litchi moth (*C. peltastica*), are important pests of litchi and macadamia. *Cryptophlebia* larvae are intercepted very frequently at U.S. ports of entry, although it is likely that these are not distinguished from larvae of related species, such as false codling moth (*Thaumatotibia leucotreta*), which was previously placed in *Cryptophlebia*.

Cryptophlebia illepida **(Butler)**

Nomenclature

Cryptophlebia illepida (Butler) (Tortricidae: Olethreutinae: Grapholitini)

Common names: koa seedworm

Synonyms: *fulva* (var.), *suffusa* (var.), *tetrao* (*Cryptophlebia*), *vulpes* (*Cryptophlebia*)

Adult Recognition

Adults are brown to reddish brown with a dark-brown pretornal spot that is reduced or absent in males. Males have sex scales on the hindwing, hind tibia, and abdomen. Male genitalia are characterized by swollen valvae with two large inner spines on the cucullus and several rows of smaller spines along the distal margin. Female genitalia are characterized by a wide, V-shaped sterigma and two signa in the corpus bursae. Males lack a forewing costal fold. FWL: 7.0-11.0 mm.

Adults of most *Cryptophlebia* species are superficially similar and are often mixed in museum collections (Bradley 1953). A genitalic dissection is usually necessary to confirm identity. The three species treated here, *C. illepida*, *C. ombrodelta*, and *C. peltastica*, can be separated by genitalic characters and geographic distribution, as outlined in Table 80.

Both *C. illepida* and *C. ombrodelta* occur in Hawaii; the former is assumed to be native as it is has not been found in any other locality, and the latter has been introduced. In addition to the genitalic differences listed in Table 80, adults of these two species can be separated by a character on the male hind tibia: in *C. ombrodelta* there is an ovate bare patch that is absent in *C. illepida* (Komai 1999, Zimmerman 1978).

Table 80: Comparison of *Cryptophlebia* species.

***Cryptophlebia* species**	**Male valva**	**Female sterigma**	**Distribution**
illepida	Two large spines, multiple rows of marginal spines	Wide, V-shaped	Hawaii
ombrodelta	Three large spines	Narrow, V-shaped, separate	Australia, Guam, Japan, India, Southeast Asia, Hawaii (int.)
peltastica	Three large spines, margin densely setose	Narrow, ovate, deeply inset	Africa, Seychelles, Mauritius, Guam (int.)

Larval Morphology

Late instar larvae are approximately 13-20 mm long. The abdomen is yellowish white, turning reddish in the final instar. Pinacula are large and darker than body color in most species; however, pinacula are not heavily sclerotized and may be difficult to observe in preserved individuals. The head and prothoracic shield are black or dark brown in early instars, turning pale or yellowish brown in the final instar. An anal comb is usually absent, although some individuals may have a rudimentary anal comb with 4-6 small teeth (especially common in *C. ombrodelta*). Other diagnostic features of *Cryptophlebia* larvae include: T1 prespiracular pinaculum extends below the spiracle; SV counts on A1,2,7,8,9 as 3:3:2(3):2(1):1; SV seta on A8 and A9 bisetose; spiracle on A8 near posterior margin of segment and displaced dorsally; L group on A9 usually trisetose (occasionally bisetose); D1 and SD1 setae on same pinaculum on A9; and D2 setae on shared saddle pinaculum on A9 (Komai 1999, Zimmerman 1978).

Biology

Cryptophlebia illepida completes continuous generations and adults are present year-round. The following describes the *C. illepida* life cycle on macadamia (from Namba 1957). Females lay eggs singly on the fruit (nut) of the host; as many as 15 eggs may be found on a single fruit. Larvae bore into the husk and are generally not able to penetrate the shell after hardening. Average larval development time is 16 days. Pupation occurs in a tunnel near an exit hole in the husk. Husk damage is often responsible for nut drop prior to to maturity.

This species is an important pest of macadamia, litchi, mango, and koa in Hawaii (Jones 1994, 1995, Namba 1957). Larvae are moderately polyphagous and have been recorded feeding on a variety of other plants (Table 81).

Table 81: *Cryptophlebia illepida* host plants

Family	**Genus/species**	**Common name**
Anacardiaceae	*Mangifera indica* L.	mango
Euphorbiaceae	*Nephelium lappaceum* L.	rambutan
Fabaceae	*Acacia confusa* Merr.	small Philippine acacia
Fabaceae	*Acacia farnesiana* (L.) Willd.	sweet acacia
Fabaceae	*Acacia koa* A. Gray	koa
Fabaceae	*Acacia koaia* Hillebr.	koaoha
Fabaceae	*Acacia* Mill.	acacia
Fabaceae	*Bauhinia purpurea* L.	butterfly tree
Fabaceae	*Caesalpinia kavaiense* H. Mann	
Fabaceae	*Inga edulis* Mart.	icecreambean
Fabaceae	*Mezonevron kauaiense* Hillebr.	
Fabaceae	*Parkinsonia aculeata* L.	Jerusalem thorn
Fabaceae	*Phaseolus* L.	bean
Fabaceae	*Pithecellobium dulce* (Roxb.) Benth.	monkeypod
Fabaceae	*Senna sulfurea* (DC. ex Collad.) Irwin & Barneby	smooth senna
Proteaceae	*Macadamia* F. Muell.	macadamia
Proteaceae	*Macadamia integrifolia* Maiden & Betche	macadamia nut
Rutaceae	*Aegle marmelos* (L.) Corr. Serr.	Indian bael
Sapindaceae	*Alectryon macrococcus* Radlk.	Hawaii alectryon
Sapindaceae	*Dodonaea viscosa* (L.) Jacq.	Florida hopbush
Sapindaceae	*Litchi chinensis* Sonn. [excluded]	lychee
Sapindaceae	*Sapindus oahuensis* Hillebr. ex Radlk.	lonomea

Table 81: *Cryptophlebia illepida* host plants

Family	Genus/species	Common name
Sapindaceae	*Sapindus saponaria* L.	wingleaf soapberry

Distribution

Cryptophlebia illepida has only been recorded only from Hawaii; however, Zimmerman (1978) suspects that it may be an immigrant.

Cryptophlebia ombrodelta **(Lower)**

Nomenclature

Cryptophlebia ombrodelta (Lower) (Tortricidae: Olethreutinae: Grapholitini)

Common names: litchi fruit moth, macadamia nut borer

Synonyms: *carpophaga* (*Cryptophlebia*)

Adult Recognition

Adults are brown to reddish brown with a dark-brown pretornal spot that is more pronounced in females. Males have sex scales on the hindwing, hind tibia, and abdomen. Male genitalia are characterized by swollen valvae with three large spines on the cucullus. Female genitalia are characterized by a narrow, V-shaped sterigma that is separate from the posterior margin of sternum VII and two signa in the corpus bursae. Males lack a forewing costal fold. FWL: 7.0-11.5 mm.

Adults of most *Cryptophlebia* species are superficially similar and a genitalic dissection is usually necessary to confirm identity. Differences between the species treated here are listed under the description of *C. illepida* and in Table 80.

Both *C. illepida* and *C. ombrodelta* occur in Hawaii; the former is assumed to be native as it is has not been found in any other locality, and the latter has been introduced. In addition to the genitalic differences listed in Table 80, adults of these two species can be separated by a character on the male hind tibia: in *C. ombrodelta* there is an ovate bare patch that is absent in *C. illepida* (Komai 1999, Zimmerman 1978).

Larval Morphology

Late instar larvae are approximately 13-20 mm long. The abdomen is yellowish white, turning reddish in the final instar. Pinacula are large and darker than body color in most species; however, pinacula are not heavily sclerotized and may be difficult to observe in preserved individuals. The head and prothoracic shield are black or dark brown in early instars, turning pale or yellowish brown in the final instar. An anal comb is usually absent, although some individuals may have a rudimentary anal comb with 4-6 small teeth (especially common in *C. ombrodelta*). Other diagnostic features of *Cryptophlebia* larvae include: T1 prespiracular pinaculum extends below the spiracle; SV counts on A1,2,7,8,9 as 3:3:2(3):2(1):1; SV seta on A8 and A9 bisetose; spiracle on A8 near posterior margin of segment and displaced dorsally; L group on A9 usually trisetose (occasionally bisetose); D1 and SD1 setae on same pinaculum on A9; and D2 setae on shared saddle pinaculum on A9 (Komai 1999, Zimmerman 1978).

Biology

The biology of *C. ombrodelta* is similar to that of *C. illepida*. Development is continuous and adults are present year-round. Females lay eggs on the fruits or pods of the host plant. Larvae bore into the fruit or pod and feed on the seeds. Pupation occurs in the fruit or pod or in the ground (Zimmerman 1978).

Cryptophlebia ombrodelta is an important pest of macadamia, litchi, and longan fruit in Asia, Australia, and Hawaii (Jones 1994, 1995, Zimmerman 1978). Larvae are moderately polyphagous and have been recorded feeding on plants in several families (Table 82).

Distribution

Cryptophlebia ombrodelta is recorded from Australia, Guam, India, Japan, Java, Philippines, Sri Lanka, Taiwan, and Thailand. It has been introduced into Hawaii (Bradley 1953, Horak 2006, Zimmerman 1978).

Table 82: *Cryptophlebia ombrodelta* host plants

Family	**Genus/species**	**Common name**
Arecaceae	*Cocos nucifera* L.	coconut palm
Euphorbiaceae	*Nephelium lappaceum* L.	rambutan
Fabaceae	*Acacia farnesiana* (L.) Willd.	sweet acacia
Fabaceae	*Acacia* Mill.	acacia
Fabaceae	*Acacia nilotica* (L.) Willd. ex Delile	gum arabic tree
Fabaceae	*Adenanthera pavonina* L.	red beadtree
Fabaceae	*Bauhinia hirsuta* Weinm.	orchid tree
Fabaceae	*Bauhinia* L.	bauhinia
Fabaceae	*Bauhinia malabarica* Roxb.	orchid tree
Fabaceae	*Bauhinia purpurea* L.	butterfly tree
Fabaceae	*Caesalpinia decapetala* (Roth) Alston	shoofly
Fabaceae	*Caesalpinia pulcherrima* (L.) Sw.	pride-of-Barbados
Fabaceae	*Caesalpinia sappan* L.	sappanwood
Fabaceae	*Cassia fistula* L.	golden shower
Fabaceae	*Cassia javanica* L. X *fistula* L.	rainbow shower
Fabaceae	*Cassia* L.	cassia
Fabaceae	*Delonix* Raf.	delonix
Fabaceae	*Delonix regia* (Bojer ex Hook.) Raf.	royal poinciana
Fabaceae	*Glycine max* (L.) Merr.	soybean
Fabaceae	*Indigofera suffruticosa* Mill.	anil de pasto
Fabaceae	*Parkinsonia aculeata* L.	Jerusalem thorn
Fabaceae	*Phaseolus* L.	bean
Fabaceae	*Phaseolus lunatus* L.	sieva bean
Fabaceae	*Phaseolus vulgaris* L.	kidney bean
Fabaceae	*Pithecellobium dulce* (Roxb.) Benth.	monkeypod
Fabaceae	*Poinciana* L.	
Fabaceae	*Prosopis juliflora* (Sw.) DC.	mesquite
Fabaceae	*Prosopis pallida* (Humb. & Bonpl. ex Willd.) Kunth	kiawe
Fabaceae	*Senna alata* (L.) Roxb.	emperor's candlesticks
Fabaceae	*Senna bicapsularis* (L.) Roxb.	Christmasbush
Fabaceae	*Senna* Mill.	senna
Fabaceae	*Senna occidentalis* (L.) Link	septicweed
Fabaceae	*Senna septemtrionalis* (Viv.) H. Irwin & Barneby	arsenic bush
Fabaceae	*Senna sophera* (L.) Roxb.	algarrobilla
Fabaceae	*Sesbania bispinosa* (Jaq.) W. Wight	dunchi fiber
Fabaceae	*Sesbania grandiflora* (L.) Poir.	vegetable hummingbird
Fabaceae	*Sesbania* Scop.	riverhemp
Fabaceae	*Tamarindus indica* L.	tamarind
Fabaceae	[unspecified]	
Oxalidaceae	*Averrhoa carambola* L.	carambola
Polygonaceae	*Coccoloba uvifera* (L.) L.	seagrape

Table 82: *Cryptophlebia ombrodelta* host plants

Family	**Genus/species**	**Common name**
Proteaceae	*Buckinghamia celsissima* F. Muell.	
Proteaceae	*Macadamia* F. Muell.	macadamia
Rutaceae	*Aegle marmelos* (L.) Corr. Serr.	Indian bael
Rutaceae	*Citrus* L.	citrus
Rutaceae	*Citrus* X *sinensis* (L.) Osbeck (pro sp.) [*maxima* X *reticulata*]	sweet orange
Rutaceae	*Limonia* L.	limonia
Rutaceae	*Limonium acidissima* L.	Indian woodapple
Rutaceae	[unspecified]	
Sapindaceae	*Cupaniopsis anacardioides* (A. Rich.) Radlk.	carrotwood
Sapindaceae	*Dimocarpus longan* Lour.	longan
Sapindaceae	*Filicium decipiens* Thwaites	
Sapindaceae	*Litchi chinensis* Sonn. [excluded]	lychee
Sapindaceae	[unspecified]	

Cryptophlebia peltastica **(Meyrick)**

Nomenclature

Cryptophlebia peltastica (Meyrick) (Tortricidae: Olethreutinae: Grapholitini)

Common names: litchi moth

Adult Recognition

Adults are brown to reddish brown with a dark-brown pretornal spot that is reduced in males. Males have sex scales on the hindwing, hind tibia, and abdomen. Male genitalia are characterized by swollen valvae with three large spines on the cucullus and a densely setose distal margin. Female genitalia are characterized by an ovate sterigma that is surrounded with, and fused to, the posterior margin of sternum VII and two signa in the corpus bursae. Males lack a forewing costal fold. FWL: 8.0-13.0 mm.

Adults of most *Cryptophlebia* species are superficially similar and a genitalic dissection is usually necessary to confirm identity. Differences between the species treated here are listed under the description of *C. illepida* and in Table 80.

Larval Morphology

Late instar larvae are approximately 13-20 mm long. The abdomen is yellowish white, turning reddish in the final instar. Pinacula are large and darker than body color in most species; however, pinacula are not heavily sclerotized and may be difficult to observe in preserved individuals. The head and prothoracic shield are black or dark brown in early instars, turning pale or yellowish brown in the final instar. An anal comb is usually absent, although some individuals may have a rudimentary anal comb with 4-6 small teeth (especially common in C. ombrodelta). Other diagnostic features of *Cryptophlebia* larvae include: T1 prespiracular pinaculum extends below the spiracle; SV counts on A1,2,7,8,9 as 3:3:2(3):2(1):1; SV seta on A8 and A9 bisetose; spiracle on A8 near posterior margin of segment and displaced dorsally; L group on A9 usually trisetose (occasionally bisetose); D1 and SD1 setae on same pinaculum on A9; and D2 setae on shared saddle pinaculum on A9 (Komai 1999, Zimmerman 1978).

Biology

Development of *C. peltastica* is continuous, and adults are present year-round. Females lay eggs singly on fruit. Larvae tunnel through the skin and into the fruit, and feed on the seeds. Larval development is complete in 2-4 weeks. Pupation occurs in the fruit or in the ground. Adults emerge in 1-2 weeks (Manrakhan et al. 2008).

Cryptophlebia peltastica is the primary pest of litchi in South Africa and Mauritius, with larvae damaging up to 20% of fruits in commercial orchards. This species is also a pest of macadamia in South Africa (Manrakhan et al. 2008, Timm et al. 2006). Larvae have been recorded feeding on a variety of plants (Table 83), and their polyphagous nature allows them to continue development when primary hosts are unavailable.

Distribution

Cryptophlebia peltastica is broadly distributed in Africa and has also been recorded from Seychelles, Madagascar, and Mauritius. It has been presumably introduced to Guam (Bradley 1953, Komai 1999, Timm et al. 2006).

Table 83: *Cryptophlebia peltastica* host plants

Family	**Genus/species**	**Common name**
Fabaceae	*Acacia karroo* Hayne	karroothorn
Fabaceae	*Bauhinia* L.	bauhinia
Fabaceae	*Caesalpinia pulcherrima* (L.) Sw.	pride-of-Barbados
Fabaceae	*Canavalia* Adans.	jackbean
Fabaceae	*Canavalia ensiformis* (L.) DC.	wonderbean
Fabaceae	*Ceratonia siliqua* L.	St. John's bread
Fabaceae	*Delonix regia* (Bojer ex Hook.) Raf.	royal poinciana
Fabaceae	*Gleditsia triacanthos* L.	honeylocust
Fabaceae	*Piptadenia* Benth.	
Fabaceae	*Schotia afra* Thunb.	
Fabaceae	*Tamarindus indica* L.	tamarind
Rutaceae	*Citrus* X *sinensis* (L.) Osbeck (pro sp.) [*maxima* X *reticulata*]	sweet orange
Sapindaceae	*Litchi chinensis* Sonn. [excluded]	lychee
Sapindaceae	*Litchi* Sonn.	lychee

Cydia Hübner

Overview

Cydia is a worldwide genus that contains approximately 200 described species (Baixeras et al. 2008). Adults are small to medium sized and many species are gray brown with a distinct ocellus. The genus is characterized by a thickened anal fold in the male hindwing that contains a hair pencil and modified sex scales (Horak 2006). Male genitalia are characterized by the absence of an uncus and socii and constriction of the valval neck. Females usually have two signa in the corpus bursae.

Cydia larvae are borers in the fruit, nuts, cones, and pods of mainly Fabaceae, Fagaceae, Pinaceae, and Rosaceae. Larvae are whitish to reddish and resemble those of other internal-feeding olethreutines. *Grapholita* larvae are similar in appearance but the two genera can usually be separated by the anal comb, which is present in many *Grapholita* and absent in most *Cydia*.

This genus contains many pest species, such as *C. pomonella*, which is the most widely distributed and important pest of apple, pear, and walnuts in the world. *Cydia* larvae are the most commonly intercepted tortricid larvae at U.S. ports of entry, and the species treated here cover the most commonly intercepted taxa, which arrive primarily from Asia, Europe, and South America.

***Cydia araucariae* (Pastrana)**

Nomenclature

Cydia araucariae (Pastrana) (Tortricidae: Olethreutinae: Grapholitini)

Adult Recognition

Forewings are brown to golden brown. Metallic bands, or striae, extend diagonally from the costa to the ocellus and continue vertically to the dorsum. Hindwings are brown to near black. FWL: 7.0-10.0 mm. Adults may appear similar to other species of *Cydia*. A genitalic dissection can be used to confirm identity.

Larval Morphology

Late instar larvae are approximately 20 mm in length and are pinkish with medium to large, pale-brown pinacula. The head is brown and the prothoracic shield is light brown. Diagnostic characters include: L pinaculum of T1 rather large, sometimes notched; L setae on A9 sometimes all on the same pinacula; SV-group setal counts 3:3:2(3):1(2):1; number of crochets 34-37 (Mecke 2002, Pastrana 1950).

Biology

Cydia araucariae completes several generations per year. Adults are active during the day. Females usually lay eggs on the female cones, but eggs may also be found on the needles, twigs, terminal buds, and male cones. Larvae tunnel into the seeds of Brazilian pine (Table 84), where they complete development. Larvae may also damage terminal buds. Pupation occurs in a silken cocoon (Mecke 2002, Pastrana 1950).

Table 84: *Cydia araucariae* host plants

Family	Genus/species	Common name
Araucariaceae	*Araucaria angustifolia* (Bertol.) Kuntze	Brazilian pine

Distribution

Cydia araucariae is found in association with its larval host in Brazil, Argentina, and possibly Chile (Mecke 2002, Pastrana 1950).

***Cydia caryana* (Fitch)**

Nomenclature

Cydia caryana (Fitch) (Tortricidae: Olethreutinae: Grapholitini)

Common names: hickory shuckworm

Adult Recognition

Forewings are dark brown to black with a faint ocellus. Hindwings are dark brown with a white patch on the upper basal half. Males have a row of black sex scales on the anal margin of the hindwing and lack a forewing costal fold. FWL: 5.0-7.0 mm. *Cydia caryana* can be separated from most other Nearctic and Palearctic *Cydia* by the combination of dark brown to black forewings and white patch on the hindwings (Gilligan et al. 2008).

Larval Morphology

Late instar larvae are approximately 10 mm in length with a pale abdomen. The head and prothoracic shield are yellow brown. An anal comb is absent (MacKay 1959).

Biology

Cydia caryana completes 4-5 generations in southern States. Adults are present April through September. Larvae tunnel into husks and nuts of the host plant; overwintering occurs in husks on the ground. Larvae may also inhabit galls formed by Hemiptera and other insects. Members of the Juglandaceae (pecan, hickory, and walnut) are the only recorded hosts (Table

85). Larvae are occasional pests in commercial pecan orchards (MacKay 1959, Payne and Heaton 1975).

Table 85: *Cydia caryana* host plants

Family	**Genus/species**	**Common name**
Juglandaceae	*Carya illinoinensis* (Wangenh.) K. Koch	pecan
Juglandaceae	*Carya ovata* (Mill.) K. Koch	shagbark hickory
Juglandaceae	*Carya* Nutt.	hickory
Juglandaceae	*Juglans nigra* L.	black walnut

Distribution

Cydia caryana occurs in eastern North America and has spread as far west as New Mexico (Gilligan et al. 2008).

Cydia coniferana (Saxesen)

Nomenclature

Cydia coniferana (Saxesen) (Tortricidae: Olethreutinae: Grapholitini)

Synonyms: *ericicolana* (*Coccyx*), *separatana* (*Tortrix*)

Adult Recognition

Adults are small and brown. Many individuals have a pair of white strigulae that run continuous from costa to dorsum; expression of these markings may vary considerably. FWL: 4.5-6.5 mm. Male genitalia are characterized by a toothlike projection of the ventral margin of the valva and numerous small cornuti in the aedeagus. Female genitalia are characterized by a short ductus bursae and a pair of small signa in the corpus bursae. Adults of *C. coniferana* are similar to other *Cydia* and *Grapholita*. A genitalic dissection may be necessary to confirm identity.

Larval Morphology

Late instar larvae are white or yellowish with a light-brown head that may have darker posterolateral shading. The prothoracic shield is yellowish brown and lightly sclerotized and the anal shield is marked with dark-brown spots (Bradley et al. 1979, LaGasa and Passoa 2007).

Biology

Cydia coniferana completes 1-2 generations per year in Europe. Adults are present from mid-May to August. Larvae create a silk-lined tunnel under the bark of the host tree and feed in the cambium. Larval damage is characterized by loose bark and exuded frass mixed with resin. Larvae often infest trees that are infected with fungus or those that are injured. Larvae have been reported feeding on various species of fir, spruce, and pine (Table 86). This species is generally characterized as an occasional pest of young pines (Bradley et al. 1979, LaGasa and Passoa 2007, Razowski 2003).

Table 86: *Cydia coniferana* host plants

Family	**Genus/species**	**Common name**
Pinaceae	*Abies* Mill.	fir
Pinaceae	*Picea* A. Dietr.	spruce
Pinaceae	*Picea abies* (L.) Karst.	Norway spruce
Pinaceae	*Picea alba* Link	
Pinaceae	*Picea sitchensis* (Bong.) Carriere	Sitka spruce
Pinaceae	*Pinus nigra* Arnold	Austrian pine
Pinaceae	*Pinus resinosa* Aiton	red pine
Pinaceae	*Pinus sylvestris* L.	Scots pine

Distribution

Cydia coniferana is distributed from Europe east to China and Mongolia (Razowski 2003). The first North American record is from New York in the late 1950s but the population never established. It was not detected again in North America until an adult was collected in Washington in 2000 (LaGasa and Passoa 2007). Additional surveys have shown that it is established and widespread in Washington and Oregon, although unpublished molecular data suggests that this recent introduction may be a different species (E. LaGasa, pers. comm.).

***Cydia deshaisiana* (Lucas)**

Nomenclature

Cydia deshaisiana (Lucas) (Tortricidae: Olethreutinae: Grapholitini)

Common names: Mexican jumping bean moth

Synonyms: *saltatoria* (*Carpocapsa*), *saltitans* (*Carpocapsa*), *sebastianiae* (*Grapholitha*)

Adult Recognition

Forewings are gray to light gray with dark-brown to black markings. Many individuals have a row of black dots or dashes along the termen and a white dot below two dark markings on the apex. Hindwings are brown. FWL: 8.0-11.0 mm. Adults may appear similar to many other species of *Cydia*. A genitalic dissection can be used to confirm identity.

Larval Morphology

Larvae are whitish and unmarked with a brown head. Although similar to those of many other *Cydia*, larvae are unlikely to be encountered outside of a Euphorbiaceae seed.

Biology

Larvae tunnel inside the seeds of various species of Euphorbiaceae (Table 87) and feed inside. The seeds eventually drop from the host plant during the summer rainy season. Larvae are able to move or “jump” the seeds, or “beans,” by rapid twitching; this behavior is triggered by temperature and may be a mechanism to move the seed to a more favorable location (such as out of direct sunlight). Larvae overwinter within the seed and pupation occurs the following spring. Adults emerge from the seed through a circular “door” that is cut by the larva prior to pupation (Cranshaw 2010, Wagner 2005).

Table 87: *Cydia deshaisiana* host plants

Family	**Genus/species**	**Common name**
Euphorbiaceae	*Croton* L.	croton
Euphorbiaceae	*Sapium* Jacq.	milktree
Euphorbiaceae	*Sebastiania bilocularis* S. Watson	arrow poision plant
Euphorbiaceae	*Sebastiania pavoniana* (Mull. Arg.) Mull. Arg.	Mexican jumping bean
Euphorbiaceae	*Sebastiania* Spreng.	Sebastian-bush

Distribution

Cydia deshaisiana is a native of Mexico and is found in the northern states of Sinoloa and Sonora (Wagner 2005). The "Mexican jumping bean capitol of the world" is supposedly near the town of Alamos, Sonora.

"Mexican jumping beans" are frequently imported into the U.S., occasionally in large numbers. Several websites are available that supply the "beans," complete with live larvae, on a commercial scale. Such sites include: http://www.jbean.com/ and http://www.jumpingbeansrus.com/.

***Cydia fabivora* (Meyrick)**

Nomenclature

Cydia fabivora (Meyrick) (Tortricidae: Olethreutinae: Grapholitini)

Synonyms: *leguminis* (*Laspeyresia*), *prosecta* (*Eulia*)

Adult Recognition

Adults are grayish brown with indistinct black, brown, and gray markings. Many individuals have a black patch near the apex of the forewing that may extend to the termen. FWL: 7.0-11.0 mm. Male genitalia are characterized by a triangular cucullus with a row of dense spines. Female genitalia are characterized by a ductus bursae that is partially sclerotized and two thornlike signa. Adults appear similar to other Grapholitini, including *C. torostoma*, which feeds on beans in Central America (Clarke 1972). A genitalic dissection may be necessary to confirm identity.

Larval Morphology

Late instar larvae are approximately 18 mm in length with a whitish abdomen. The head, prothoracic shield, and thoracic legs are brown. Abdominal pinacula are large, brown, and conspicuous. An anal comb is absent (MacKay 1959).

Larvae are similar to many species in the *Cryptophlebia-Ecdytolopha* group, with an enlarged L-pinaculum on the prothorax that extends beneath (and usually beyond) the spiracle. Adamski and Brown (2001) hypothesize that *C. fabivora* is placed in the wrong genus based on this and other characters.

Larval damage to beans is similar to that caused by *Crocidosema aporema*; however, larvae of *C. aporema* feed primarily on young leaflets while those of *C. fabivora* feed in the stems and pods. An anal comb is present in *C. aporema* but is absent in *C. fabivora*. Other bean-feeding tortricids in South and Central America include *Amorbia* spp., *C. torostoma*, *Lusterala phaseolana*, and *Platynota* spp.

Biology

The following life history information is summarized from Clarke (1972), Heinrich (1943), and Stansly and Sanchez (1990). *Cydia fabivora* completes several generations per year. In areas where host plants are available year-round, adults may be present at any time.

Females lay eggs on the leaves, petioles, stems, flowers, and pods, of soybean; oviposition behavior varies with plant age. Larvae tunnel into the petioles, stems, or pods, sometimes causing considerable damage. Larvae may also feed on terminals and flowers. Pupation occurs in cocoons at the larval feeding site.

Cydia fabivora is a pest of beans (lima beans, string beans, soybeans – Table 88) in Central and South America. Larval damage is characterized by stunted plants, pod loss, or complete plant death in the case of tunnelling in the main stem.

Table 88: *Cydia fabivora* host plants

Family	Genus/species	Common name
Fabaceae	*Glycine max* (L.) Merr.	soybean
Fabaceae	*Phaseolus lunatus* L.	sieva (lima) bean
Fabaceae	*Phaseolus vulgaris* L.	kidney bean
Fabaceae	*Phaseolus* L.	bean

Distribution

Cydia fabivora is widely distributed in Central and South America (Clarke 1972, Stansly and Sanchez 1990).

***Cydia fagiglandana* (Zeller)**

Nomenclature

Cydia fagiglandana (Zeller) (Tortricidae: Olethreutinae: Grapholitini)

Synonyms: *grossana* (*Tortrix*)

Subspecies: *corsica* (Corsica, France)

Adult Recognition

Adults are brownish gray with numerous white markings. A subtriangular dark patch is present proximal to the poorly defined ocellus. Males have a whitish-gray patch of sex scales on the dark brown hindwings. FWL: 6.0-9.5 mm.

Similar species include *C. splendana* in Europe and *C. kurokoi* in Asia, although both are generally lighter gray in color and have a large subtriangular patch surrounding the ocellus on the distal one-third of the forewing.

Larval Morphology

Late instar larvae are approximately 14 mm in length. The abdomen is whitish with orange or red suffused longitudinal stripes. Abdominal pinacula are orange to red. The head is light brown and the prothoracic shield is reddish yellow. An anal comb is absent (Brown and Komai 2008).

Other tortricid *Castanea* pests include *C. glandicolana*, *C. kurokoi*, *C. splendana*, *Fibuloides aestuosa*, and *Pammene fasciana*. Brown and Komai (2008) provide a description and key to larvae of these species. *Cydia fagiglandana* larvae can be distinguished by the following characters: abdomen reddish white with orange to red pinacula; distance between V setae on A9 usually the same as on A8; more than 18 crochets on abdominal prolegs; anal comb absent.

Biology

Cydia fagiglandana completes a single generation per year. Adults are present from May to July. On chestnut, females deposit eggs near the fruits. Larvae bore into the fruit and consume the seeds. Larvae may complete development within a single fruit or move to other fruits in host species with small seeds. Overwintering occurs as a late instar larva. Pupation occurs in the spring. Larvae are a common pest of beech, chestnut, and oak (Table 89) (Bogenschutz 1991, Bradley et al. 1979, Brown and Komai 2008).

Table 89: *Cydia fagiglandana* host plants

Family	**Genus/species**	**Common name**
Betulaceae	*Corylus avellana* L.	common filbert
Fagaceae	*Castanea* Mill.	chestnut
Fagaceae	*Castanea sativa* Mill.	European chestnut
Fagaceae	*Fagus sylvatica* L.	European beech
Fagaceae	*Quercus ilex* L.	holly oak
Fagaceae	*Quercus ilex* subsp. *rotundifolia* (Lam.) Tab. Morais	
Fagaceae	*Quercus* L.	oak
Fagaceae	*Quercus suber* L.	cork oak

Distribution

Cydia fagiglandana occurs from Europe east to northern Iran and central Asia (Razowski 2003).

Cydia garacana (Kearfott)

Nomenclature

Cydia garacana (Kearfott) (Tortricidae: Olethreutinae: Grapholitini)

Synonyms: *septicola* (*Enarmonia*)

Adult Recognition

Adults are gray with two pairs of grayish-white dorsal strigulae. Males lack a forewing costal fold. FWL: 5.5-6.5 mm.

Cydia garacana is similar to other Nearctic *Cydia*, such as *C. populana* and *C. flexiloqua*. A genitalic dissection is recommended to confirm identity; consult Heinrich (1926) for genitalic differences between species listed here.

Larval Morphology

The larva of *C. garacana* is undescribed. It is assumed to be similar to that of other *Cydia* species, including *C. pomonella*.

Biology

Life history information is not recorded for this species. Larvae feed on the leaves of *Populus* (cottonwood) (Table 90) (Gilligan et al. 2008).

Table 90: *Cydia garacana* host plants

Family	Genus/species	Common name
Salicaceae	*Populus* L.	cottonwood

Distribution

Cydia garacana is distributed from Ontario to Minnesota, south to Texas (Gilligan et al. 2008).

Cydia glandicolana **(Danilevsky)**

Nomenclature

Cydia glandicolana (Danilevsky) (Tortricidae: Olethreutinae: Grapholitini)

Adult Recognition

Forewings are gray, darker basally, with a subtriangular dorsal patch proximal to the ocellus. Hindwings are grayish brown. FWL: 6.5-10.0 mm.

Other similar species include *C. splendana* and *C. kurokoi*. A dark patch surrounds the ocellus on the distal one-third of the forewing in both of these species. A genitalic dissection can be used to confirm identity.

Larval Morphology

Late instar larvae are approximately 17 mm in length. The abdomen is yellowish red with concolorous inconspicuous pinacula. The head is yellowish brown and the prothoracic shield is pale yellow brown with mottling along the posterior margin. An anal comb is absent (Brown and Komai 2008).

Other tortricid *Castanea* pests include *C. fagiglandana*, *C. kurokoi*, *C. splendana*, *Fibuloides aestuosa*, and *Pammene fasciana*. Brown and Komai (2008) provide a description and key to larvae of these species. *Cydia glandicolana* larvae can be distinguished by the following characters: reddish abdomen with inconspicuous pinacula; D2 pinacula fused to each other on A9; distance between V setae on A9 greater than on A8; 19-26 crochets on abdominal prolegs; anal comb absent.

Biology

Cydia glandicolana completes one generation per year. Adults are present July through September. Larvae feed within chestnuts and acorns. Overwintering occurs in leaf litter as a late instar. This species is a pest of chestnut in China. In Japan it has only been recorded feeding on oak (Komai and Ishikawa 1987). A list of documented host plants is provided in Table 91.

Table 91: *Cydia glandicolana* host plants

Family	**Genus/species**	**Common name**
Fagaceae	*Castanea mollissima* Blume	Chinese chestnut
Fagaceae	*Castanea* Mill.	chestnut
Fagaceae	*Quercus dentata* Thunb.	Daimyo oak
Fagaceae	*Quercus mongolica* Fisch. ex Ledeb.	mongolian oak
Fagaceae	*Quercus serrata* Thunb.	bao li
Fagaceae	*Quercus* sp.	oak

Distribution

Cydia glandicolana is distributed throughout China, Korea, and Japan. There are also records from southeastern Russia (Razowski 2003).

Cydia kurokoi **(Amsel)**

Nomenclature

Cydia kurokoi (Amsel) (Tortricidae: Olethreutinae: Grapholitini)

Adult Recognition

Forewings are light gray and brown. The distal one-third of the wing is marked with a dark-brown to black subtriangular patch that surrounds the ocellus. Hindwings are grayish brown. FWL: approx. 10 mm.

Adults may appear similar to other species of *Cydia*, including the European *C. splendana*. A dissection can be used to confirm identity.

Larval Morphology

Late instar larvae are whitish with a yellow-brown head and prothoracic shield. Abdominal pinacula are dark and conspicuous (Brown and Komai 2008).

Other tortricid *Castanea* pests include *C. fagiglandana*, *C. glandicolana*, *C. splendana*, *Fibuloides aestuosa*, and *Pammene fasciana*. Brown and Komai (2008) provide a description and key to larvae of these species. *Cydia kurokoi* larvae can be distinguished by the following characters: whitish abdomen with darker pinacula; D2 pinacula fused to each other on A9; all three L setae on same pinaculum on A9; distance between V setae on A9 greater than on A8; 25-35 crochets on abdominal prolegs; anal comb absent.

Biology

Cydia kurokoi completes one generation per year. Adults are present in August and September. Larvae tunnel into and feed inside chestnuts. Overwintering occurs in leaf litter as a late instar. This species is a pest of chestnuts in Japan. Larvae have also been recorded on *Quercus* (Table 92) (Brown and Komai 2008, Komai and Ishikawa 1987).

Table 92: *Cydia kurokoi* host plants

Family	**Genus/species**	**Common name**
Fagaceae	*Castanea crenata* Siebold & Zucc.	Japanese chestnut
Fagaceae	*Castanea mollissima* Blume	Chinese chestnut
Fagaceae	*Castanea seguinii* Dode	Seguin's chestnut
Fagaceae	*Castanea* Mill.	chestnut
Fagaceae	*Quercus acutissima* Carruth.	sawtooth oak
Fagaceae	*Quercus* L.	oak

Distribution

Cydia kurokoi occurs in China, Korea, and Japan (Brown and Komai 2008).

***Cydia latiferreana* (Walsingham)**

Nomenclature

Cydia latiferreana (Walsingham) (Tortricidae: Olethreutinae: Grapholitini)

Common names: filbertworm

Synonyms: *aurichalceana* (*Melissopus*), *inquilina* (*Cydia*)

Adult Recognition

Adult forewing color is extremely variable and ranges from pale tan to orange red to dark brown. Most individuals are marked with two metallic transverse bands that run from costa to dorsum. FWL: 6.0-9.5 mm.

Male genitalia are equally variable. Heinrich (1926) designated seven different forms (type A-G) based on variation in the dorsally projecting extensions of the tegumen and lateral projection off the aedeagus. The different forms are somewhat geographically isolated, and are assumed to constitute a species complex (Brown 1983), although there have been no conclusive studies that demonstrate this to be the case.

Larval Morphology

Last instar larvae are approximately 12-15 mm in length with a whitish-gray abdomen. The head is yellowish brown. The prothoracic shield is pale brown with faint dark mottling. An anal comb is absent (MacKay 1959, Passoa 2008).

Biology

Cydia latiferreana completes multiple generations over most of its range. Adults may be present March to November in southern locations with reduced flight periods in the North. Larvae feed within the acorns, nuts, and burrs of *Quercus* (oak), *Fagus* (beech), *Corylus* (hazelnut and filbert), and *Castanea* (chestnut) (Table 93). Overwintering occurs in the soil or leaf litter, and pupation occurs the following spring. Larvae have also been reported to infest oak galls produced by cynipid wasps (Gilligan et al. 2008, MacKay 1959).

Table 93: *Cydia latiferreana* host plants

Family	**Genus/species**	**Common name**
Betulaceae	*Corylus avellana* L.	common filbert
Betulaceae	*Corylus* L.	hazelnut
Fagaceae	*Castanea* Mill.	chestnut
Fagaceae	*Fagus* L.	beech
Fagaceae	*Quercus agrifolia* Nee	California live oak
Fagaceae	*Quercus alba* L.	white oak
Fagaceae	*Quercus chrysolepis* Liebm.	canyon live oak
Fagaceae	*Quercus douglasii* Hook. & Arn.	blue oak
Fagaceae	*Quercus falcata* Michx.	southern red oak
Fagaceae	*Quercus kelloggii* Newb.	California black oak
Fagaceae	*Quercus* L.	oak
Fagaceae	*Quercus lobata* Nee	valley oak
Fagaceae	*Quercus macrocarpa* Michx.	bur oak
Fagaceae	*Quercus nigra* L.	water oak
Fagaceae	*Quercus rubra* L.	red oak
Fagaceae	*Quercus velutina* Lam.	black oak
Fagaceae	*Quercus wislizeni* A. DC.	interior live oak
Juglandaceae	*Juglans regia* L.	English walnut
Proteaceae	*Macadamia* F. Muell.	macadamia
Punicaceae	*Punica granatum* L.	pomegranate
Rosaceae	*Prunus dulcis* (Mill.) D. A. Webb	sweet almond
Rosaceae	*Prunus ilicifolia* (Nutt. ex Hook. & Arn.) D. Dietr. ssp. *lyonii* (Eastw.) P. H. Raven	hollyleaf cherry
Rosaceae	*Prunus* L.	

Distribution

Cydia latiferreana is widely distributed across North America and northern Mexico (Gilligan et al. 2008, Powell and Opler 2009).

Cydia pomonella **(Linnaeus)**

Nomenclature

Cydia pomonella (Linnaeus) (Tortricidae: Olethreutinae: Grapholitini)

Common names: codling moth

Synonyms: *aeneana* (*Tortrix*), *glaphyrana* (*Carpocapsa splendana* ab.), *pomana* (*Pyralis*), *pomonana* (*Tortrix*)

Subspecies: *simpsonii* (Idaho)

Cydia pomonella is often referred to as *Carpocapsa pomonella* and *Laspeyresia pomonella* in older literature. The correct generic name is *Cydia*; refer to Brown (1979) and Brown (2006) for more information.

Adult Recognition

Forewings are gray with silvery striations. The ocellus is dark purplish brown and is edged with metallic gold or copper scales. FWL: 6.5-11.0 mm. Hindwings are grayish brown and males have a fold along the base of the cubital vein that contains a hair pencil with long black sex scales. Male genitalia are characterized by a ventrally projecting spur at the base of the cucullus. Female genitalia are characterized by a short sclerotized ductus bursae.

Adults may appear similar to dark individuals of *C. splendana*. *Cydia pomonella* can be separated from *C. splendana* by the metallic scales surrounding the ocellus and the hair pencil on the male hindwing. A genitalic dissection can be used to confirm identity.

Larval Morphology

Late instar larvae are approximately 15-19 mm in length with a whitish or pale-yellow abdomen and moderately large pinacula. The head is yellowish brown and may be mottled or unmarked. The prothoracic and anal shields are yellowish brown with distinct mottling. An anal comb is absent (MacKay 1959).

Larvae may appear similar to those of other Grapholitini, especially other *Cydia* and *Grapholita*. *Cydia pomonella* can be separated from most *Grapholita* by the absence of an anal comb. Larvae of *C. splendana* are similar but lack the mottling on the prothoracic and anal shields. Molecular diagnoses may be necessary to positively identify some individuals or early instars.

Biology

The following life history information is compiled from Barnes (1991), Falcon and Huber (1991), Gilligan et al. (2008), and Wearing et al. 2001. *Cydia pomonella* completes 2-4 generations per year. In North America, adults are present in many locations from April through September.

Females lay eggs singly on fruits, stems, or leaves of the host. Larvae tunnel into fruit to feed on the seeds. Larval damage to fruit is characterized by entry and exit holes, rot that surrounds larval feeding areas, and frass accumulation. Mature larvae exit the fruit and create a cocoon under tree bark or in leaf litter; overwintering occurs as a prepupa.

Cydia pomonella is the most widely distributed and important pest of apple, pear, and walnuts in the world. Other larval hosts include almond, apricot, fig, macadamia, nectarine, peach, plum, prune, and quince (Table 94). Wearing et al. 2001 provide evidence that sweet cherry (*Prunus avium*) is not a larval host. Larvae were intercepted by the USDA on Citrus in 2009 originating from Australia, confirming Rutaceae as a larval host.

Table 94: *Cydia pomonella* host plants

Family	**Genus/species**	**Common name**
Fagaceae	*Castanea sativa* Mill.	European chestnut
Juglandaceae	*Juglans* L.	walnut
Juglandaceae	*Juglans regia* L.	English walnut
Moraceae	*Ficus carica* L.	edible fig
Proteaceae	*Macadamia* F. Muell.	macadamia
Rosaceae	[unspecified]	
Rosaceae	*Cydonia oblonga* Mill.	quince
Rosaceae	*Malus domestica* Borkh.	apple
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Malus sylvestris* (L.) Mill.	European crab apple
Rosaceae	*Prunus* L.	
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Pyrus communis* L.	common pear
Rosaceae	*Pyrus* L.	pear

Table 94: *Cydia pomonella* host plants

Family	Genus/species	Common name
Rosaceae	*Sorbus aria* (L.) Crantz	chess-apple
Rutaceae	*Citrus* L.	citrus

Distribution

Cydia pomonella is found in nearly all temperate pome fruit-growing regions of the world. It is notably absent from Japan and Korea (Barnes 1991).

Cydia splendana **(Hübner)**

Nomenclature

Cydia splendana (Hübner) (Tortricidae: Olethreutinae: Grapholitini)

Common names: chestnut fruit tortrix, acorn moth

Synonyms: *bicolorana* (*Tinea*), *bicolorata* (*Tinea*), *glandella* (*Tortrix*), *pencleriana* (*Tortrix*), *penkleriana* (*Tortrix*), *reaumurana* (*Carpocapsa*)

Adult Recognition

Forewings are light gray to gray. The distal one-third of the wing is marked with a dark-brown to black subtriangular patch that surrounds the purplish ocellus. Hindwings are dark grayish brown. FWL: 6.5-10.5 mm.

Adults may appear similar to those of other *Cydia* species. *Cydia kurokoi* is very similar but only occurs in Asia. *Cydia fagiglandana* is darker overall and lacks a well-defined purplish ocellus. *Cydia pomonella* is a more uniform gray and the ocellus is edged with gold or copper scales. A genitalic dissection can be used to confirm identity of the species listed here.

Larval Morphology

Late instar larvae are approximately 13-15 mm in length with a whitish gray-green to pale-yellow abdomen. The head is yellowish brown and the prothoracic shield is paler than the head and weakly sclerotized. According to Brown and Komai (2008), an anal comb is absent; Meijerman and Ulenberg (2000) incorrectly list the anal comb as present.

Other tortricid *Castanea* pests include *C. fagiglandana*, *C. glandicolana*, *C. kurokoi*, *Fibuloides aestuosa*, and *Pammene fasciana*. Brown and Komai (2008) provide a description and key to larvae of these species. *Cydia splendana* larvae can be distinguished by the following characters: whitish abdomen with concolorous pinacula; distance between V setae on A9 slightly to conspicuously greater than those on A8; number of crochets on prolegs less than 18; anal comb absent.

Biology

Cydia splendana completes a single annual generation. Adults are present in June to July in Central and Northern Europe and August to September in Southern Europe. Females lay eggs singly on young fruit or on leaves near fruit. Early instars tunnel into fruit and feed inside and a single fruit generally supports one larva. Larval-infested fruit drop to the ground early. Overwintering occurs as a late instar lava under bark or in the soil. Pupation occurs the following spring (Bogenschutz 1991, Bradley et al. 1979, Brown and Komai 2008).

Larvae of *Cydia splendana* are important pests of chestnut (*Castanea sativa*). Other preferred hosts include *Quercus*, *Fagus*, and *Juglans* (Table 95).

Table 95: *Cydia splendana* host plants

Family	**Genus/species**	**Common name**
Fagaceae	*Castanea* Mill.	chestnut
Fagaceae	*Castanea sativa* Mill.	European chestnut
Fagaceae	*Fagus sylvatica* L.	European beech
Fagaceae	*Quercus* L.	oak
Fagaceae	*Quercus petraea* (Mattuschka) Liebl.	durmast oak
Fagaceae	*Quercus robur* L.	English oak
Fagaceae	*Quercus rubra* L.	red oak
Juglandaceae	*Juglans* L.	walnut

Distribution

Cydia splendana is widely distributed thoughout Europe (Razowski 2003). Records from Asia are likely of a different species (see *Cydia kurokoi*).

Cydia strobilella **(Linnaeus)**

Nomenclature

Cydia strobilella (Linnaeus) (Tortricidae: Olethreutinae: Grapholitini)

Synonyms: *kollarana* (*Tortrix*), *strobilana* (*Tortrix*), *youngana* (*Enarmonia*)

Adult Recognition

Adults are dark brown with lighter, metallic, transverse markings. Hindwings are brown. Males lack a forewing costal fold. FWL: 4.5-7.0 mm. *Cydia strobilella* is similar to other Palearctic pine-feeding *Cydia*, including *C. conicolana*, *C. cosmophorana*, and *C. pactolana*. A genitalic dissection can be used to confirm identity.

Larval Morphology

Larvae are pale yellow to white with a light or dark-brown head. The prothoracic shield is light brown with darker posterolateral shading (Bradley et al. 1979). In North America, larvae of *C. strobilella* are intercepted on spruce cones imported from Europe.

Biology

Cydia strobilella completes 1-2 annual generations. In Europe, a single generation is most common and adults are present April to May in the South and May to June in the North. In North America, two generations are common and adults are present in April and May and again in August and September. Adults are active during the afternoon. Females lay eggs under the scales of young cones. Early instar larvae feed on the scales, later moving to the seeds. Mid- to late instars tunnel inside the cone and form a double, branched mine. Pupation occurs in the cone. Each cone may support several larvae (Bradley et al. 1979, Brockerhoff and Kenis 1996). A list of documented host plants is provided in Table 96.

Distribution

Cydia strobilella occurs from Europe to China and Japan. It is also present in the northern United States and Canada (Razowski 2003).

Table 96: *Cydia strobilella* host plants

Family	**Genus/species**	**Common name**
Pinaceae	*Abies alba* Mill.	silver fir
Pinaceae	*Abies* Mill.	fir
Pinaceae	*Picea* A. Dietr.	spruce
Pinaceae	*Picea abies* (L.) Karst.	Norway spruce
Pinaceae	*Picea engelmannii* Parry ex Engelm.	Engelmann spruce
Pinaceae	*Picea glauca* (Moench) Voss	white spruce
Pinaceae	*Picea jezoensis* (Siebold & Zucc.) Carriere	
Pinaceae	*Picea koyamae* Shiras.	
Pinaceae	*Picea mariana* (Mill.) Britton, Sterns & Poggenb.	black spruce
Pinaceae	*Picea obovata* Ledeb.	
Pinaceae	*Picea omorika* (Pancic) Purk.	
Pinaceae	*Picea pungens* Engelm.	blue spruce
Pinaceae	*Picea rubens* Sarg.	red spruce
Pinaceae	*Picea sitchensis* (Bong.) Carriere	Sitka spruce
Pinaceae	*Pinus pungens* Lamb.	Table Mountain pine
Pinaceae	*Pinus sylvestris* L.	Scots pine

Cydia toreuta **(Grote) [complex]**

Nomenclature

[Species complex] (Tortricidae: Olethreutinae: Grapholitini)

Cydia toreuta (Grote)

Cydia piperana Kearfott

Common names: eastern pine seedworm, ponderosa pine seed moth

Adult Recognition

Forewings are dark grayish brown with three transverse metallic bars running from costa to dorsum. Males lack a forewing costal fold. FWL: 5.0-7.5 mm (*C. toreuta*); 8.0-10.0 mm (*C. piperana*).

Cydia toreuta belongs to a complex of similar species that includes names such as *C. ingens*, *C. injectiva*, *C. miscitata*, and *C. piperana*. Placement to complex is straightforward based on the distinctive forewing pattern. It is unknown how many species are in this complex, and it most likely includes several undescribed species. The common eastern species is *C. toreuta* and the common western species is *C. piperana* (Abrahamson and Kraft 1965, Gilligan et al. 2008, Heinrich 1926).

Larval Morphology

Late instar larvae are approximately 12 mm in length with a pale abdomen and yellowish-brown head and prothoracic shield. An anal comb is absent (MacKay 1959).

Biology

Members of this complex generally complete one generation per year. Adults are present in the East from late May to mid-August and on the West Coast from Februrary to June. Females deposit eggs on green cones. Larvae bore between the cone scales and consume the seeds. Overwintering occurs in a tunnel at the center of the cone (Abrahamson and Kraft 1965, Powell and Opler 2009). A list of documented host plants is provided in Table 97.

Table 97: *Cydia toreuta* host plants

Family	**Genus/species**	**Common name**
Pinaceae	*Pinus banksiana* Lamb.	jack pine
Pinaceae	*Pinus contorta* Douglas ex Louden	lodgepole pine
Pinaceae	*Pinus echinata* Mill.	shortleaf pine
Pinaceae	*Pinus resinosa* Aiton	red pine
Pinaceae	*Pinus taeda* L.	loblolly pine
Pinaceae	*Pinus virginiana* Mill.	Virginia pine

Distribution

Cydia toreuta is distributed from Pennsylvania to Minnesota, south to Texas. *Cydia piperana* is widely distributed in the West. *Cydia ingens* is the name applied to the species found in Flordia. Other undescribed species range into Mexico (Gilligan et al. 2008, Heinrich 1926, Powell and Opler 2009).

***Ecdytolopha* Zeller**

Overview

The genus *Ecdytolopha* consists of 10 species and ranges from Peru, north through Central America and the U.S. to southern Canada (Adamski and Brown 2001). Forewings are dark brown basally and light gray or white apically with a dark spot near the tornus in many species. Male genitalia are distinguished by the following characters: tegumen rounded; uncus and socii absent; valvae elongate, cucullus densely setose; and vesica with numerous deciduous cornuti. Female genitalia are distinguished by two signa in the corpus bursae, and the location of the ostium, which is usually located in a deep invagination of sternum VII. Adamski and Brown (2001) provide keys to males and females.

Larvae are similar to other species in the *Cryptophlebia-Ecdytolopha* group, with an enlarged L-pinaculum on the prothorax that extends beneath (and usually beyond) the spiracle. Larvae of *Gymnandrosoma* can be separated from those of *Ecdytolopha* by the distance between the V setae on A9: approximately the same as the distance between Vs on A8 in *Ecdytolopha* and 1.2-2.0 times the distance between Vs on A8 in *Gymnandrosoma* (Adamski and Brown 2001).

The single species treated here, *E. insiticiana*, is the locust twig borer from economic literature. Larvae feed within new growth on locust and can cause tree disfigurement (Solomon 1995). There are many records of *Ecdytolopha* larval interceptions at U.S. ports of entry; however, most of these probably refer to *Gymnandrosoma*, as species such as *G. aurantianum* were placed in *Ecdytolopha* prior to 2001.

Ecdytolopha insiticiana **Zeller**

Nomenclature

Ecdytolopha insiticiana Zeller (Tortricidae: Olethreutinae: Grapholitini)

Common names: locust twig borer

Adult Recognition

Forewings are dark brown basally and light gray or white apically with a dark-brown mark on the dorsum near the tornus. FWL: 9.0-13.5 mm. Male genitalia are characterized by a rounded tegumen, long valvae, and deciduous cornuti in the vesica. Female genitalia are characterized by a deep invagination of sternum VII where the ostium is located, two signa in the corpus bursae, and a sclerotized patch near the junction of the corpus bursae and ductus bursae.

Ecdytolopha insiticiana appears similar to other members of the genus. Males can be separated from other *Ecdytolopha* species by a hair pencil in a narrow concavity on the anal margin of the hindwing. Females can be separated by genitalic characters; see Adamski and Brown (2001) for keys to females.

Larval Morphology

Late instar larvae are approximately 15-18 mm long with a pinkish-red abdomen and large, conspicuous pinacula. The head and prothoracic shield is yellowish or reddish brown and may have faint mottling. An anal comb is absent. Other laval characters include: prothoracic L-pinaculum subrectangular; D2s on A8 on same pinaculum; and SV setal counts on A1,2,7,8,9 as 3:3:3:2:2 (Adamski and Brown 2001, Solomon 1995).

Larvae are similar to other species in the *Cryptophlebia-Ecdytolopha* group, with an enlarged L-pinaculum on the prothorax that extends beneath (and usually beyond) the spiracle. Larvae of *Gymnandrosoma* can be separated from those of *Ecdytolopha* by the distance between the V setae on A9: approximately the same as the distance between Vs on A8 in

Ecdytolopha and 1.2-2.0 times the distance between Vs on A8 in *Gymnandrosoma* (Adamski and Brown 2001).

Biology

Ecdytolopha insiticiana completes a single annual generation in the North and two annual generations in the South. Adults are present May-June, and again in July-September for the second generation. Females lay eggs on news shoots of the host. Larvae bore into new growth and induce the formation of elongate galls. Larvae complete seven instars. Those of the last generation leave the gall and overwinter in flattened cocoons constructed in leaf litter. Pupation occurs in the spring. The primary larval host is black locust (*Robinia pseudoacacia*) (Table 98). This species can be a pest in nurseries and plantations, causing disfiguration of trees (Harman and Berisford 1979, Solomon 1995).

Table 98: *Ecdytolopha insiticiana* host plants

Family	**Genus/species**	**Common name**
Fabaceae	*Robinia* L.	locust
Fabaceae	*Robinia pseudoacacia* L.	black locust
Fabaceae	*Wisteria* Nutt.	wisteria

Distribution

Ecdytolopha insiticiana is widely distributed in the continental U.S. and southern Canada, although it is more common in the East (Gilligan et al. 2008).

Grapholita Treitschke

Overview

The worldwide genus *Grapholita* contains approximately 125 described species. Its members are divided into two subgenera, *Grapholita* and *Aspila*, and are defined by a pair of coremata and short lateral projections on the eighth sternite in the male (Komai 1999). Adults are small (FWL 3.5-8.0 mm) and brown, usually with white markings and a well-defined ocellus.

Many species have bright white costal strigulae and one or more pairs of dorsal strigulae. Male genitalia are characterized by the absence of an uncus and socii, and a well-defined, densely setose cucullus. Female genitalia are characterized by a short ductus bursae and two thornlike signa in the corpus bursae in many species.

This genus contains many well known tortricid pests, such as the oriental fruit moth (*G. molesta*), cherry fruit worm (*G. packardi*), lesser apple worm (*G. prunivora*), and plum fruit moth (*G. funebrana*). Larvae are internal feeders in fruit and can cause significant economic damage to stone-fruits (almonds, apricots, cherries, nectarines, peaches, pears, plums, etc.). The white to reddish larvae resemble those of other internal-feeding olethreutines, such as *Cydia*, but larvae of the two genera can usually be separated by the anal fork, which is present in many *Grapholita* and absent in most *Cydia*.

With the exception of *G. funebrana*, all of the species treated here are present in the U.S. and are listed as non-targets. *Grapholita funebrana* is one of the most important lepidopteran pests of stone-fruit in Europe and its exclusion from North America is a top priority. *Grapholita* larvae are commonly intercepted at U.S. ports of entry on a variety of products originating from Europe, Central America, and South America.

Grapholita delineana **(Walker)**

Nomenclature

Grapholita delineana (Walker) (Tortricidae: Olethreutinae: Grapholitini)

Synonyms: *apicatana* (*Grapholita*), *mundana* (*Grapholitha*), *quadristriana* (*Laspeyresia*), *sinana* (*Grapholitha*), *terstrigana* (*Grapholitha*), *tetragrammana* (*Grapholitha*)

Adult Recognition

Adults are dark brown with two pair of white dorsal strigulae on each forewing. FWL: 4.5-6.5 mm. Hindwings are dark brown. Male genitalia are characterized by an aedeagus that narrows abruptly. Female genitalia are characterized by two signa in the corpus bursae.

Wing pattern is similar to other *Grapholita*, especially *G. tristrigana* in eastern North America. The dorsal strigulae in *G. delineana* are usually narrow and distinctly separated, while those in *G. tristrigana* may be confluent. The two species can be separated by genitalia: in *G. tristrigana* the male aedeagus tapers evenly and females lack a signum in the corpus bursae (Gilligan et al. 2008).

Larval Morphology

Larvae are similar to those of other *Grapholita*. An anal comb is absent. Komai (1999) provides a complete setal map.

Biology

Grapholita delineana completes 2-3 generations per year. Adults are present in mid-May to August in North America. Females lay eggs on the leaves and stems of the host plant. Larvae feed in the stems, on developing seeds, and also on flowers. Infested plants show a swelling in the stem and possibly and entrance hole with extruded frass. Pupation occurs in the soil. Larvae can be pests on marijuana or hop (Cannabaceae) and have also been recorded on knotweed (Polygonaceae) (Table 99) (Gilligan et al. 2008, Miller 1982).

Table 99: *Grapholita delineana* host plants

Family	**Genus/species**	**Common name**
Cannabaceae	*Cannabis sativa* L.	marijuana
Cannabaceae	*Humulus japonicus* Siebold & Zucc.	Japanese hop
Cannabaceae	*Humulus lupulus* L.	common hop
Polygonaceae	*Polygonum* L.	knotweed

Distribution

Grapholita delineana is a native of East Asia that has spread into Europe, Asia Minor, Transcausasia, and North America along with the commercial production of hemp. It was first recorded from North America in 1943 and it is currently distributed from New York west to Minnesota and south to Missouri (Gilligan et al. 2008, Miller 1982).

***Grapholita funebrana* (Treitschke)**

Nomenclature

Grapholita funebrana (Treitschke) (Tortricidae: Olethreutinae: Grapholitini)

Common names: plum fruit moth, red plum maggot

Synonyms: *cerasana* (*Laspeyresia*)

Grapholita funebrana is often referred to as *Cydia funebrana* in older literature. Komai (1999) provides a taxonomic history of the Grapholitini.

Adult Recognition

Forewings are grayish brown with indistinct silvery-gray markings on the dorsum and around the poorly-defined ocellus. FWL: 4.0-7.5 mm. Hindwings are brown. Male genitalia are characterized by a thornlike projection off the ventral margin of the valva. Female genitalia are characterized by a rounded sterigma, an irregular sclerite in the ductus bursae, and two long, curved, thornlike signa in the corpus bursae.

Adults are similar to other species of *Grapholita*, including *G. molesta* and *G. tenebrosana*; a genitalic dissection may be necessary to confirm species identity, especially if individuals are recovered from sticky traps. In *G. molesta*, males lack the thornlike projection off the ventral margin of the valva and the female sterigma is laterally elongate with small posterolateral projections. In *G. tenebrosana*, the male valva is more elongate with the anal angle sharply pronounced and the female sterigma is large with triangular lateral lobes.

Pheromone traps using "Funemone" lures are commonly used to trap male *G. funebrana*. Alford (1978) provided trapping data for "Funemone" traps operating in England over the course of three years. Non-target tortricids attracted to the "Funemone" pheromone include: *Cnephasia stephensiana*, *Cnephasia* sp., *Celypha rufana*, *Hedya nubiferana*, *Pammene argyrana*, *Pammene fasciana*, and *G. tenebrosana*. As the lure is not species-specific, it will also attract other species of *Grapholita*, including *G. molesta*.

Larval Morphology

Late instar larvae are approximately 9-12 mm in length with a bright reddish-pink abdomen. The head is dark brown and the prothoracic shield is yellowish brown. The anal shield is light brown with dark mottling. An anal comb is present with 4-10 small teeth. Early instars are whitish with a black head and prothoracic shield (Bradley et al. 1979).

Larvae may appear similar to those of many other species of *Grapholita* and *Cydia*. *Cydia pomonella* larvae can be separated from *G. funebrana* by the absence of an anal fork. Other species of *Grapholita* cannot be reliably separated from *G. funebrana* based solely on larval morphology. Chen and Dorn (2009) provide a molecular assay to distinguish *G. funebrana* larvae from similar species using a polymerase chain reaction-restriction fragment length polymorphism (PCR-RFLP) analysis.

Biology

The following life history information is summarized from Bradley et al. (1979), Dickler (1991), and Komai (1999). *Grapholita funebrana* completes 1-3 generations per year; two generations are most common over most of its range. Adults are present from late May to September.

First generation females lay eggs singly on fruitlets. Second generation females lay eggs near the base of maturing fruit. Larvae tunnel into the fruit and feed inside. Last instar larvae bore out of the fruit and overwinter in a cocoon spun on tree bark or in the soil. Pupation occurs the following spring. Larvae of the second generation cause the most damage to fruits such as plum that mature in mid- to late summer.

Grapholita funebrana is one of the most important lepidopteran pests of fruit in Europe. Larvae can cause significant damage to apricot, cherry, peach, plum, and other *Prunus* species (Table 100).

Distribution

A native of Europe, *G. funebrana* has spread to most other fruit-growing regions of the Palearctic. It is currently present from Europe and northern Africa across Asia Minor and Central Asia to China, Korea, and Japan (Dickler 1991, Razowski 2003).

Table 100: *Grapholita funebrana* host plants

Family	**Genus/species**	**Common name**
Rosaceae	*Prunus armeniaca* L.	apricot
Rosaceae	*Prunus avium* (L.) L.	sweet cherry
Rosaceae	*Prunus domestica* L.	European plum
Rosaceae	*Prunus domestica* L. var. *insititia* (L.) Fiori & Paoletti	European plum
Rosaceae	*Prunus fruticosa* Pall.	European dwarf cherry
Rosaceae	*Prunus japonica* Thunb.	Japanese bush cherry
Rosaceae	*Prunus* L.	
Rosaceae	*Prunus maximowiczii* Rupr.	Korean cherry
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Prunus spinosa* L.	blackthorn
Rosaceae	*Pyrus ussuriensis* Maxim.	
Rosaceae	*Rosa davurica* Pall.	Amur rose

***Grapholita interstinctana* (Clemens)**

Nomenclature

Grapholita interstinctana (Clemens) (Tortricidae: Olethreutinae: Grapholitini)

Common names: clover seed caterpillar

Synonyms: *distema* (*Grapholitha*), *scitana* (*Dichrorampha*)

Adult Recognition

Forewings are dark brown with a single pair of white dorsal strigulae per wing. FWL: 4.0-6.0 mm. Male genitalia are characterized by the valvae, which have a straight costal margin and a V-shaped ventral invagination at the neck.

Adults appear similar to many other species of *Grapholita*. In eastern North America, *G. fana* is similar, but the two species are easily distinguished by the larger ocellus in *G. fana*. Genitalia of the two species are different (Gilligan et al. 2008).

Larval Morphology

Late instar larvae are approximately 5-6 mm in length with a pale abdomen. The head and prothoracic shield are yellowish brown. An anal comb is absent (MacKay 1959).

Biology

Grapholita interstinctana completes 2-3 generations per year. Adults fly from late May to September. Larvae feed on seeds in the heads of clover; usually one larva is present per flower head. Overwintering occurs as a mature larva and pupation occurs in the spring (Wehrle 1924).

In the early part of the 20th century, *G. interstinctana* was considered a significant pest of clover. Larvae have also been reported feeding on sunflower (Asteraceae) (Table 101) (Folsom 1909, Wehrle 1924).

Table 101: *Grapholita interstinctana* host plants

Family	**Genus/species**	**Common name**
Asteraceae	*Helianthus* L.	sunflower
Fabaceae	*Trifolium incarnatum* L.	crimson clover
Fabaceae	*Trifolium* L.	clover

Distribution

Grapholita interstinctana is distributed in eastern North America from Maine to Alberta, south to Missouri (Gilligan et al. 2008).

***Grapholita molesta* (Busck)**

Nomenclature

Grapholita molesta (Busck) (Tortricidae: Olethreutinae: Grapholitini)

Common names: oriental fruit moth

Adult Recognition

Forewings are dull grayish brown with a row of black dots near the apex and termen. FWL: 5.0-6.5 mm. Male genitalia are characterized by an elongate valva with rounded cucullus. Female genitalia are characterized by rectangular lateral extensions of the sterigma with sharply pointed posterolateral projections.

Adults are similar to other species of *Grapholita*, including *G. funebrana*, *G. libertina*, *G. tenebrosana*, and several others. A genitalic dissection may be necessary to confirm species identity, especially if individuals are recovered from sticky traps. Males of *G. funebrana* can be distinguished by the thornlike projection off the ventral margin of the valva, which is lacking in *G. molesta*. Gilligan et al. (2008) provide illustrations of male and female genitalia for many common Nearctic *Grapholita*. Synthetic pheromones are not species-specific, and *G. molesta* lures will attract other species of *Grapholita*, including *G. funebrana*.

Larval Morphology

Last instar larvae are approximately 10-12 mm in length with a pinkish abdomen and large pale pinacula. The head and prothoracic shield are yellowish brown. The anal shield is light brown without mottling. An anal comb is present with ca. 5 teeth (MacKay 1959). Early instars are assumed to be whitish with a black head and prothoracic shield.

Larvae may appear similar to those of many other species of *Grapholita* and *Cydia*. *Cydia pomonella* larvae can be separated from *G. molesta* by the absence of an anal fork. Other species of *Grapholita* cannot be reliably separated from *G. molesta* based solely on larval morphology. Chen and Dorn (2009) provide a molecular assay to distinguish *G. molesta* larvae from similar species using a polymerase chain reaction-restriction fragment length polymorphism (PCR-RFLP) analysis.

Biology

The following life history information is summarized from Chapman and Lienk (1971), Gilligan et al. (2008), and Rothschild and Vickers (1991). *Grapholita molesta* completes 3-7 annual generations; the exact number depends on temperature (latitude). In midwestern North America, adults are present from early May to late September. In southern locations, adults may be present year-round.

Females lay eggs singly on smooth surfaces of the host plant, which usually includes leaves, shoots, and twigs. Early instars tunnel into shoots or pedicels. Later instars continue feeding in shoots or tunnel into fruit. The final instar leaves the fruit or shoot and constructs a cocoon on the tree or in leaf litter. Larvae complete 4-5 instars. Overwintering occurs as a prepupa and pupation occurs in the spring for the overwintering generation. Larval damage is characterized by dead and wilting shoots and injured fruit. Injured fruit may fall early and is more prone to secondary infection by fungus.

Grapholita molesta is an important pest of stone-fruit crops throughout the world. Most economic damage occurs in peach and nectarine, or when other fruit crops are grown adjacent to peach (Rothschild and Vickers 1991). In addition to the Rosaceae, larvae have been recorded feeding on plants in several families (Table 102).

Distribution

Grapholita molesta is thought to have originated in northwest China. The first North American records are from 1913-1915. It is currently widely distributed on all continents where stone-fruit is grown (Rothschild and Vickers 1991).

Table 102: *Grapholita molesta* host plants

Family	**Genus/species**	**Common name**
Cornaceae	*Cornus* L.	dogwood
Ebenaceae	*Diospyros kaki* L. f.	Japanese persimmon
Myrtaceae	*Hexachlamys edulis* (O. Berg) Kausel & D. Legrand	
Myrtaceae	*Psidium guajava* L.	guava
Rosaceae	*Chaenomeles* Lindl.	flowering quince
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Cydonia oblonga* Mill.	quince
Rosaceae	*Eriobotrya japonica* (Thunb.) Lindl.	loquat
Rosaceae	*Malus* Mill.	apple

Table 102: *Grapholita molesta* host plants

Family	**Genus/species**	**Common name**
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Malus sylvestris* (L.) Mill.	European crab apple
Rosaceae	*Photinia glabra* (Thunb.) Maxim.	Japanese photinia
Rosaceae	*Prunus armeniaca* L.	apricot
Rosaceae	*Prunus avium* (L.) L.	sweet cherry
Rosaceae	*Prunus cerasus* L.	sour cherry
Rosaceae	*Prunus dulcis* (Mill.) D. A. Webb	sweet almond
Rosaceae	*Prunus ilicifolia* (Nutt. ex Hook. & Arn.) D. Dietr.	hollyleaf cherry
Rosaceae	*Prunus* L.	[various]
Rosaceae	*Prunus mume* Siebold & Zucc.	Japanese apricot
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Prunus salicina* Lindl.	Japanese plum
Rosaceae	*Prunus serrulata* var. *spontanea* (Maxim.) E. H. Wilson	
Rosaceae	*Pyracantha* M. Roem.	firethorn
Rosaceae	*Pyrus communis* L.	common pear
Rosaceae	*Pyrus* L.	pear
Rosaceae	*Pyrus pyrifolia* (Burm. f.) Nakai	Chinese pear
Rosaceae	*Rosa* L.	rose
Sapindaceae	*Litchi chinensis* Sonn.	lychee

Grapholita packardi **(Zeller)**

Nomenclature

Grapholita packardi (Zeller) (Tortricidae: Olethreutinae: Grapholitini)

Common names: cherry fruit worm

Synonyms: *pyricolana* (*Steganoptycha*)

Adult Recognition

Adults range in color from light to dark. Light individuals have silvery striae and darker contrasting fasciae while dark individuals lack a well-defined forewing pattern. FWL: 3.5-5.0 mm. Males have a conspicuous patch of dark sex scales on the dorsal surface of the hindwing. Male genitalia are characterized by the unconstricted valval neck, which is nearly as wide as the cucullus. Female genitalia are characterized by a triangular ostium.

Dark individuals may appear similar to *G. molesta*, but are generally much smaller. In western North America, *G. libertina* is similar in appearance to dark *G. packardi* in both forewing pattern and male genitalia, but *G. libertina* males lack a patch of dark sex scales on the hindwing (Heinrich 1926).

Larval Morphology

Late instar larvae are approximately 8-9 mm in length with a pale-reddish abdomen. The head is yellowish brown with darker mottling. The prothoracic and anal shields are brown. Pinacular are moderately large. An anal comb is present with 4-6 teeth (MacKay 1959).

Larvae may appear similar to those of many other species of *Grapholita* and *Cydia*. *Cydia pomonella* larvae can be separated from *G. packardi* by the absence of an anal fork and their larger size. Larvae of *G. molesta* are similar but generally larger. Larvae of *G. packardi* and *G. prunivora* are very similar, although the pinacula on the 8th and 9th abdominal segment are more prominent in *G. packardi*, and the abdomen of *G. prunivora* is more reddish, especially in preserved specimens (Chapman and Lienk 1971, MacKay 1959).

Biology

Grapholita packardi completes 2-3 generations per year. Adults are present in May and June. Females lay eggs singly on terminal shoot leaves. Larvae feed inside the shoots or fruit; Chapman and Lienk (1971) found considerable variation in feeding records suggesting that larvae behave differently on different hosts or that a species complex is involved. Overwintering occurs on the host in a cocoon and pupation occurs the following spring. Larval damage is characterized by injured fruit, early fruit drop, and/or stunted or dead shoots (Weires and Riedel 1991).

Larvae of *G. packardi* feed on many common fruit crops in the families Rosaceae and Ericaceae (Table 103). It is possible that hawthorn (*Crataegus*) is the native host (Chapman and Lienk 1971).

Table 103: *Grapholita packardi* host plants

Family	Genus/species	Common name
Ericaceae	*Vaccinium* L.	blueberry
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Malus pumila* Mill.	paradise apple
Rosaceae	*Prunus domestica* L.	European plum
Rosaceae	*Prunus* L.	
Rosaceae	*Prunus serotina* Ehrh.	black cherry
Rosaceae	*Pyracantha* M. Roem.	firethorn
Rosaceae	*Pyrus communis* L.	common pear
Rosaceae	*Rosa* L.	rose

Distribution

Grapholita packardi is widely distributed in eastern North America. It is also present in the Pacific Northwest (Washington and British Columbia) (E. LaGasa, pers. comm.) and likely other fruit-growing regions of the West.

Grapholita prunivora (Walsingham)

Nomenclature

Grapholita prunivora (Walsingham) (Tortricidae: Olethreutinae: Grapholitini)

Common names: lesser apple worm

Adult Recognition

Forewing pattern is distinctive with pale-yellow costal strigulae, silvery striae, a line of yellow scales preceeding the black terminal line, and a well-developed ocellus with 3-4 black dashes. FWL: 4.0-5.5 mm. The male valva is characterized by the angular apex and evenly rounded anal angle of the cucullus. Female genitalia are characterized by a ringlike sterigma with weakly sclerotized lateral extensions (Gilligan et al. 2008).

This is one of the smallest North American tortricids. Fresh specimens are unlikely to be confused with any other species; worn specimens can be dissected to confirm identity.

Larval Morphology

Late instar larvae are approximately 7.5-9.5 mm in length with a pale-reddish abdomen. The head is yellowish brown with darker mottling. The prothoracic and anal shields are brown; the prothoracic shield may have some dark posterolateral markings. Pinacular are moderately large. An anal comb is present with 4-6 teeth (MacKay 1959).

Larvae may appear similar to those of many other species of *Grapholita* and *Cydia*. *Cydia pomonella* larvae can be separated from *G. prunivora* by the absence of an anal fork and their larger size. Larvae of *G. molesta* are similar but generally larger. Larvae of *G. packardi* and *G. prunivora* are very similar, although the pinacula on the 8th and 9th abdominal segment are more prominent in *G. packardi*, and the abdomen of *G. prunivora* is more reddish, especially in preserved specimens (MacKay 1959).

Biology

Grapholita prunivora completes two annual generations over much of its range. Adults are present in May to June and again in August. Females lay eggs singly on young fruits or on the upper surface of leaves. Lavae tunnel into the fruit at the calyx end and feed inside. In apple, larvae may feed directly under the skin of the fruit, creating a blotchy mine. Overwintering

Table 104: *Grapholita prunivora* host plants

Family	Genus/species	Common name
Rosaceae	*Amelanchier* Medik.	serviceberry
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Photinia* Lindl.	chokeberry
Rosaceae	*Prunus* L.	[various]
Rosaceae	*Prunus pensylvanica* L. f.	pin cherry
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Prunus salicina* Lindl.	Japanese plum
Rosaceae	*Pyrus communis* L.	common pear

occurs as a mature larva and pupation occurs in the spring (Weires and Riedel 1991). In addition to feeding on various stone-fruits (Table 104), larvae of *G. prunivora* have also been recorded feeding inside galls (MacKay 1959).

Distribution

Grapholita prunivora is found throughout southern Canada and the continental U.S.; it is absent from the far South (Chapman and Lienk 1971, Gilligan et al. 2008).

Gymnandrosoma **Dyar**

Overview

Gymnandrosoma consists of seven species, all restricted to the New World. Adults are dark brown to black, usually with indistinct markings. Diagnostic genitalic characters include: male with uncus and socii undeveloped, valva with moderately constricted neck, cucullus with rounded apex and short stout setae evenly distributed along distal margin, and medial surface with ridge of stout setae near ventral margin from neck to anal angle; female with two large thornlike signa in the corpus bursae and extensive wrinkling of the membrane near the juncture with the ductus bursae. Most males have secondary sex scaling on the abdomen, terga, hind tibia, and/or anal margin on the hindwing. Adamski and Brown (2001) provide the most recent revision of the genus.

Larvae are similar to other species in the *Cryptophlebia-Ecdytolopha* group, with an enlarged L-pinaculum on the prothorax that extends beneath (and usually beyond) the spiracle. Larvae of *Gymnandrosoma* can be separated from those of *Ecdytolopha* by the distance between the V setae on A9: approximately the same as the distance between Vs on A8 in *Ecdytolopha* and 1.2-2.0 times the distance between Vs on A8 in *Gymnandrosoma* (Adamski and Brown 2001).

Gymnandrosoma larvae (including *G. aurantianum*) are intercepted frequently on a variety of imports from Central America, South America, and the Caribbean. As most species were placed in Ecdytolopha prior to Adamski and Brown's revison (2001), many interception records for *Gymnandrosoma* are listed under *Ecdytolopha*.

***Gymnandrosoma aurantianum* Lima**

Nomenclature

Gymnandrosoma aurantianum Lima (Tortricidae: Olethreutinae: Grapholitini)

Synonyms: *cnemoptila* (*Acharneodes*), *pithecolobiae* (*Gymnandrosoma*), *sideropetra* (*Argyroploce*), *sideroptera* (*Gymnandrosoma*), *torticornis* (*Argyroploce*)

Adult Recognition

Adults are brown with indistinct reddish-brown and black markings. A conspicuous white dot is present on the distal one-third of the forewing in most individuals. FWL: 6.0-10.5 mm. Male genitalia are characterized by a dense row of ca. 130 deciduous cornuti in the vesica. Female genitalia are characterized by a cestum in the ductus bursae that is closer to the ostium than the ductus seminalis. The male antenna is flattened and notched at the base.

Males can be separated from other species of *Gymnandrosoma* by the flattened, notchlike basal portion of the antenna and large hairpencil on the hind tibia (Adamski and Brown 2001). Females may need to be dissected to confirm identity.

Larval Morphology

Late instar larvae are approximately 16-19 mm in length. Abdominal pinacula are large and well defined. The head is pale yellow to pale orange and the prothoracic shield is pale yellow, usually without darker mottling. Other larval characters include: distance between V setae on A9 1.5 times the distance between V setae on A8; SV pinacula on A9 unisetose; and 40-50 crochets on the abdominal prolegs (Adamski and Brown 2001).

Larvae are similar to other species in the *Cryptophlebia-Ecdytolopha* group, with an enlarged L-pinaculum on the prothorax that extends beneath (and usually beyond) the spiracle. Larvae of *Gymnandrosoma* can be separated from those of *Ecdytolopha* by the distance between the V setae on A9: approximately the same as the distance between Vs on A8 in *Ecdytolopha* and 1.2-2.0 times the distance between Vs on A8 in *Gymnandrosoma* (Adamski and Brown 2001).

Gymnandrosoma larvae (including *G. aurantianum*) are intercepted frequently on a variety of imports from Central America, South America, and the Caribbean. As most species were placed in *Ecdytolopha* prior to Adamski and Brown's revison (2001), interception records for *Ecdytolopha* could refer to species in either genus.

Biology

Gymnandrosoma aurantianum has a relatively short life cycle (36 days from egg to adult), allowing it to complete up to 10 generations per year if the appropriate host plants are available. Adults may be present year-round. Females lay eggs on mature fruit. Larvae tunnel into fruit and consume the seeds, or they may occasionally feed on leaves and stems. Pupation occurs in the soil. Larval damage to fruit may lead to secondary infection by fungus and bacteria. Larvae of *G. aurantianum* can be pests on cultivated macadamia, citrus, and other tropical fruits (Table 105) (Adamski and Brown 2001, Blanco-Metzler et al. 2009).

Distribution

Gymnandrosoma aurantianum is found in Central America, South America, and the Caribbean (Adamski and Brown 2001).

Table 105: *Gymnandrosoma aurantianum* host plants

Family	**Genus/species**	**Common name**
Annonaceae	*Annona cherimola* L. X *squamosa* L.	
Euphorbiaceae	*Plukenetia volubilis* L.	
Fabaceae	*Cojoba arborea* (L.) Britton & Rose	
Fabaceae	*Pithecellobium dulce* (Roxb.) Benth.	monkeypod
Myrtaceae	*Psidium guajava* L.	guava
Oxalidaceae	*Averrhoa carambola* L.	carambola
Proteaceae	*Macadamia integrifolia* Maiden & Betche	macadamia nut
Punicaceae	*Punica granatum* L.	pomegranate
Rosaceae	*Eriobotrya japonica* (Thunb.) Lindl.	loquat

Table 105: *Gymnandrosoma aurantianum* host plants

Family	**Genus/species**	**Common name**
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rutaceae	*Citrus* L.	citrus
Sapindaceae	*Cupania vernalis* A. St.-Hil.	
Sapindaceae	*Litchi chinensis* Sonn.	lychee
Sapindaceae	*Sapindus saponaria* L.	wingleaf soapberry
Stericulaceae	*Theobroma cacao* L.	cacao

***Gymnandrosoma punctidiscanum* Dyar**

Nomenclature

Gymnandrosoma punctidiscanum Dyar (Tortricidae: Olethreutinae: Grapholitini)

Adult Recognition

Adults are brown to blackish gray with indistinct markings. A conspicuous white dot is present on the distal one-third of the forewing. FWL: 8.0-12.0 mm. Male genitalia are characterized by a short row of ca. 25-30 deciduous cornuti in the vesica. Female genitalia are characterized by a cestum in the ductus bursae that is closer to the ductus seminalis than the ostium. Males have distinctive sex scaling on the dorsal surface of the abdomen.

Males can be separated from those of *G. aurantianum* by sex scaling on the dorsal surface of the abdomen and differences in genitalia. Two other species of *Gymnandrosoma*, *G. linaresensis* and *G. cryptotortanum*, have similar scale structures on the abdomen, but these species are only known to occur in Mexico (Adamski and Brown 2001).

Larval Morphology

Although not described, larvae are assumed to be similar to other species in the *Cryptophlebia-Ecdytolopha* group, with an enlarged L-pinaculum on the prothorax that extends beneath (and usually beyond) the spiracle.

Biology

Little is known regarding the life cycle of this species. Collection records suggest multiple generations per year. In the midwestern U.S., adults are present in May through September. In

Florida and southern locations, adults may be present year-round. Larvae of *Gymnandrosoma punctidiscanum* have been recorded feeding on locust and red oak (Table 106) (Gilligan et al. 2008).

Table 106: *Gymnandrosoma punctidiscanum* host plants

Family	Genus/species	Common name
Fabaceae	*Robinia* L.	locust
Fagaceae	*Quercus rubra* L.	red oak

Distribution

Gymnandrosoma punctidiscanum is distributed from Massachusetts and southern Ontario west to Wisconsin and south to Florida and Texas (Gilligan et al. 2008).

Pammene fasciana **(Linnaeus)**

Nomenclature

Pammene fasciana (Linnaeus) (Tortricidae: Olethreutinae: Grapholitini)

Common names: chestnut leafroller

Synonyms: *herrichiana* (*Carpocapsa*), *juliana* (*Philalcea*), *nimbana* (*Carpocapsa*), *trinotana* (*Tortrix*)

Adult Recognition

Forewings are white and gray with a large ocellus consisting of a leaden-metallic center bar surrounded by black and yellow dashes. Adults are not sexually dimorphic. Males lack a forewing costal fold. FWL: 6.5-8.5 mm. Male genitalia are characterized by a reduced uncus and several cornuti in the vescia. Female genitalia are characterized by two thornlike signa and a short ductus bursae with a sclerotized ring.

The majority of *Pammene* are Palearctic, with over 90 species present in the region (Razowski 2003). Only six species are present in the Nearctic and all can be distinguished from

P. fasciana by wing pattern. A genitalic dissection can be used to confirm the identity of questionable or worn individuals.

Larval Morphology

Late instar larvae are approximately 13-15 mm in length with a whitish gray-green to pale-yellow abdomen. The head is yellowish brown and the prothoracic shield is paler than the head and weakly sclerotized (Brown and Komai 2008).

Other tortricid *Castanea* pests include *Cydia fagiglandana*, *C. glandicolana*, *C. kurokoi*, *C. splendana*, and *Fibuloides aestuosa*. Brown and Komai (2008) provide a description and key to larvae of these species. *Pammene fasciana* larvae can be distinguished by the following characters: whitish abdomen with large, dark, conspicuous pinacula; SV setal counts on A1,2,7,8,9 as 3:3:2:2(1):1; three L setae on A9 on single pinaculum; distance between V setae on A9 greater than distance between Vs on A8; crochets on prolegs uniordinal (or irregularly biordinal); number of crochets on prolegs 30-33; anal comb present.

Biology

Pammene fasciana completes a single generation per year. Adults are present June-July. Males fly after sunrise and again in the afternoon; females also fly at night. Females lay eggs singly or in small groups along the veins on the leaves of the host plant. Larvae bore into nuts and feed inside. When the nuts fall from the tree, the larvae bore out of the nut and create a cocoon under tree bark or moss in which to overwinter. Pupation occurs the following spring. Larvae of *P. fasciana* are important pests of chestnut (*Castanea sativa*). Other preferred hosts include *Quercus* and *Fagus* (Table 107) (Bradley et al. 1979, Brown and Komai 2008, Razowski 2003).

Distribution

Pammene fasciana is distributed from Western Europe east to the Ukraine.

Table 107: *Pammene fasciana* host plants

Family	Genus/species	Common name
Fagaceae	*Castanea* Mill.	chestnut
Fagaceae	*Castanea sativa* Mill.	European chestnut
Fagaceae	*Fagus* L.	beech
Fagaceae	*Fagus sylvatica* L.	European beech
Fagaceae	*Quercus* L.	oak
Fagaceae	*Quercus suber* L.	cork oak
Fagaceae	*Quercus robur* L.	English oak

Talponia batesi **(Heinrich)**

Talponia batesi (Heinrich) (Tortricidae: Olethreutinae: Grapholitini)

Adult Recognition

Forewings are broadly triangular with a strongly arched costa, a row of black dots along the termen, and a conspicuous notch below the apex. The basal half of the wing is olive green while the apical half is extensively marked with metallic striations. Males lack a forewing costal fold. FWL: 5.0-6.5 mm.

The only other member of this genus, *T. plummeriana*, is recorded from the eastern half of the United States where it feeds on *Asimina* spp. (pawpaw); there are also unverified records of *T. plummeriana* from Mexico (Gomez and Carrasco 2008, Heinrich 1932). The two species are similar in appearance but *T. plummeriana* is easily distinguished from *T. batesi* by the reddish color on the apical half of the forewing. Genitalia of the two species are quite different and a dissection can be used to confirm identity.

Larval Morphology

Last instar larvae are approximately 8-10 mm in length with a whitish abdomen. The head is yellowish brown and the prothoracic shield is brown to yellowish brown. Talponia larvae can be distinguished by the large, single, brown pinaculum on A9 where the D1, D2, and SD1 setae are located. Other morphological features include: SV setal counts on A1, 2, 7, 8, 9 as

2:2:2:2:1 or 2:2:2:1:1; SD1 on A8 anterodorsal to spiracle; abdominal setae short; anal comb absent (J. Brown, pers. comm., MacKay 1959).

Biology

Females lay eggs on the flowers and fruits of *Annona*. Larvae bore into the fruit and feed inside until mature. Pupation occurs in leaves outside of the fruit. The number of generations per year is unknown although it is assumed that larval development is synchronized with fruit development (Heinrich 1932). This species is a serious pest of *Annona* (Table 108), and in Mexico crop losses on cherimoya range from 45-100% if larvae are not controlled (Gomez and Carrasco 2008). *Talponia batesi* is commonly intercepted at U.S. ports of entry on *Annona* originating from the Caribbean, Mexico, or Central America.

Table 108: *Talponia batesi* host plants

Family	**Genus/species**	**Common name**
Annonaceae	*Annona cherimola* Mill.	cherimoya
Annonaceae	*Annona glabra* L.	pond apple
Annonaceae	*Annona muricata* L.	soursop
Annonaceae	*Annona* L.	custard apple

Distribution

Talponia batesi has been recorded from the Caribbean, Mexico, and Central America (Gomez and Carrasco 2008, Heinrich 1932). Larvae are commonly intercepted at U.S. ports of entry on *Annona* originating in Guatemala or Mexico.

Thaumatotibia leucotreta **(Meyrick)**

Refer to Chapter 5 for information on *Thaumatotibia leucotreta*.

Plates

The following plates illustrate wing pattern variation and male and female genitalia for each of the species treated in the TortAI adult key. A complete list of the specimen data and slide numbers for the genitalic dissections is available at the following URL: http://idtools.org/id/leps/tortai/TortAI_Genitalia_slide_log.pdf.

Adults: Figures 1–29.

Male genitalia: Figures 30–43.

Female genitalia: Figures 44–60.

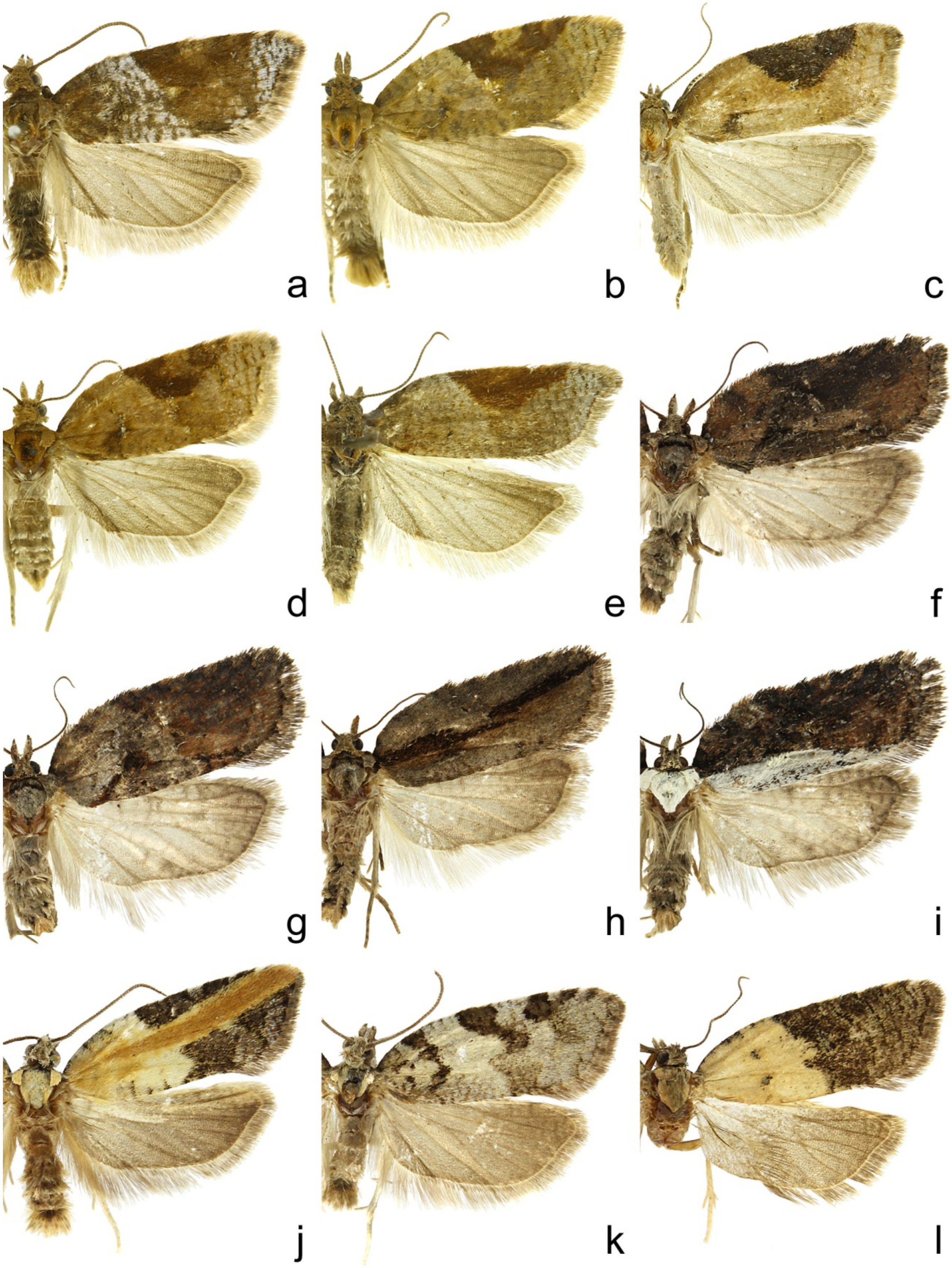


Figure 1. Tortricinae; a–e: *Acleris comariana*; f–i: *Acleris flavivittana*; j–l: *Acleris gloverana*.

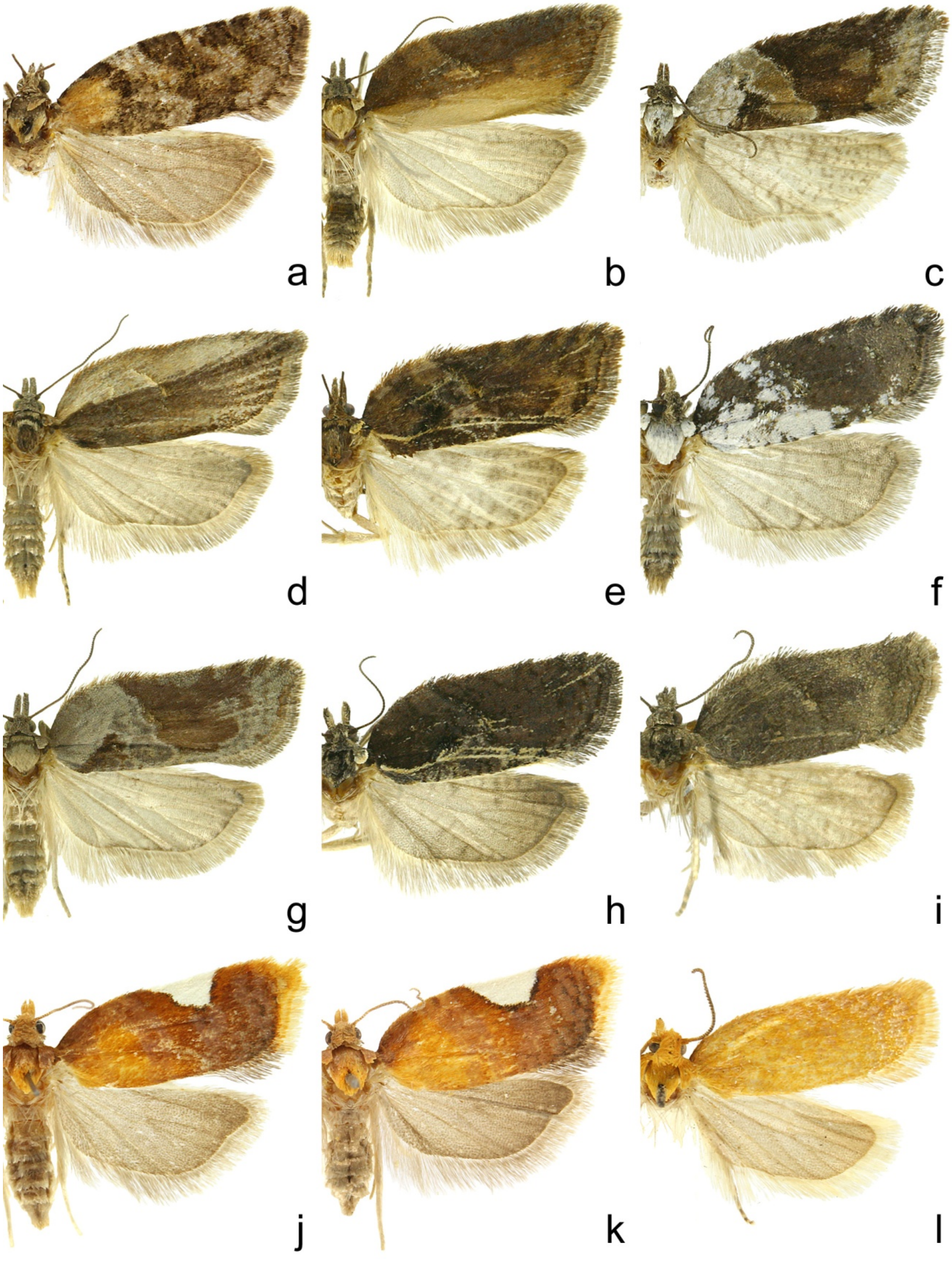


Figure 2. Tortricinae; a: *Acleris gloverana*; b–i: *Acleris hastiana*; j–k: *Acleris holmiana*; l: *Acleris minuta*.

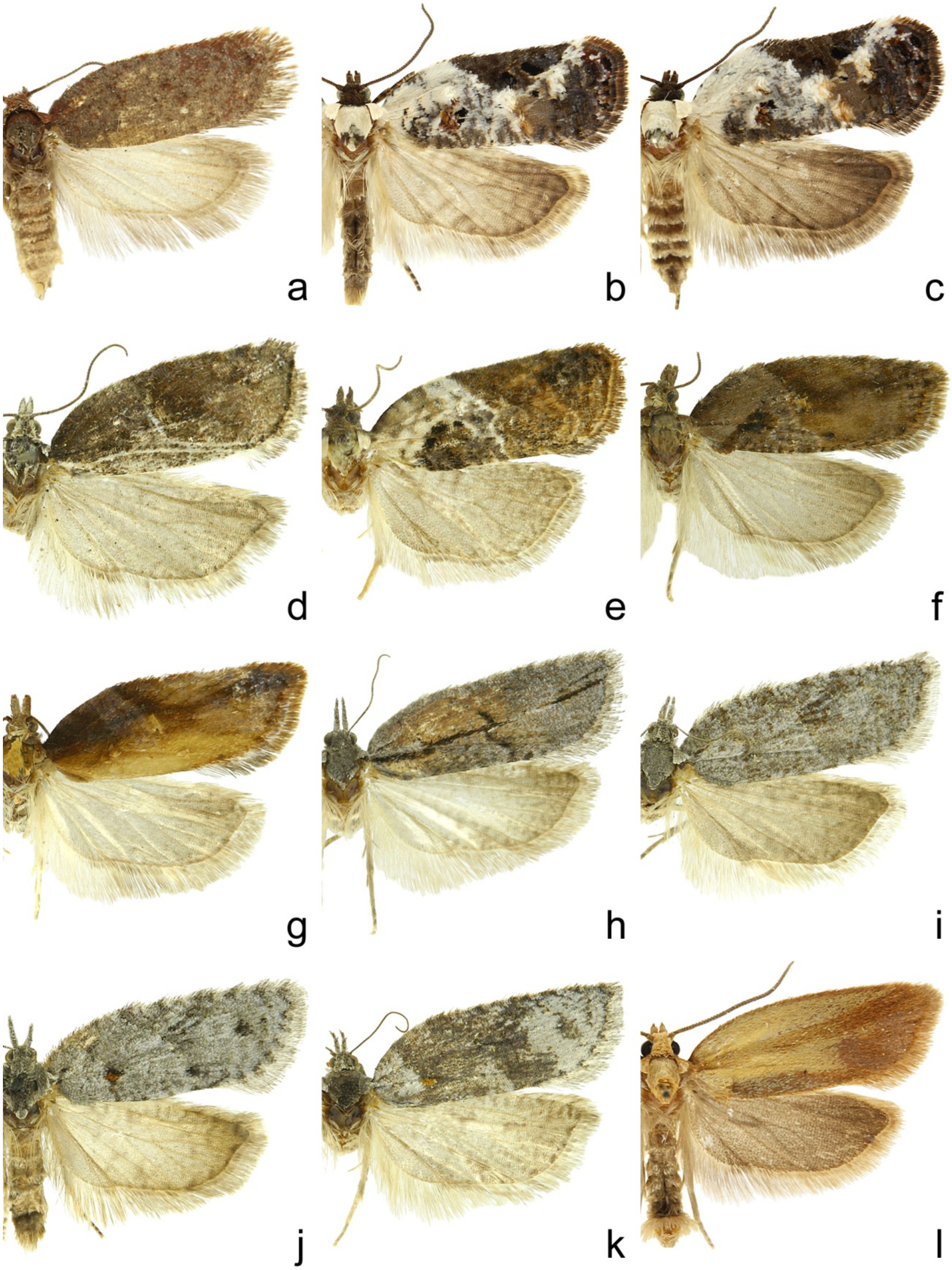


Figure 3. Tortricinae; a: *Acleris minuta*; b–c: *Acleris nivisellana*; d–g: *Acleris robinsoniana*; h–k: *Acleris senescens*; l: *Acleris variana*.

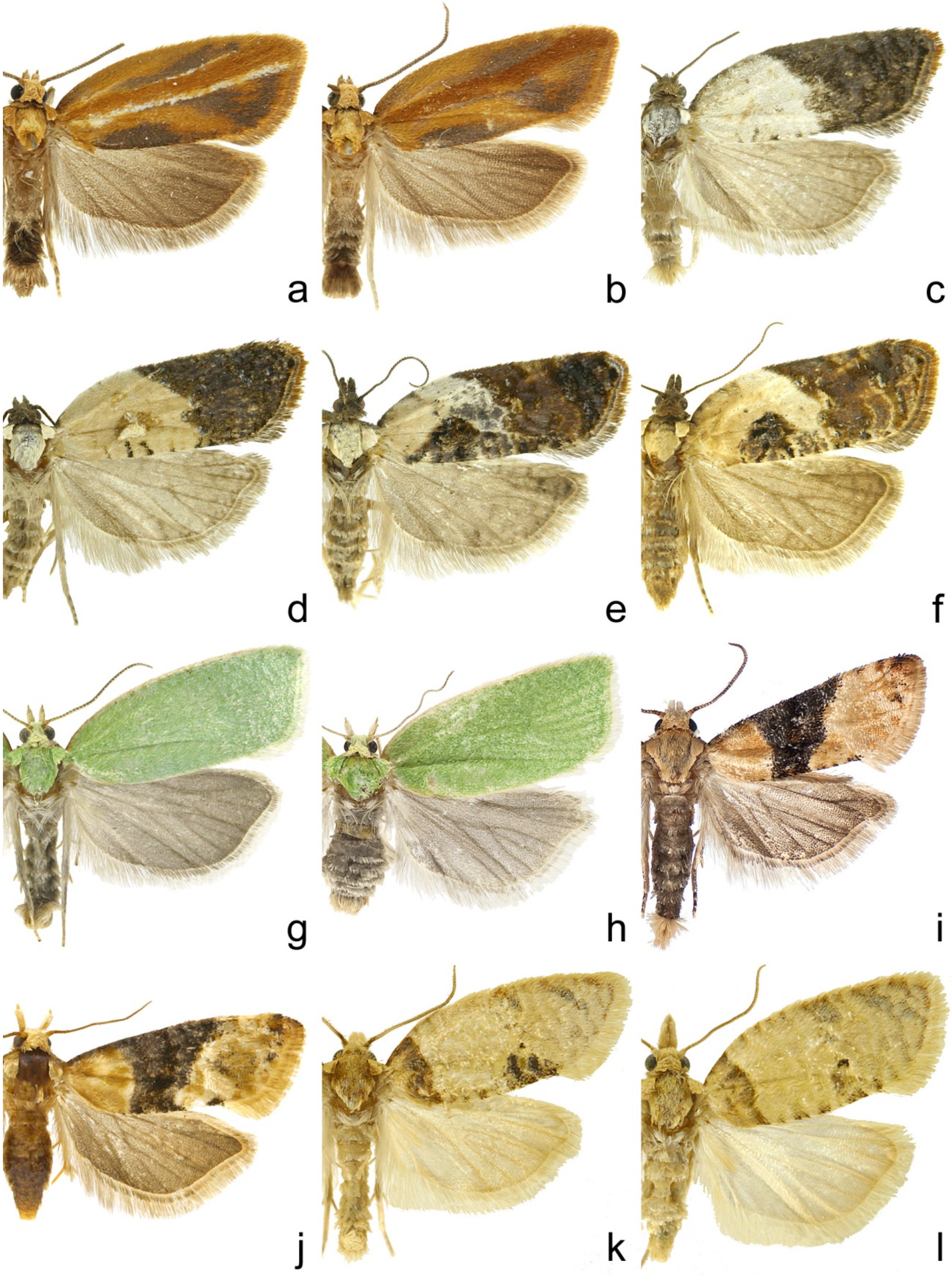


Figure 4. Tortricinae; a–b: *Acleris variana*; c–f: *Acleris variegana*; g–h: *Tortrix viridana*; i–j: *Eupoecilia ambiguella*; k–l: *Henricus umbrabasana*.

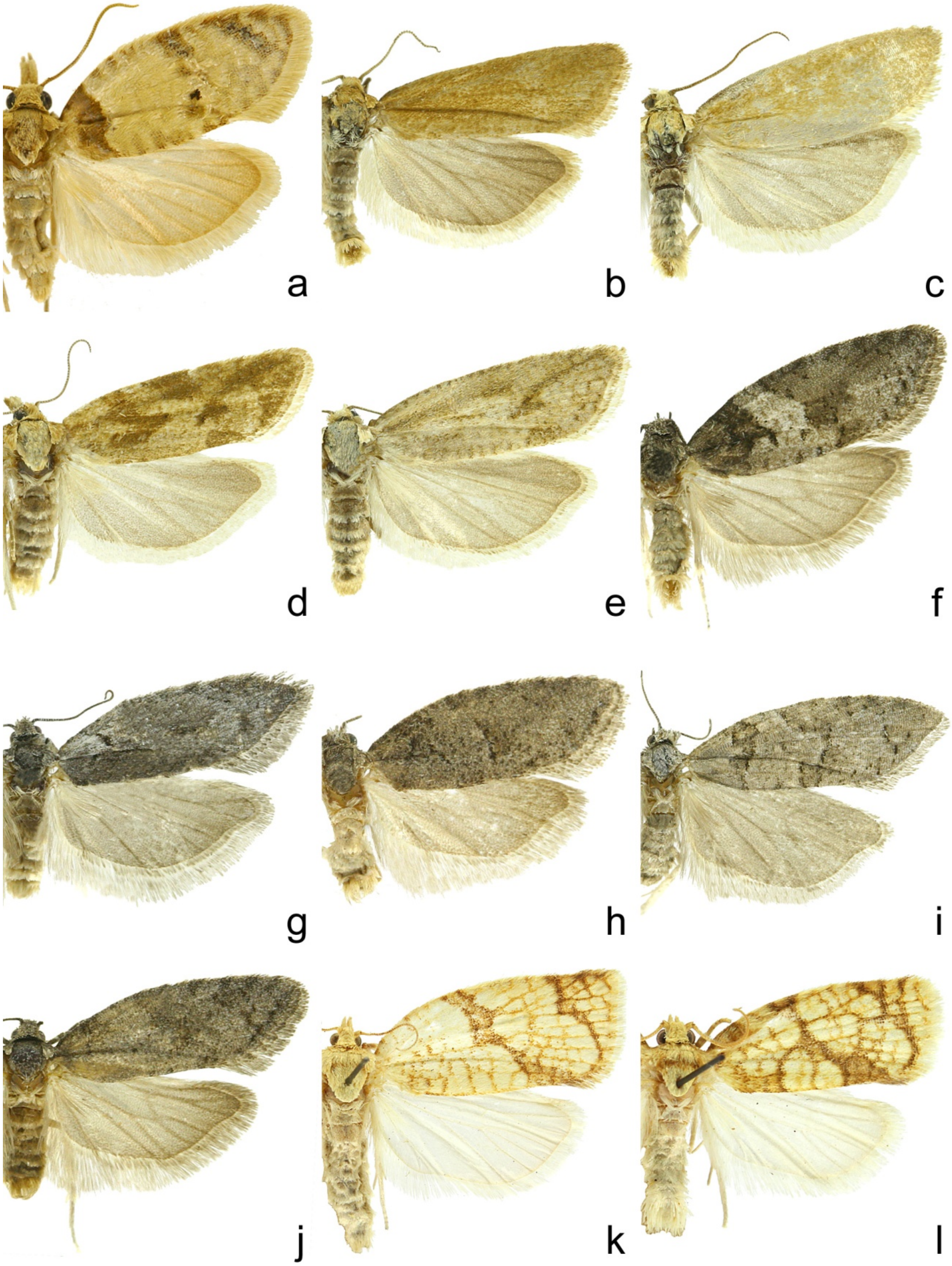


Figure 5. Tortricinae; a: *Henricus umbrabasana*; b–e: *Cnephasia longana*; f–g: *Decodes basiplagana*; h–j: *Decodes fragariana*; k–l: *Adoxophyes furcatana*.

Figure 6. Tortricinae; a: *Adoxophyes furcatana*; b–d: *Adoxophyes negundana*; e–j: *Adoxophyes orana*; k–l: *Archips argyrospila*.

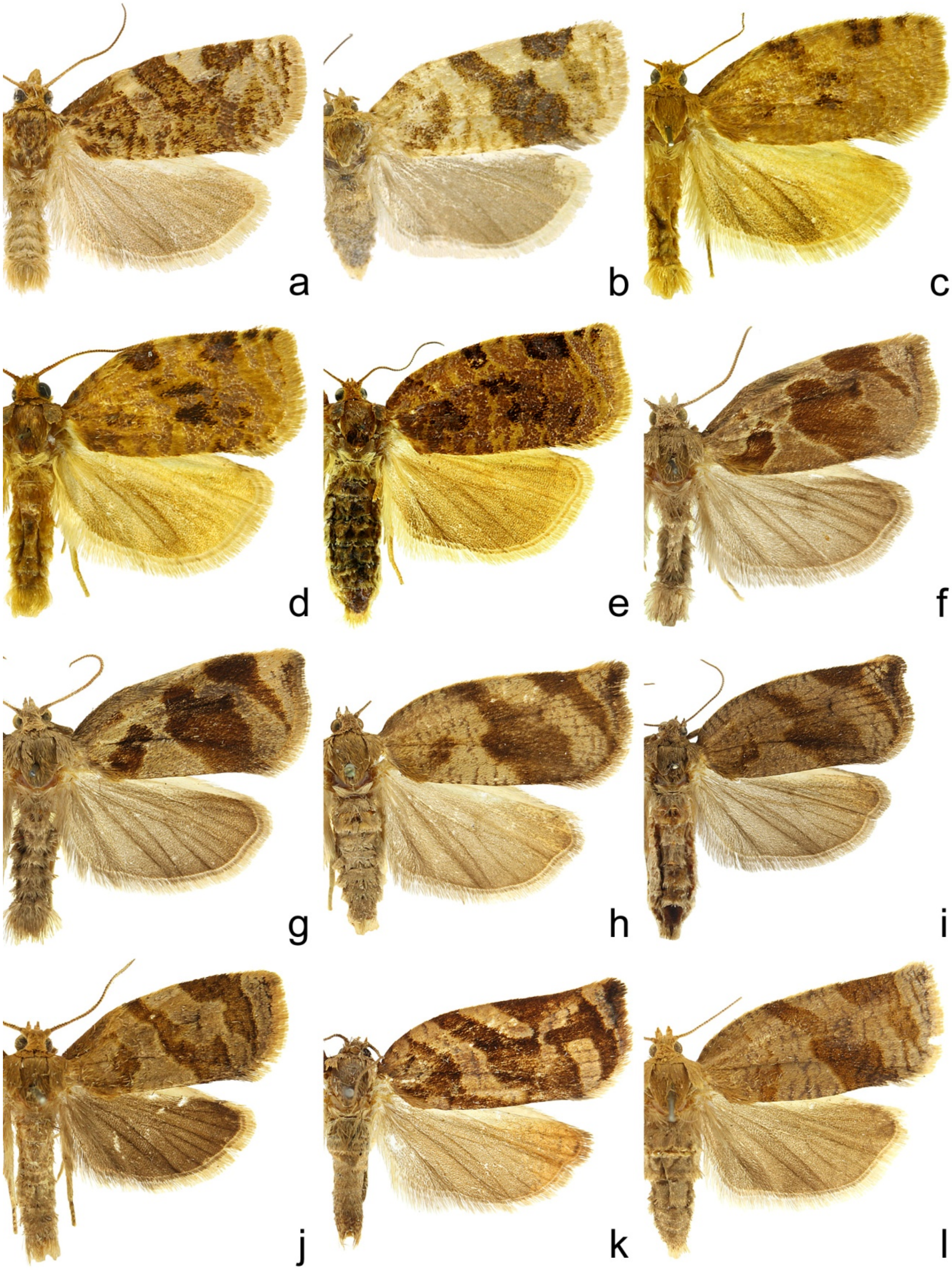


Figure 7. Tortricinae; a–b: *Archips argyrospila*; c–e: *Archips cerasivorana*; f–i: *Archips crataegana*; j–l: *Archips fuscocupreanus*.

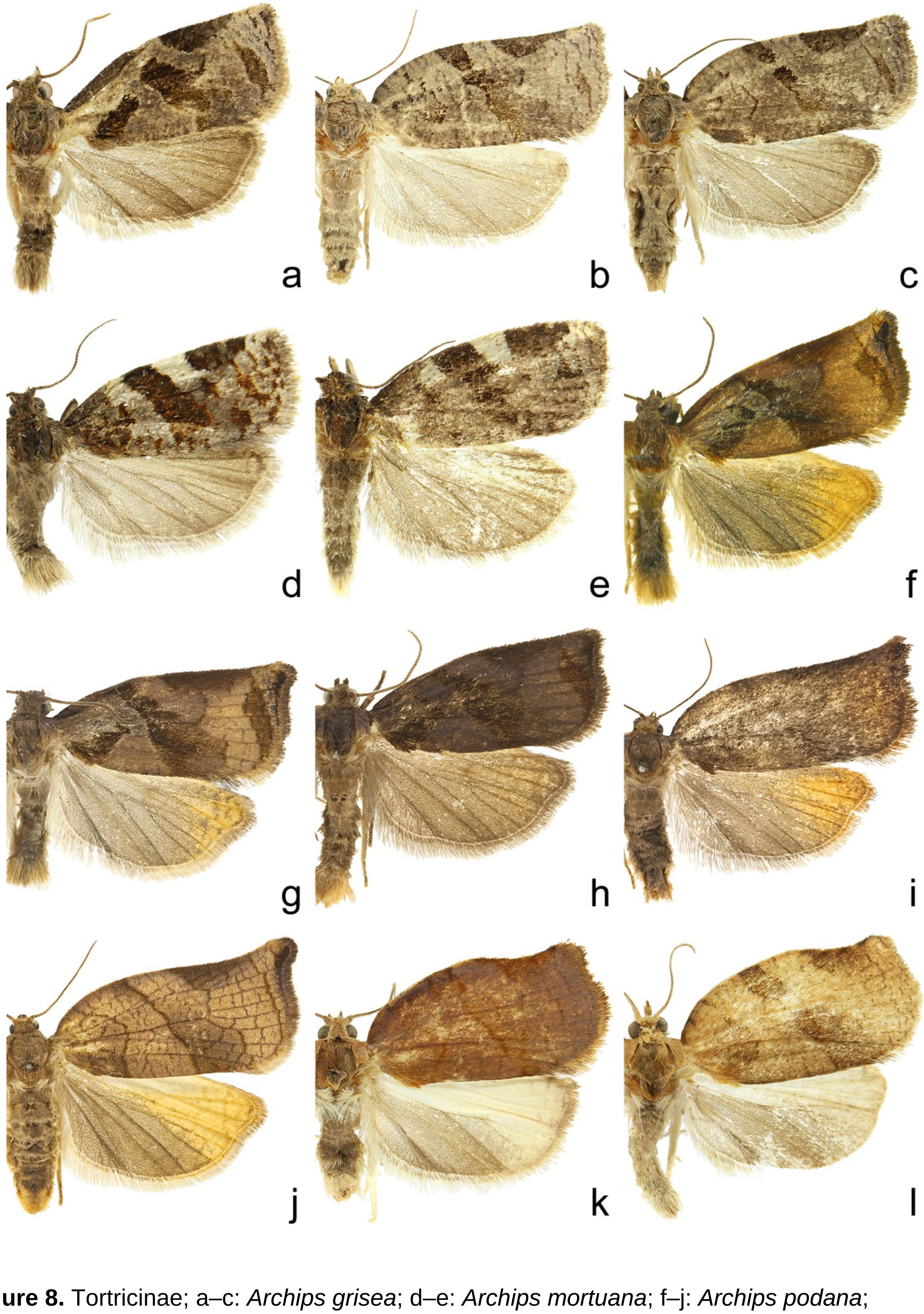


Figure 8. Tortricinae; a–c: *Archips grisea*; d–e: *Archips mortuana*; f–j: *Archips podana*; k–l: *Archips purpurana*.

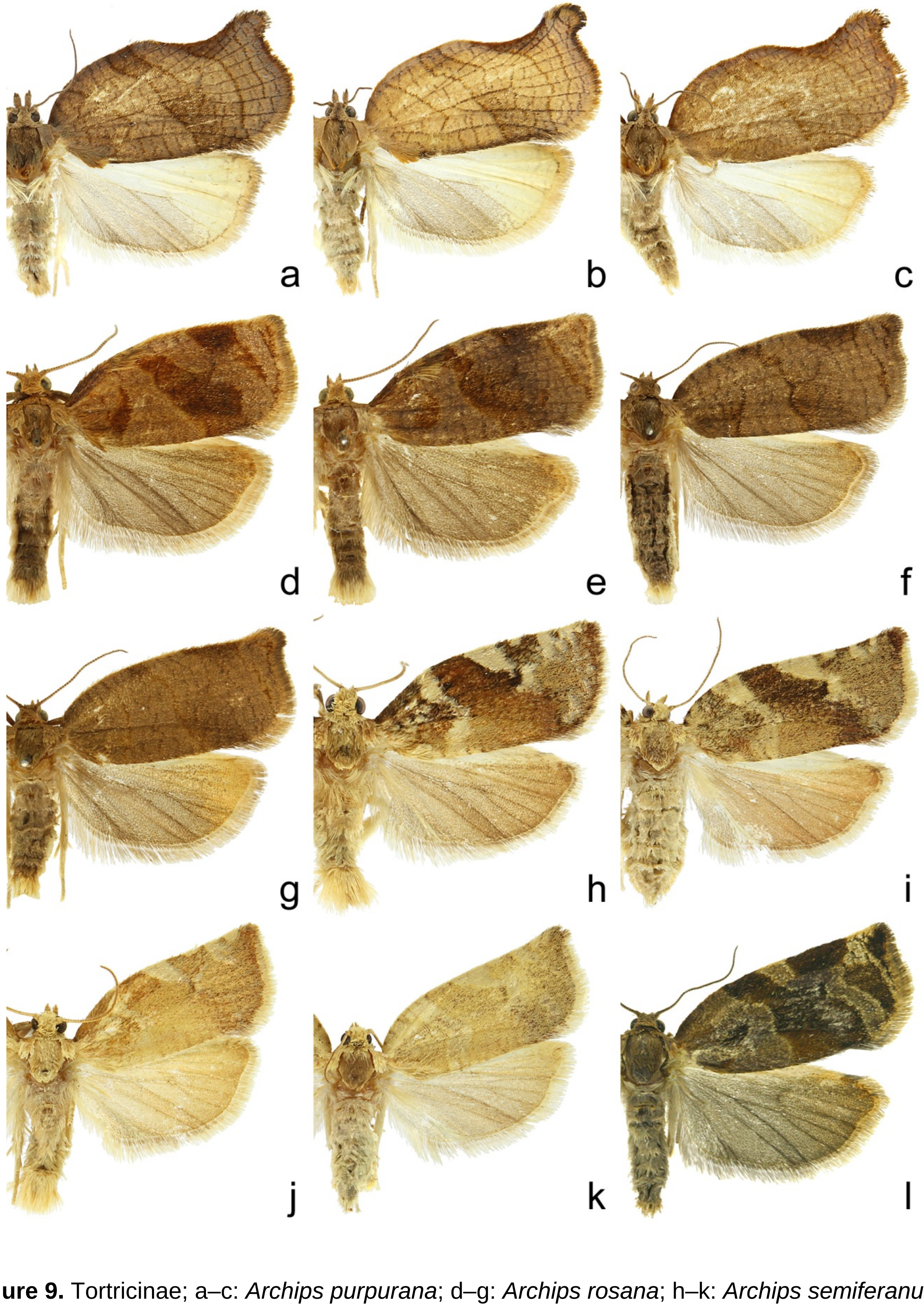


Figure 9. Tortricinae; a–c: *Archips purpurana*; d–g: *Archips rosana*; h–k: *Archips semiferanus*; l: *Archips xylosteana*.

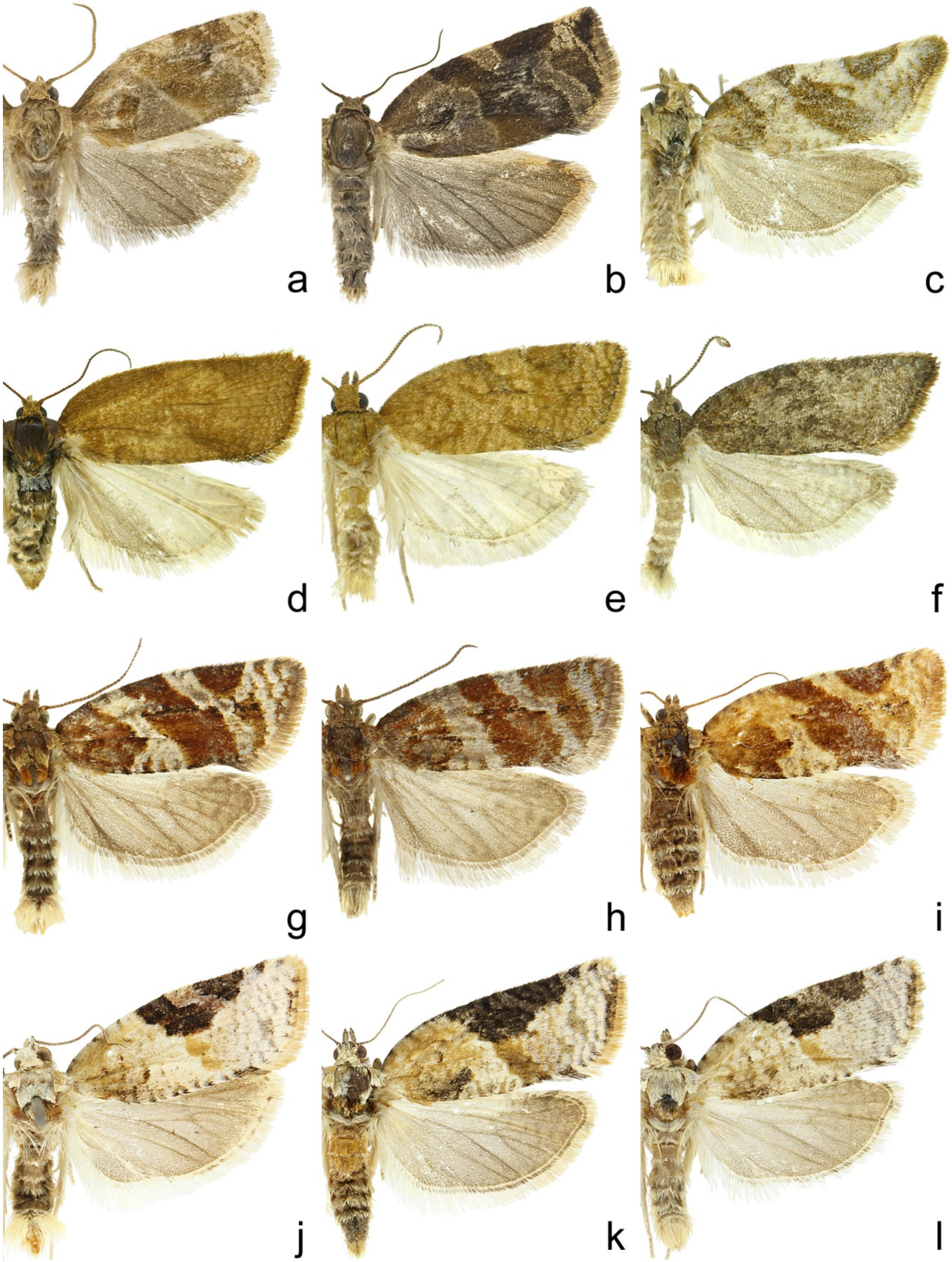


Figure 10. Tortricinae; a–b: *Archips xylosteana*; c–f: *Argyrotaenia franciscana*; g–i: *Argyrotaenia ljungiana*; j–l: *Argyrotaenia mariana*.

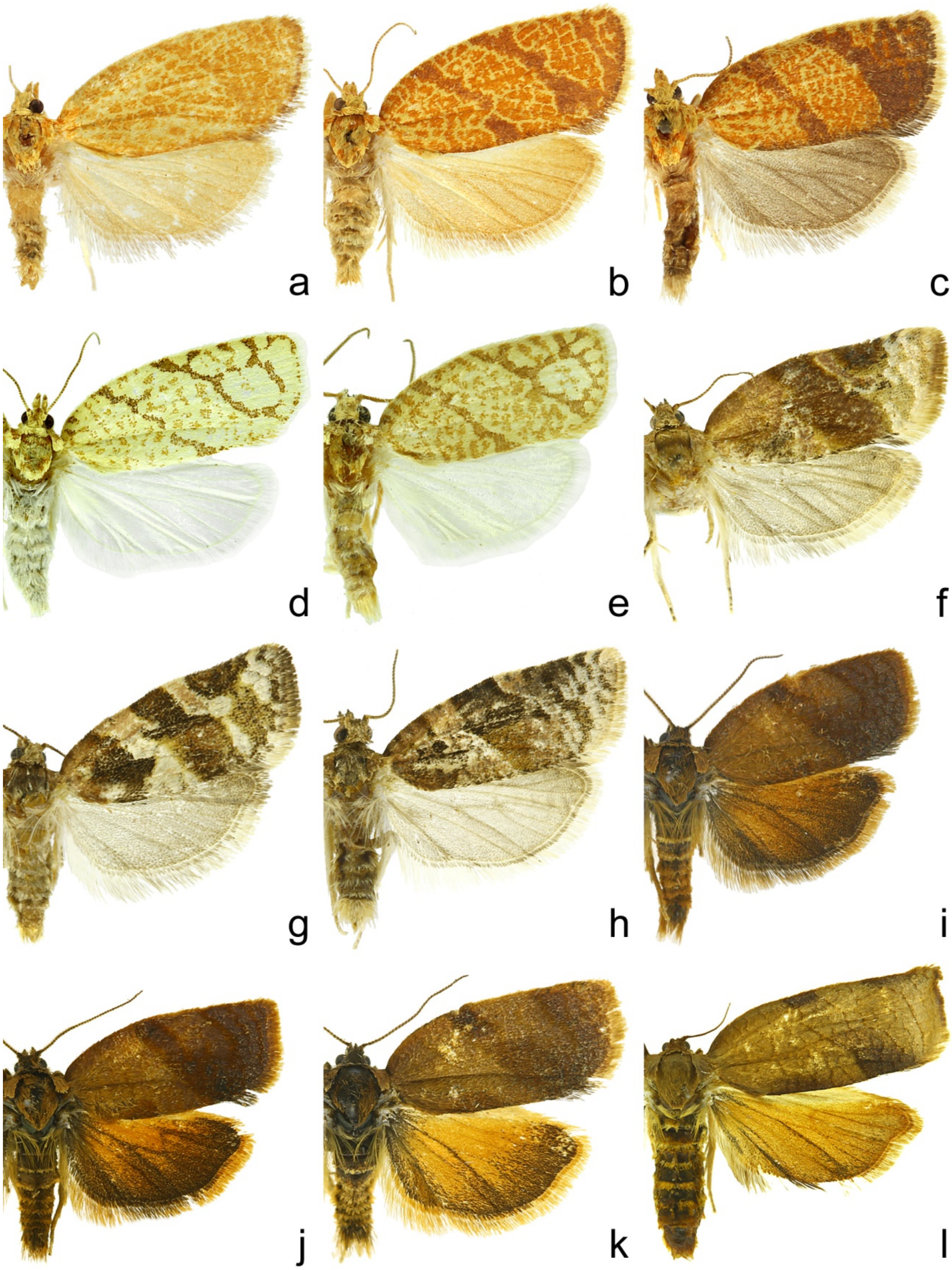


Figure 11. Tortricinae; a–c: *Argyrotaenia quadrifasciana*; d–e: *Argyrotaenia quercifoliana*; f–h: *Argyrotaenia velutinana*; i–l: *Cacoecimorpha pronubana*.

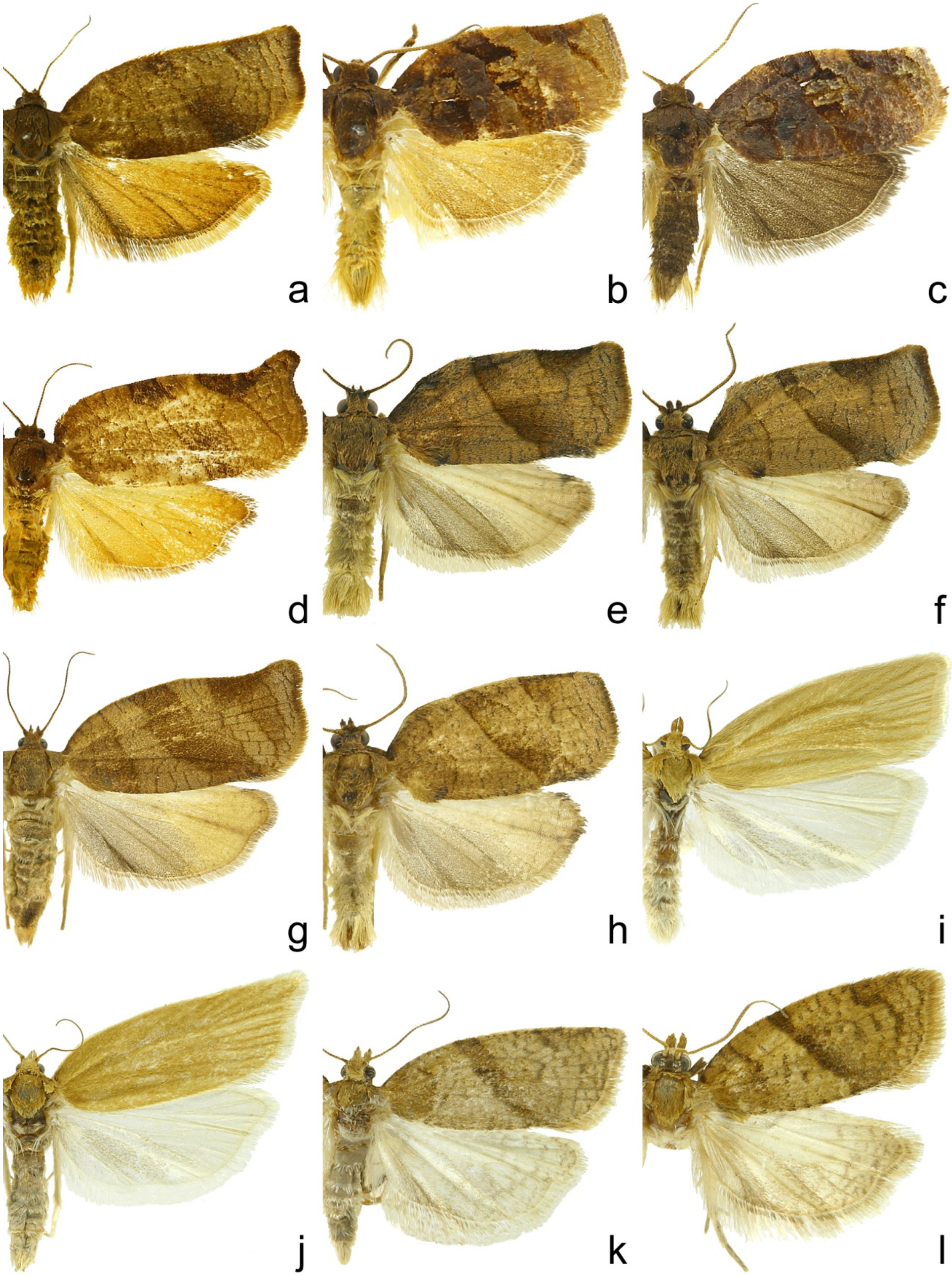


Figure 12. Tortricinae; a: *Cacoecimorpha pronubana*; b–d: *Choristoneura occidentalis*; e–h: *Choristoneura rosaceana*; i–j: *Clepsis clemensiana*; k–l: *Clepsis fucana*.

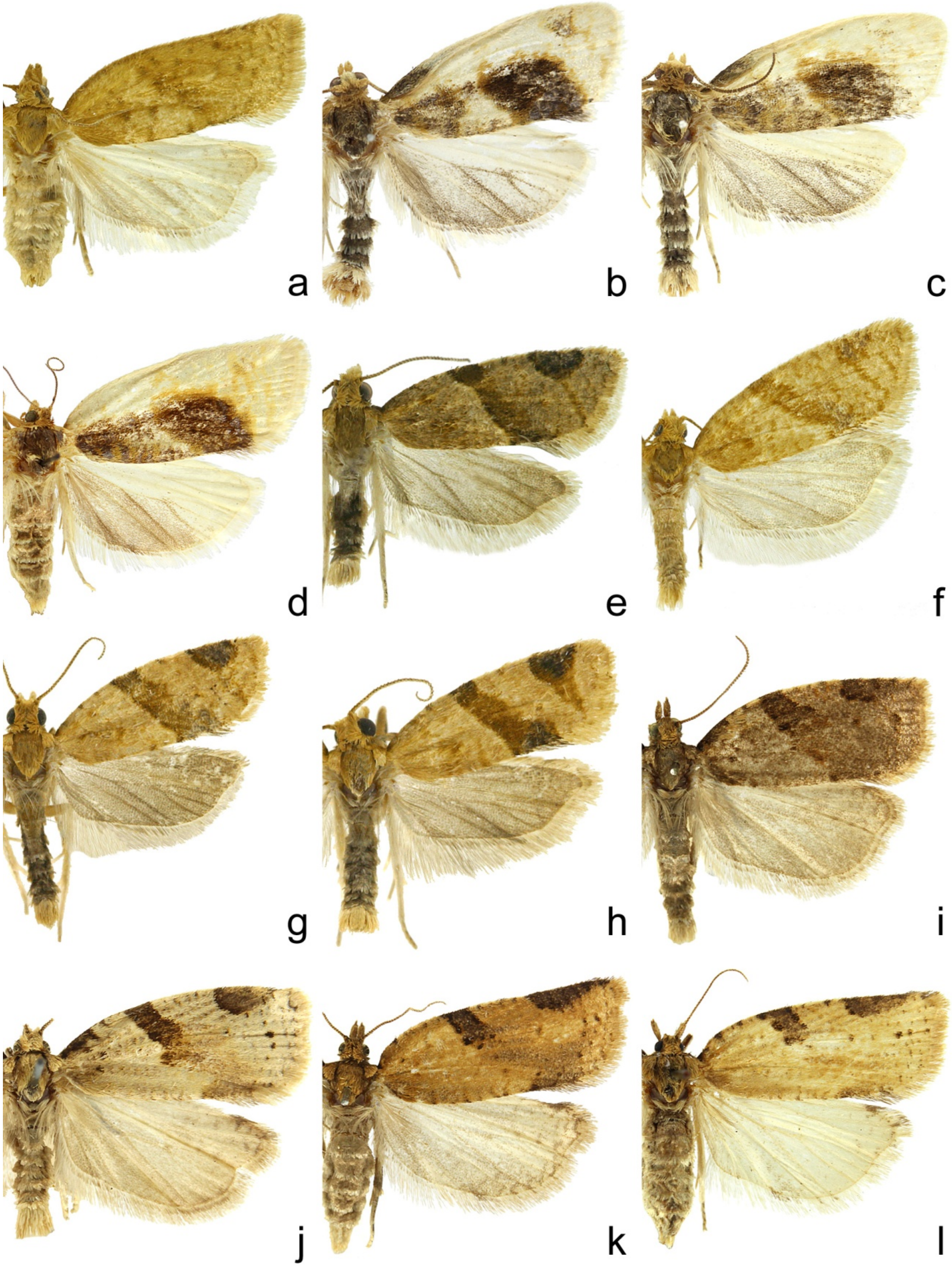


Figure 13. Tortricinae; a: *Clepsis fucana*; b–d: *Clepsis melaleucanus*; e–h: *Clepsis peritana*; i–l: *Clepsis spectrana*.

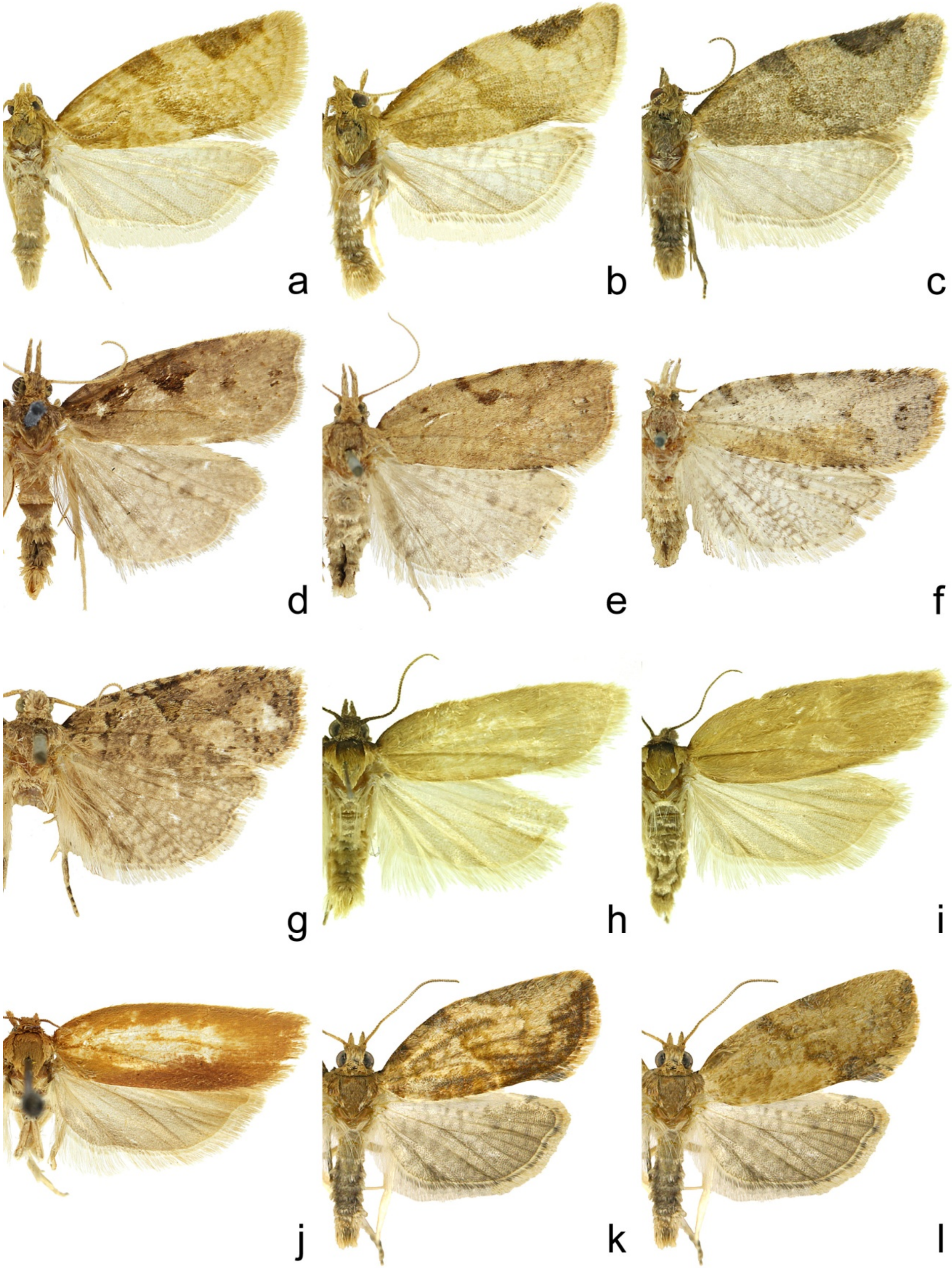


Figure 14. Tortricinae; a–c: *Clepsis virescana*; d–g: *Ctenopseustis obliquana*; h–j: *Epichoristodes acerbella*; k–l: *Epiphyas postvittana*.

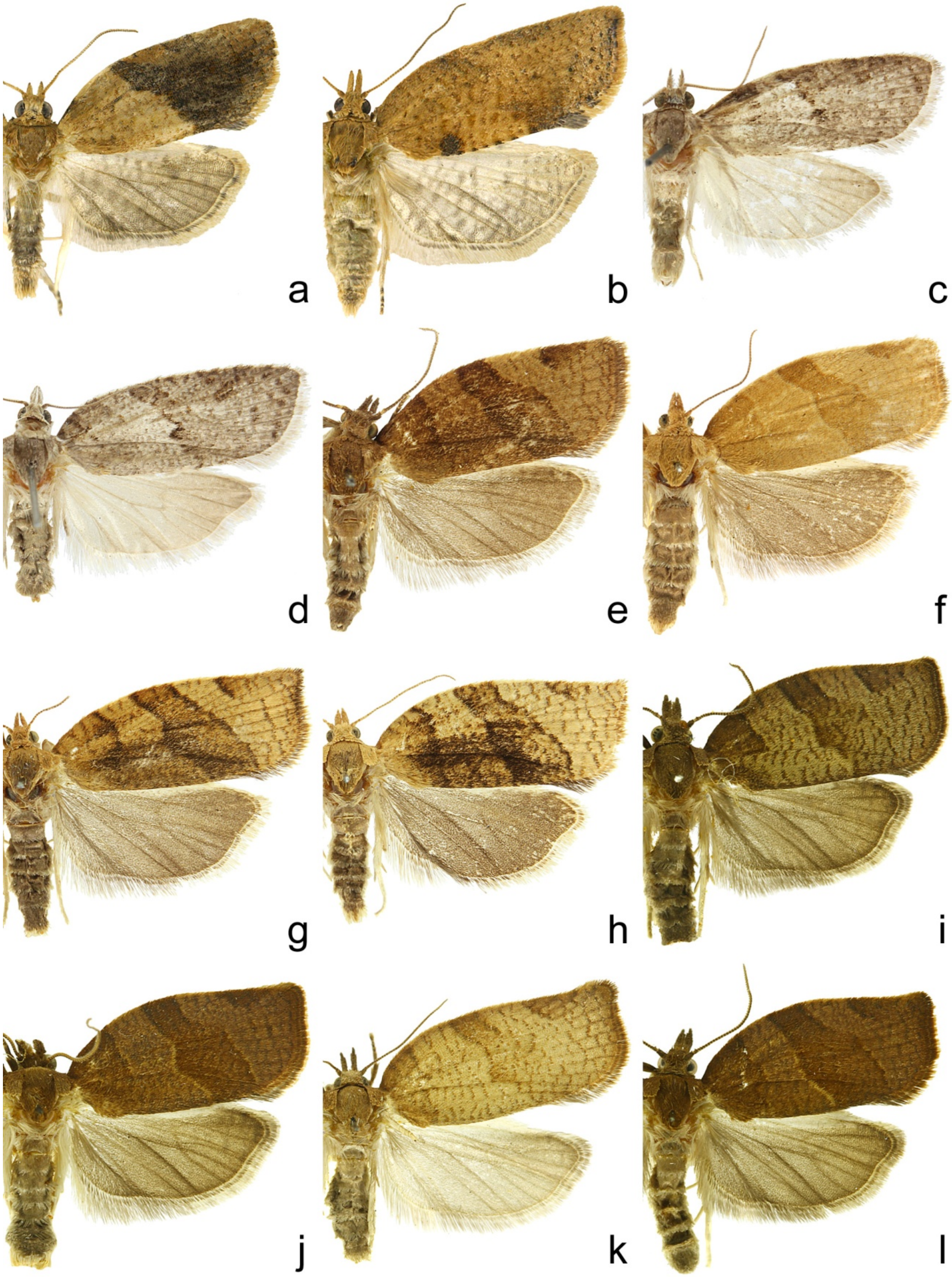


Figure 15. Tortricinae; a–b: *Epiphyas postvittana*; c–d: *Isotenes miserana*; e–h: *Pandemis cerasana*; i–l: *Pandemis heparana*.

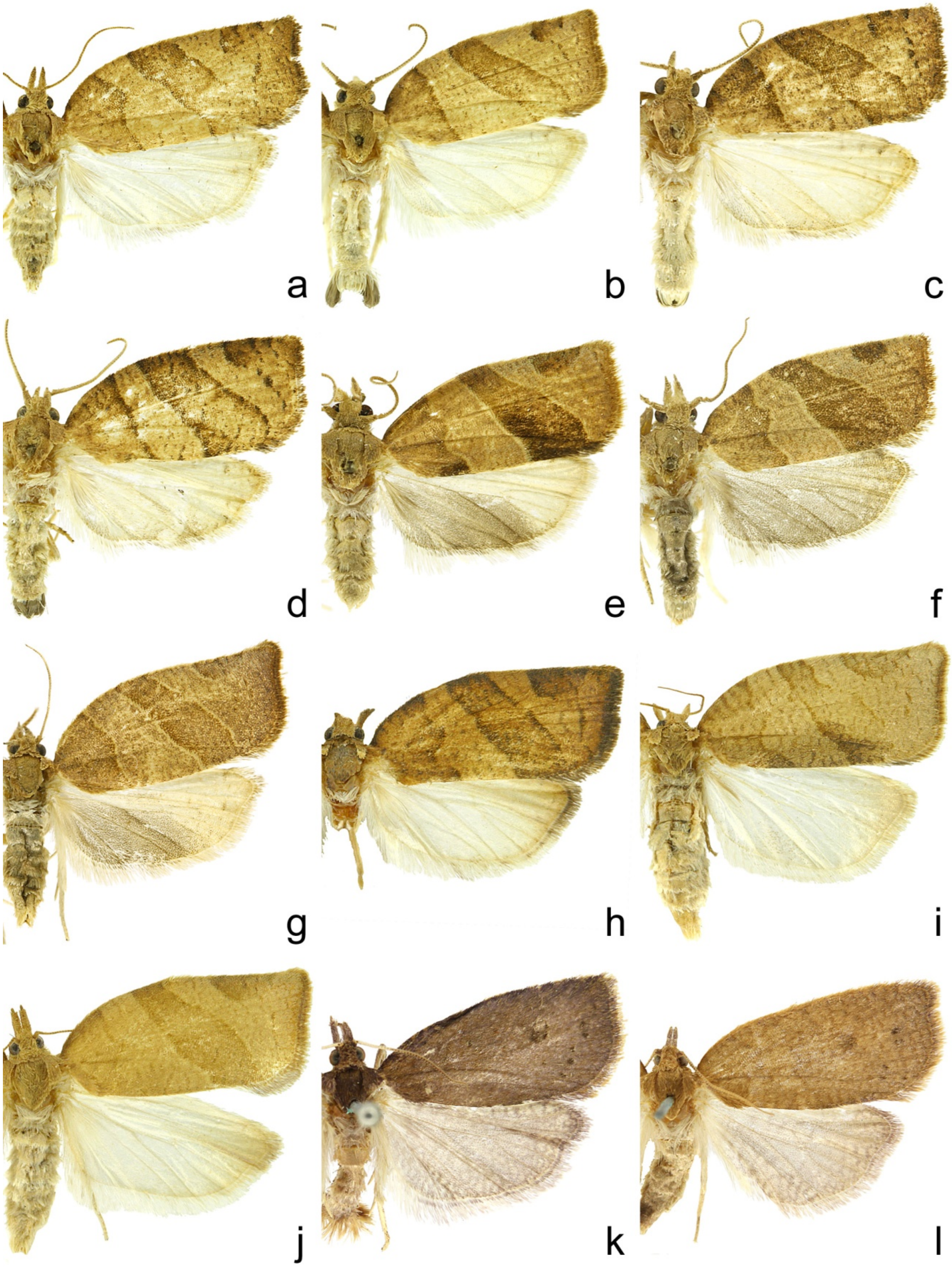


Figure 16. Tortricinae; a–d: *Pandemis lamprosana*; e–g: *Pandemis limitata*; h–j: *Pandemis pyrusana*; k–l: *Planotortrix excessana*.

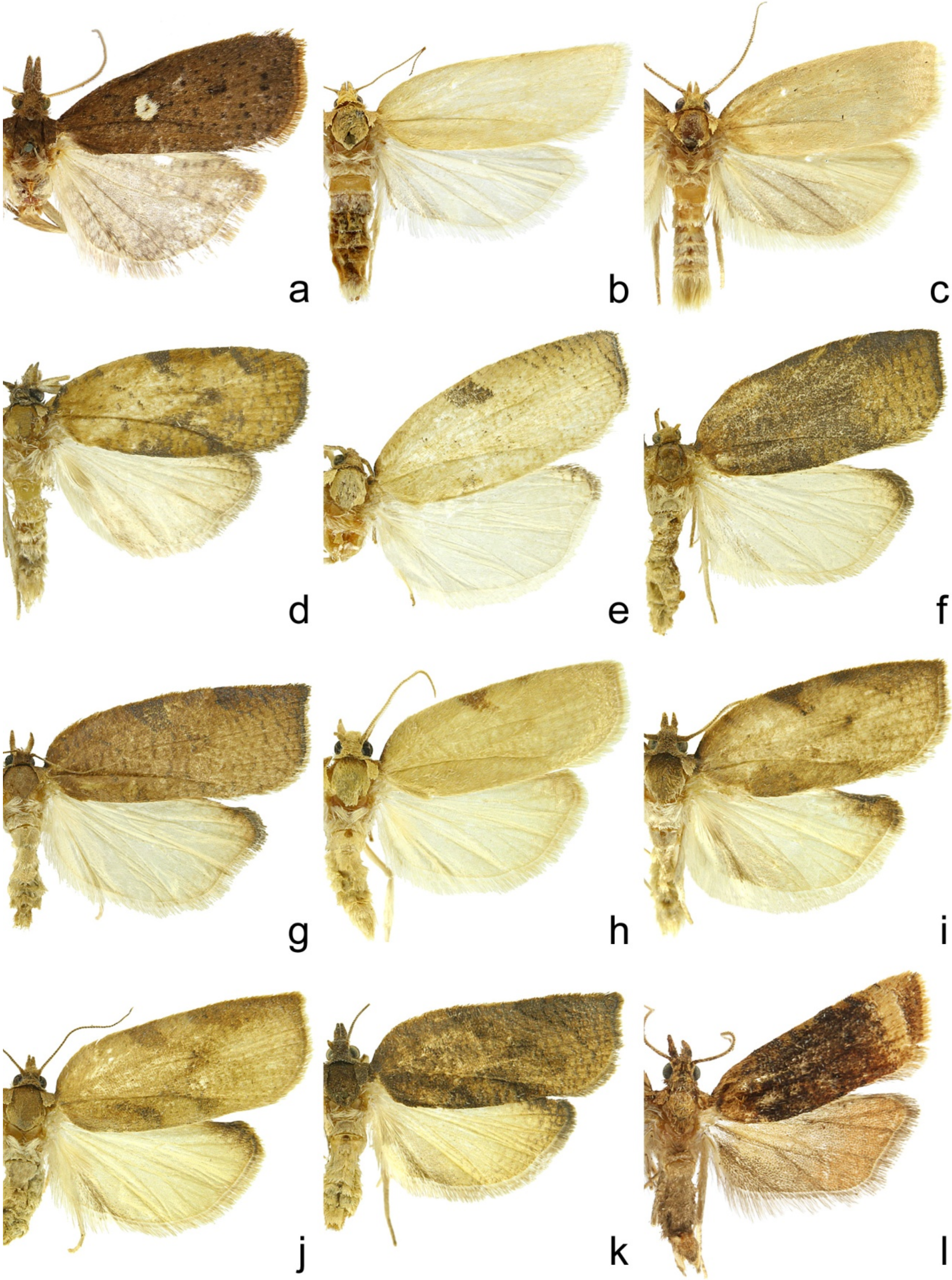


Figure 17. Tortricinae; a: *Planotortrix excessana*; b–c: *Xenotemna pallorana*; d–g: *Amorbia cuneana*; h–k: *Amorbia emigratella*; l: *Platynota flavedana*.

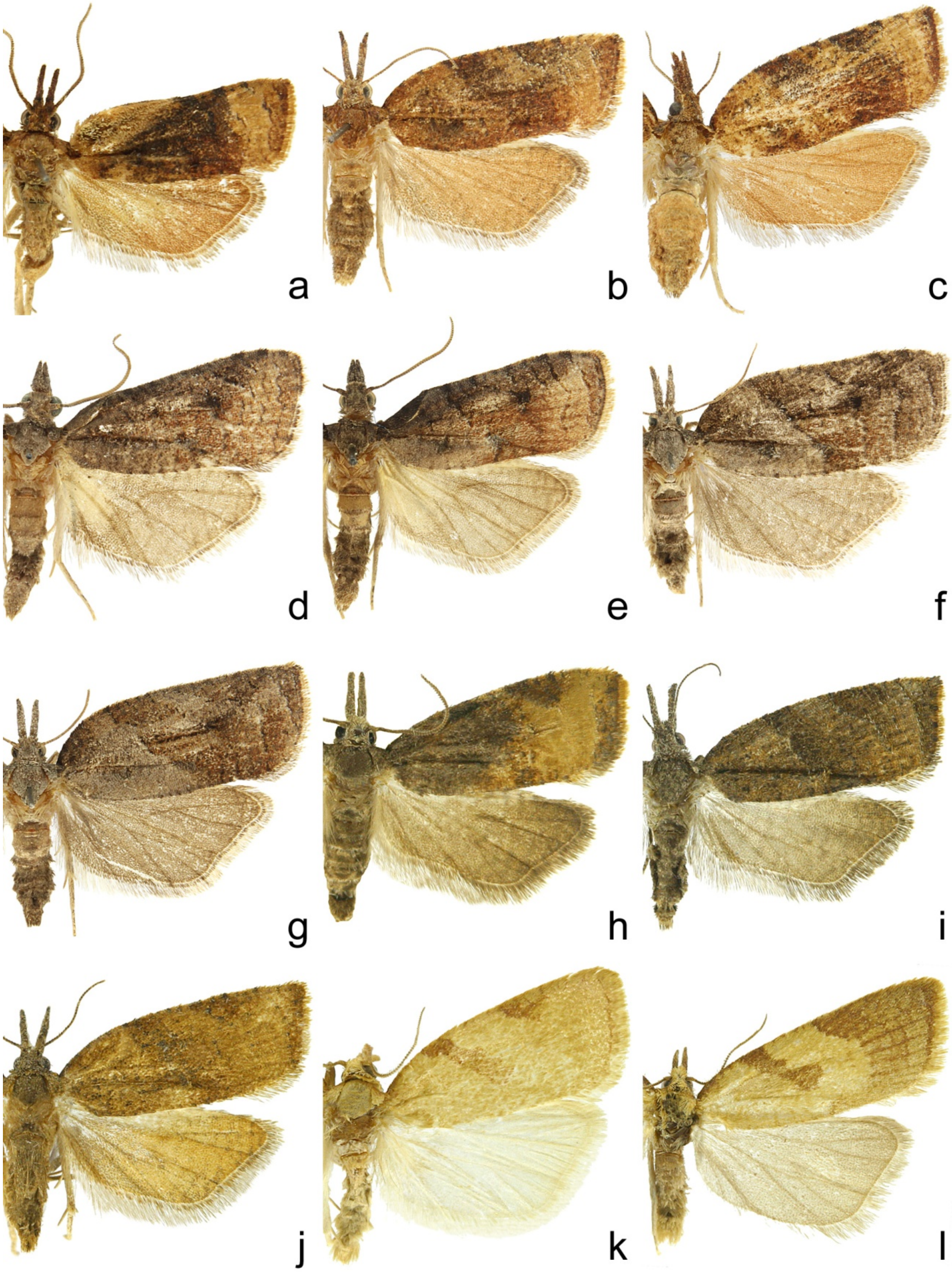


Figure 18. Tortricinae; a–c: *Platynota flavedana*; d–g: *Platynota idaeusalis*; h–j: *Platynota stultana*; k–l: *Sparganothis senecionana*.

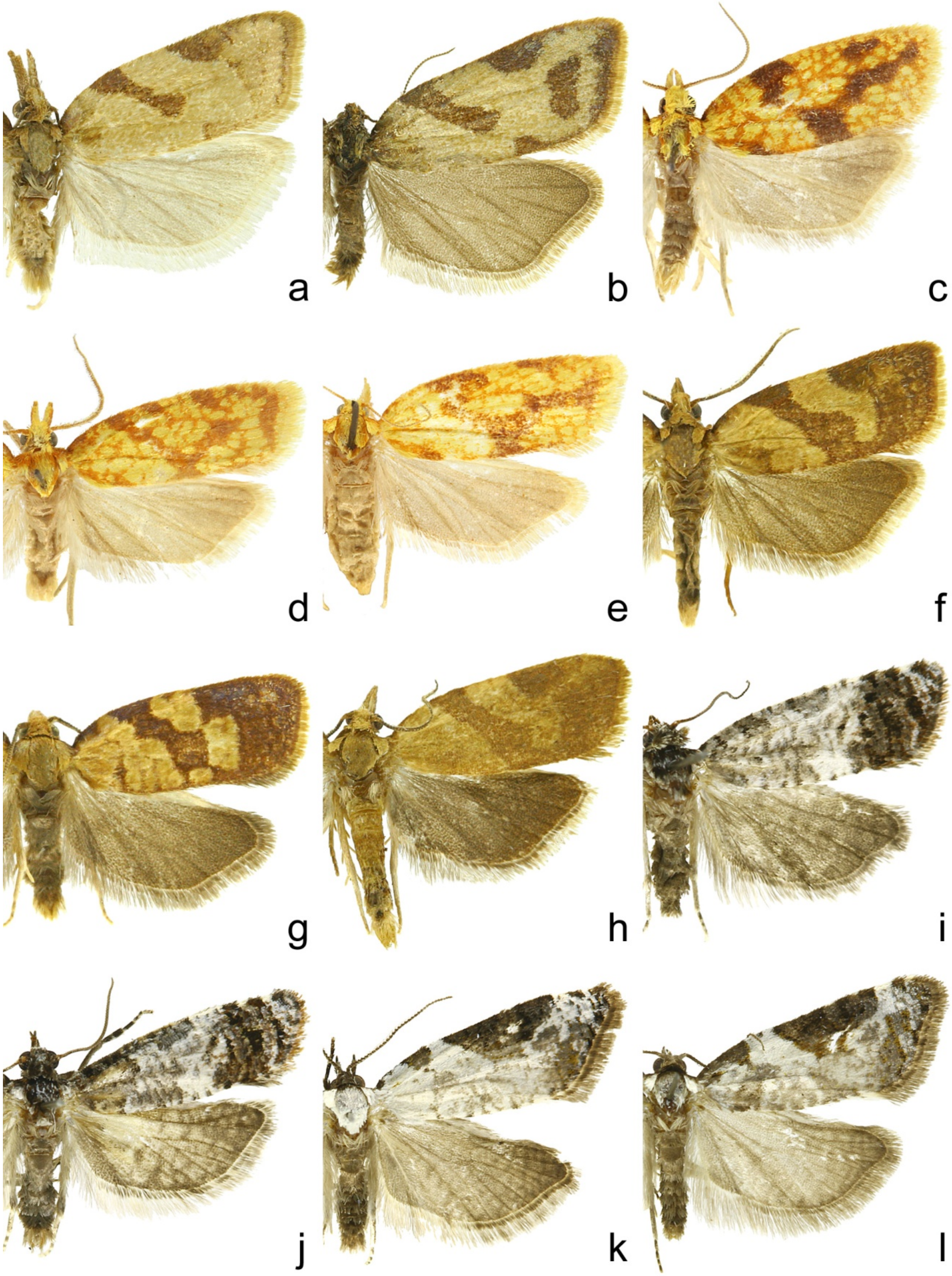


Figure 19. Tortricinae; a–b: *Sparganothis senecionana*; c–e: *Sparganothis sulfureana*; f–h: *Sparganothis tunicana*; i–j: *Accuminulia buscki*; k–l: *Chileulia stalactitis*.

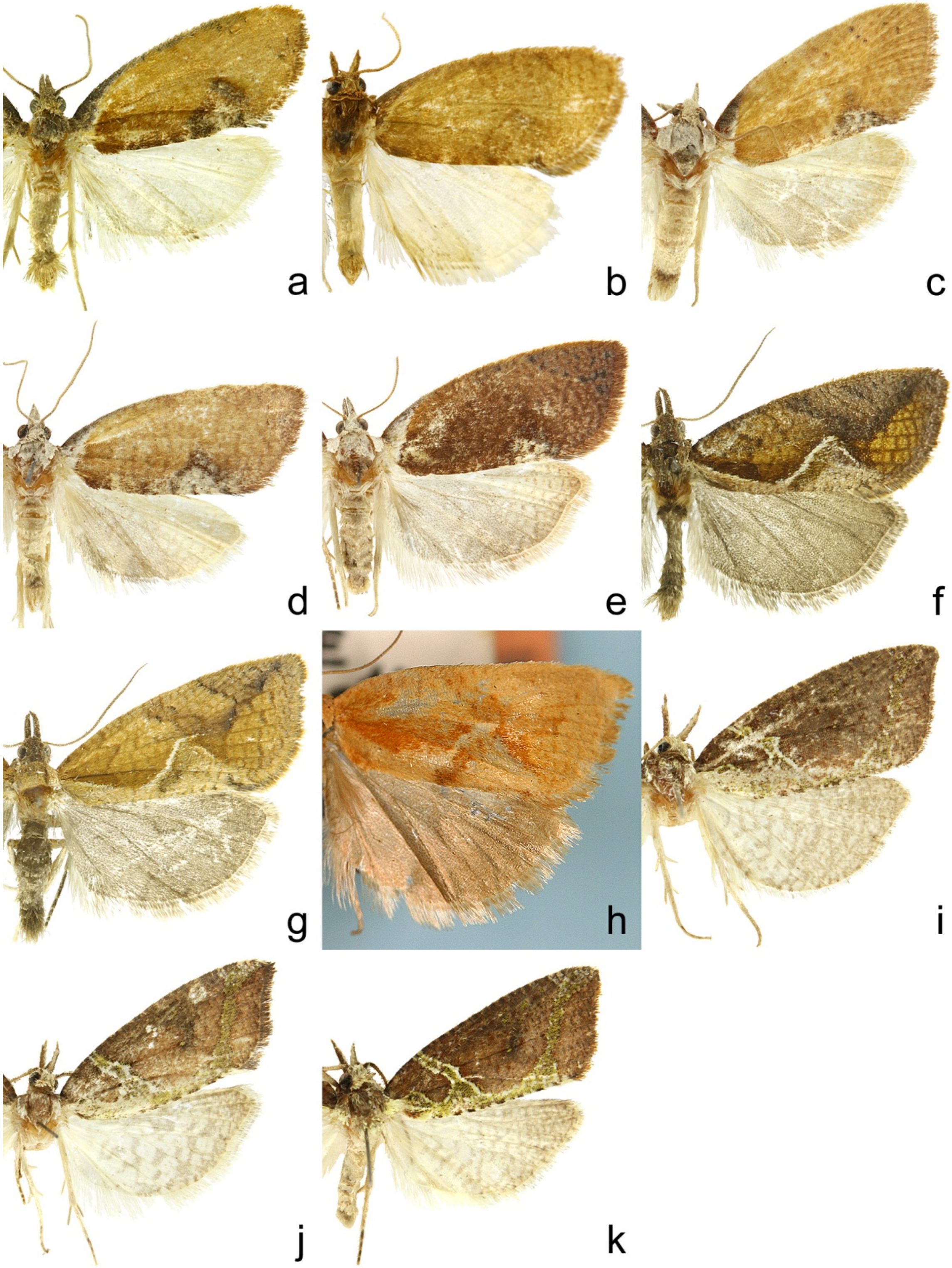


Figure 20. Tortricinae; a–b: *Proeulia auraria*; c–e: *Proeulia chrysopteris*; f–g: *Proeulia triquetra*; h: *Proeulia apospasta*; i–k: *Seticosta rubicola*.

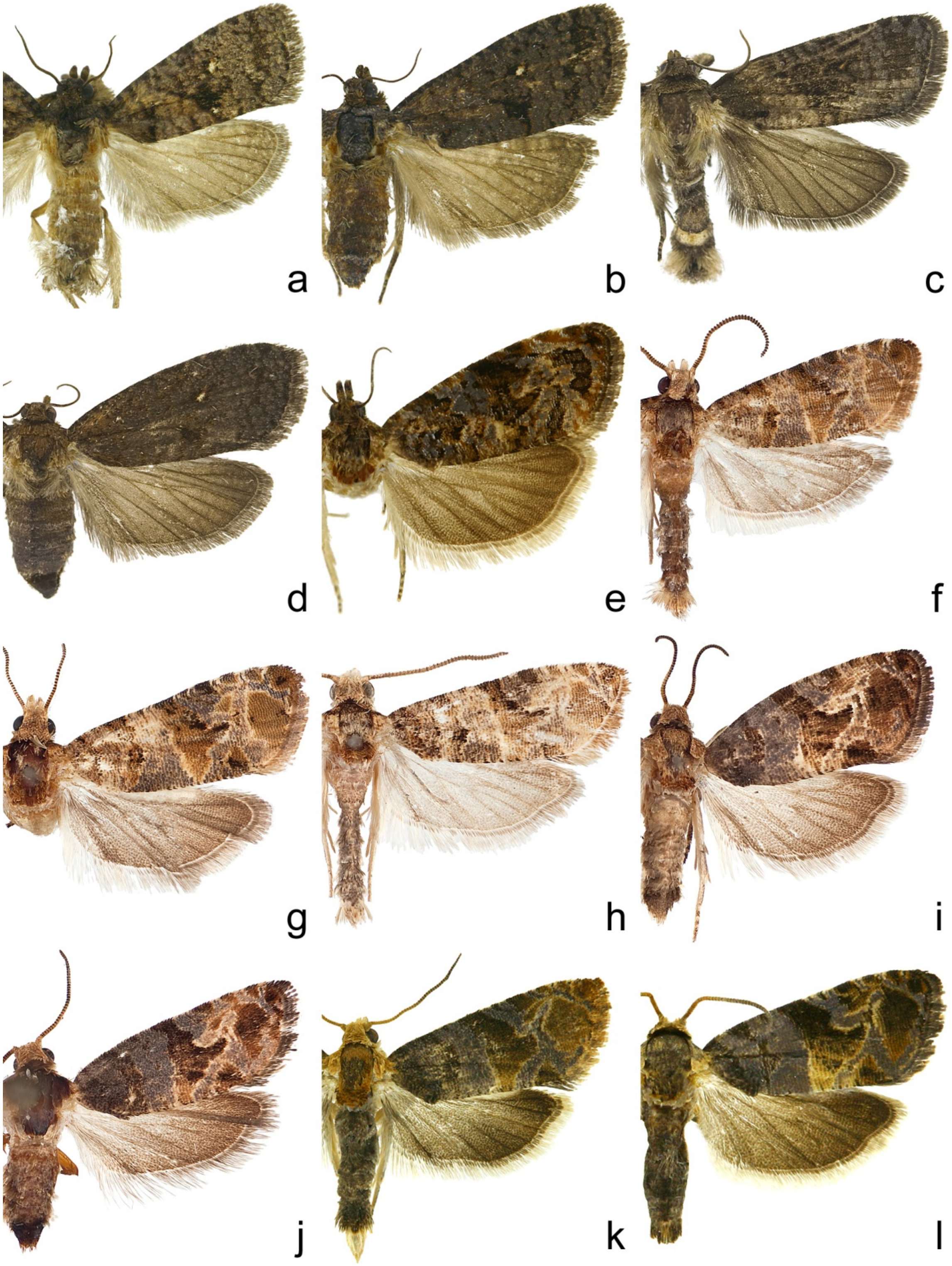


Figure 21. Olethreutinae; a–b: *Cryptaspasma bipenicilla*; c–d: *Cryptaspasma perseana*; e: *Endothenia hebesana*; f–h: *Lobesia botrana*; i–j: *Paralobesia viteana*; k: *Paralobesia lireodendrana*; l: *Paralobesia monotropana*.

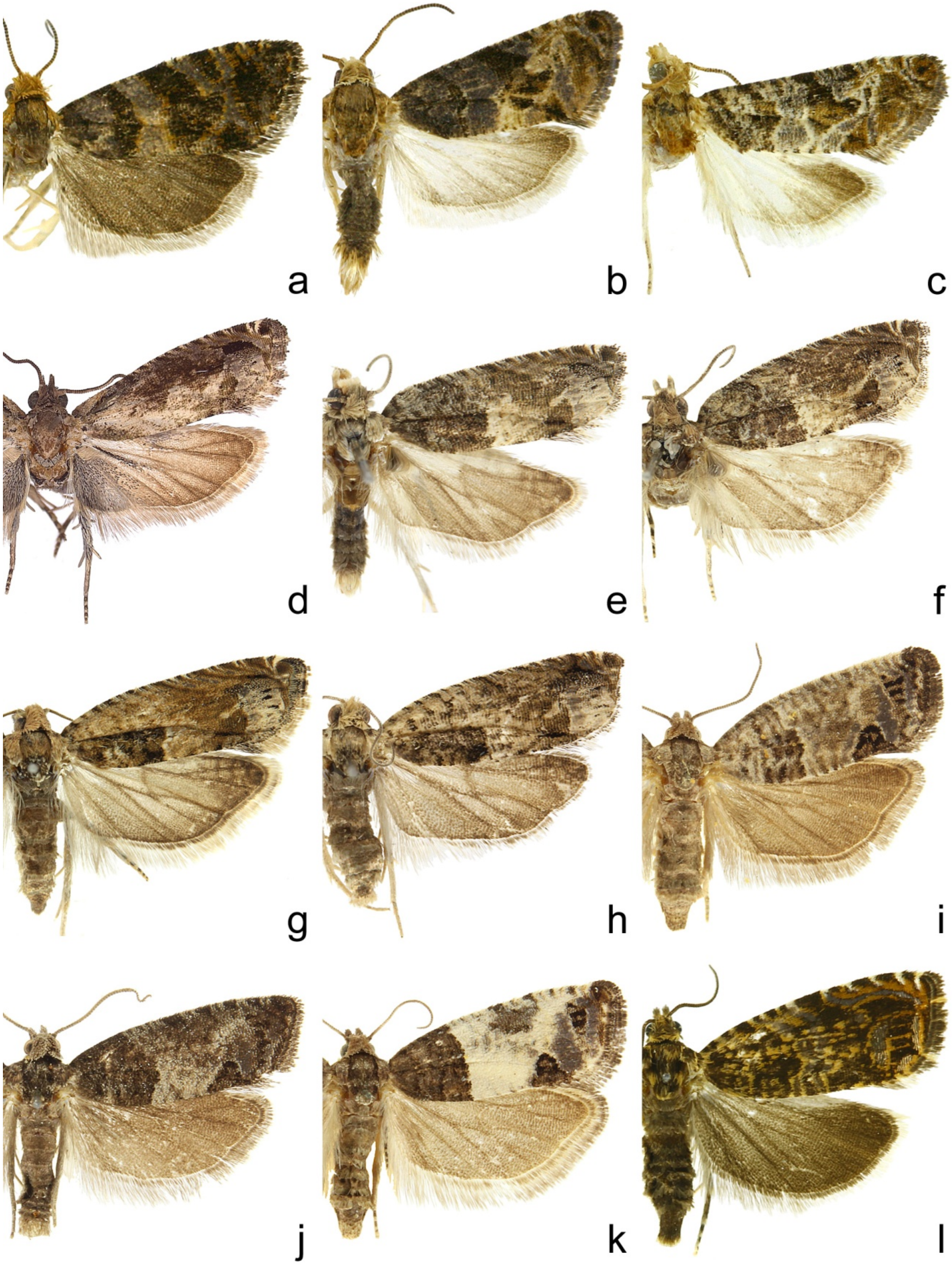


Figure 22. Olethreutinae; a: *Paralobesia palliolana*; b: *Paralobesia rhoifructana*; c: *Paralobesia yaracana*; d: *Crocidosema aporema* (photo credit Joaquín Baixeras Almela); e–h: *Crocidosema plebejana*; i–k: *Spilonota ocellana*; l: *Enarmonia formosana*.

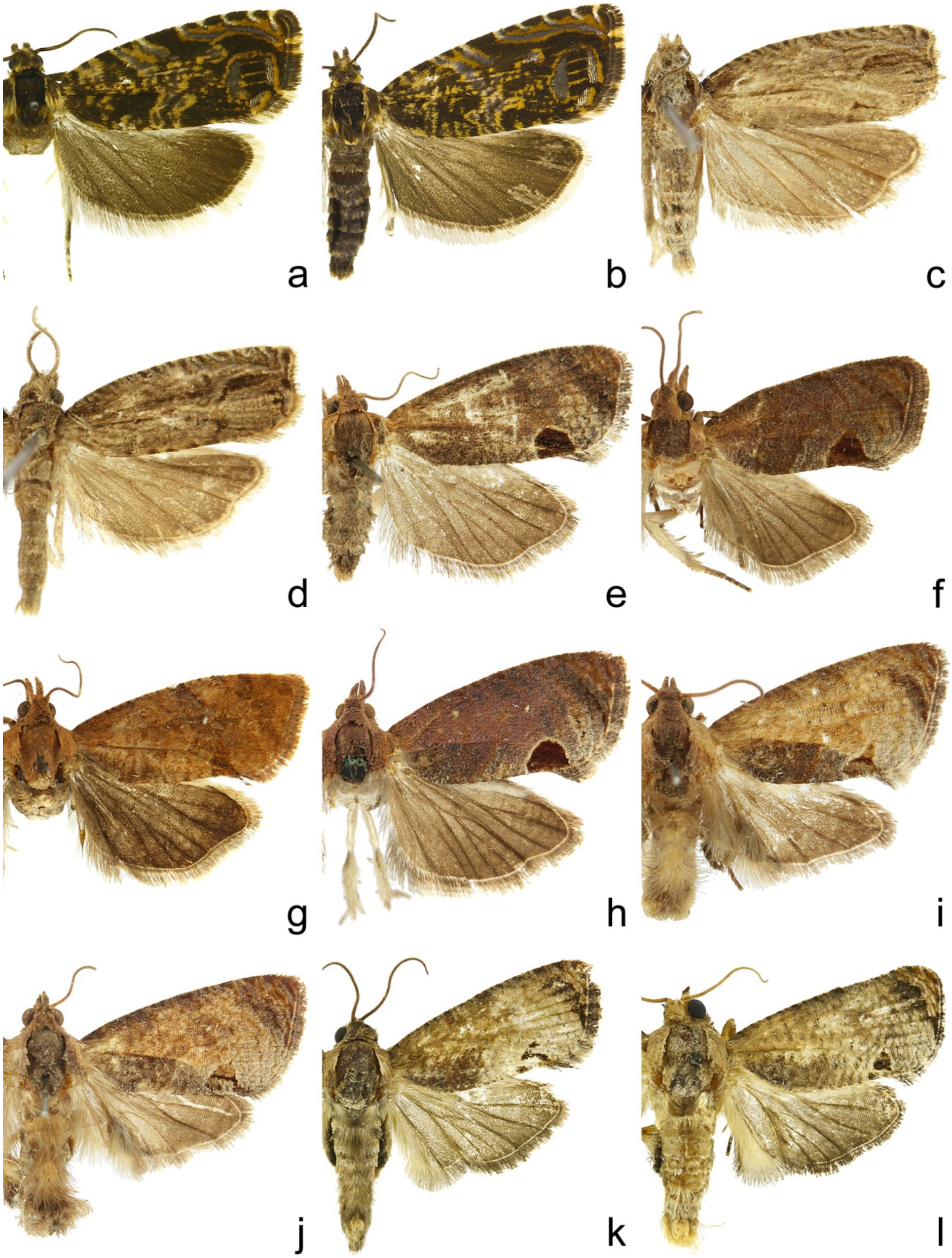


Figure 23. Olethreutinae; a–b: *Enarmonia formosana*; c–d: *Tetramoera schistaceana*; e–g: *Cryptophlebia illepida*; h–j: *Cryptophlebia ombrodelta*; k–l: *Cryptophlebia peltastica*.

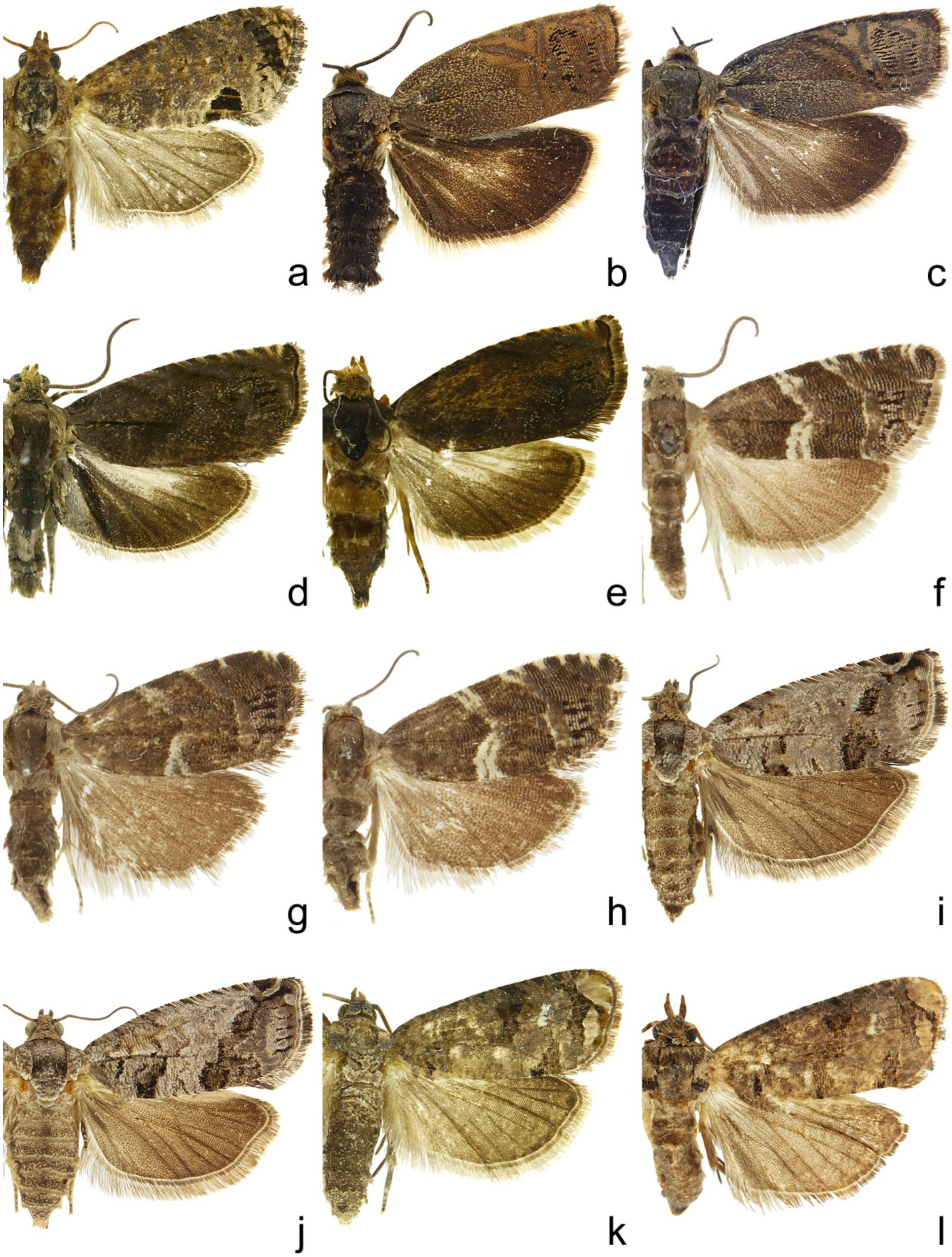


Figure 24. Olethreutinae; a: *Cryptophlebia peltastica*; b–c: *Cydia araucariae*; d–e: *Cydia caryana*; f–h: *Cydia coniferana*; i–j: *Cydia deshaisiana*; k–l: *Cydia fabivora*.

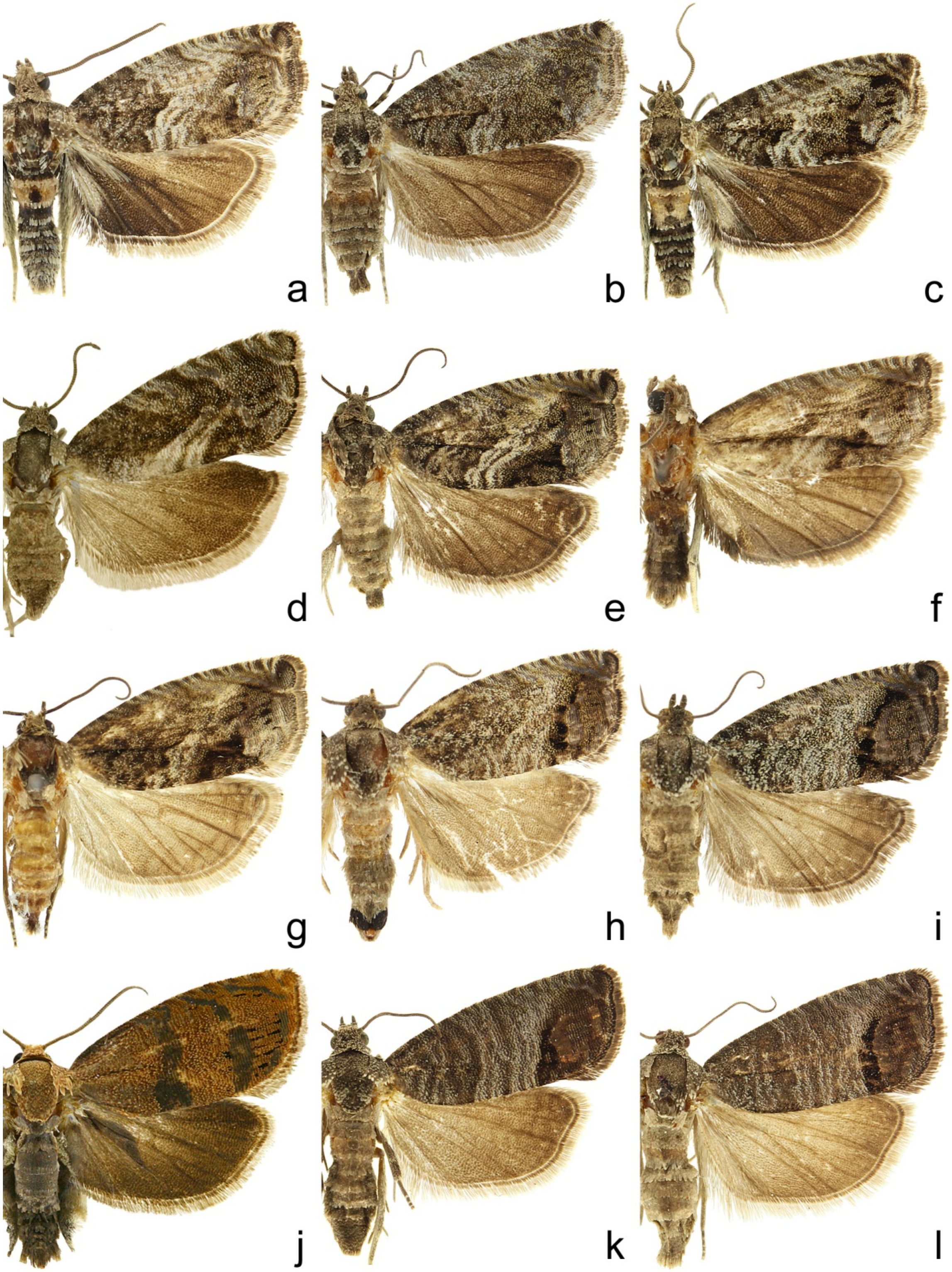


Figure 25. Olethreutinae; a–c: *Cydia fagiglandana*; d: *Cydia garacana*; e–g: *Cydia glandicolana*; h–i: *Cydia kurokoi*; j: *Cydia latiferreana*; k–l: *Cydia pomonella*.

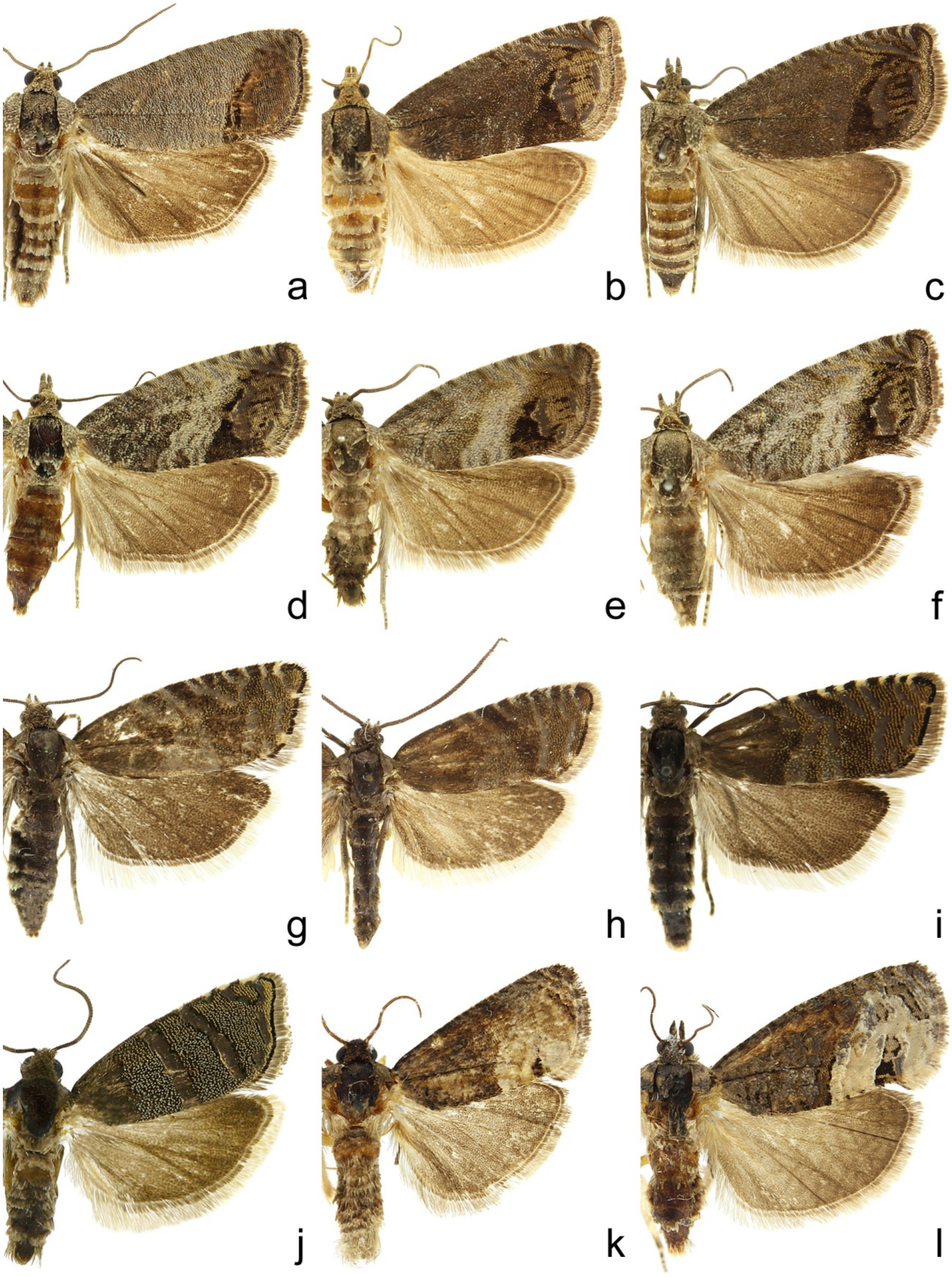


Figure 26. Olethreutinae; a: *Cydia pomonella*; b–f: *Cydia splendana*; g–i: *Cydia strobilella*; j: *Cydia toreuta*; k–l: *Ecdytolopha insiticiana*.

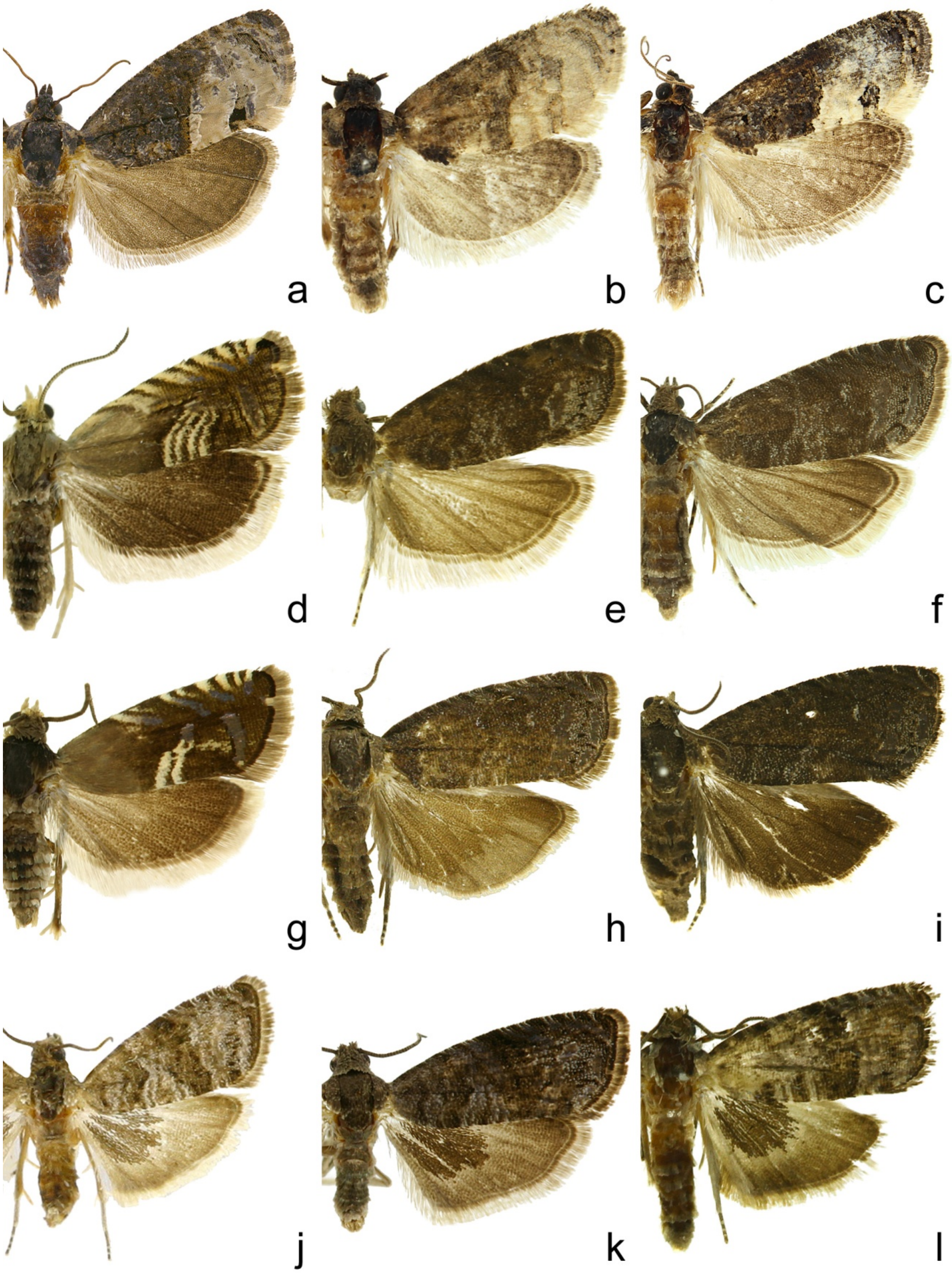


Figure 27. Olethreutinae; a: *Ecdytolopha insiticiana*; b: *Ecdytolopha mana*; c: *Ecdytolopha occidentana*; d: *Grapholita delineana*; e–f: *Grapholita funebrana*; g: *Grapholita interstinctana*; h–i: *Grapholita molesta*; j–l: *Grapholita packardi*.

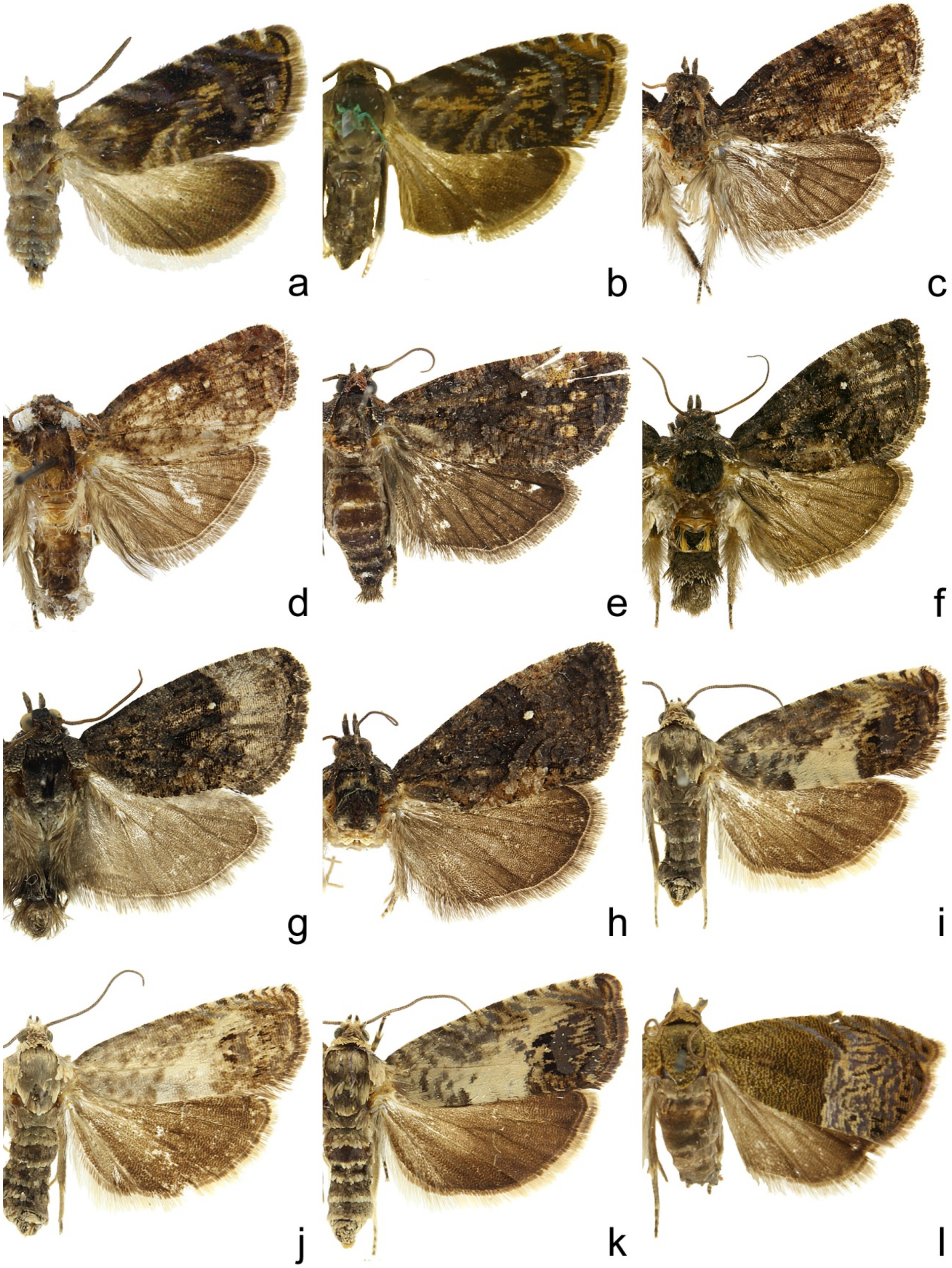


Figure 28. Olethreutinae; a–b: *Grapholita prunivora*; c–e: *Gymnandrosoma aurantianum*; f–h: *Gymnandrosoma punctidiscanum*; i–k: *Pammene fasciana*; l: *Talponia batesi*.

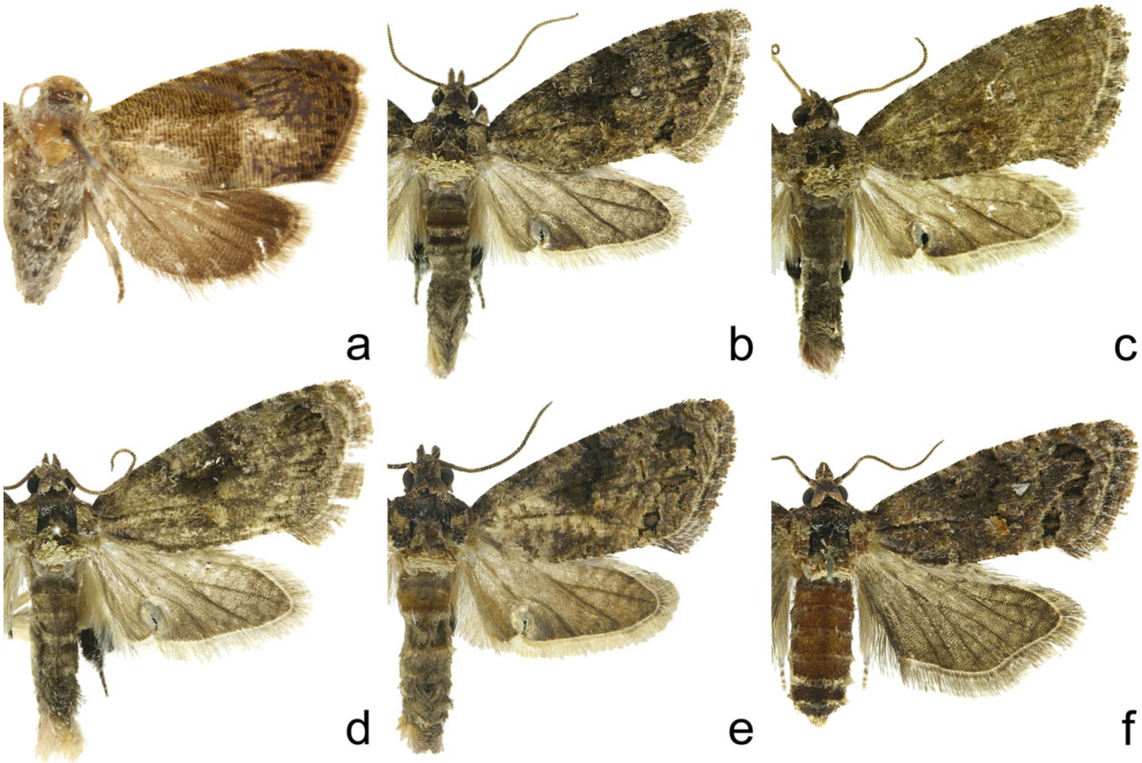


Figure 29. Olethreutinae; a: *Talponia batesi*; b–f: *Thaumatotibia leucotreta*.

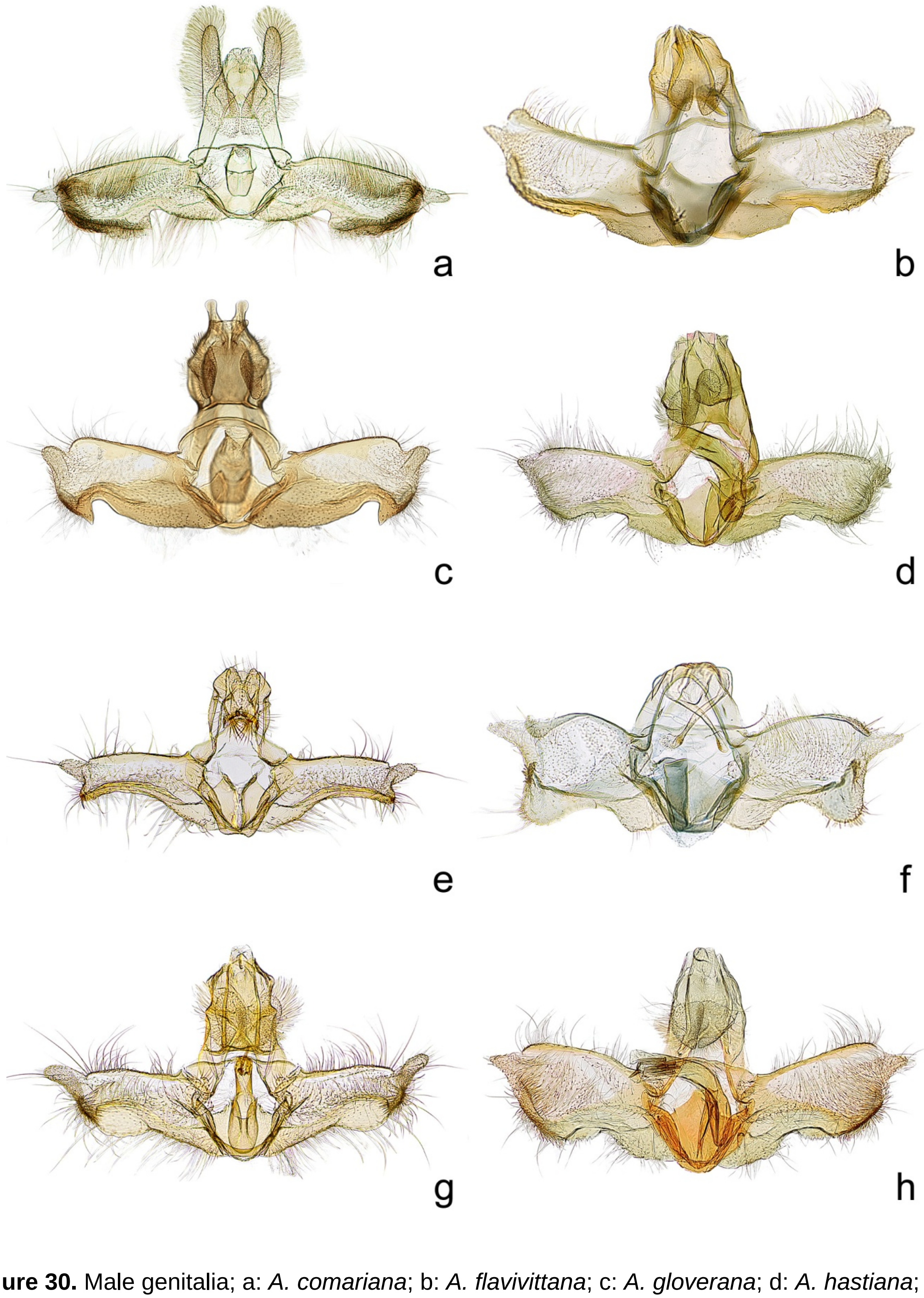


Figure 30. Male genitalia; a: *A. comariana*; b: *A. flavivittana*; c: *A. gloverana*; d: *A. hastiana*; e: *A. holmiana*; f: *A. minuta*; g: *A. nivisellana*; h: *A. robinsoniana*.

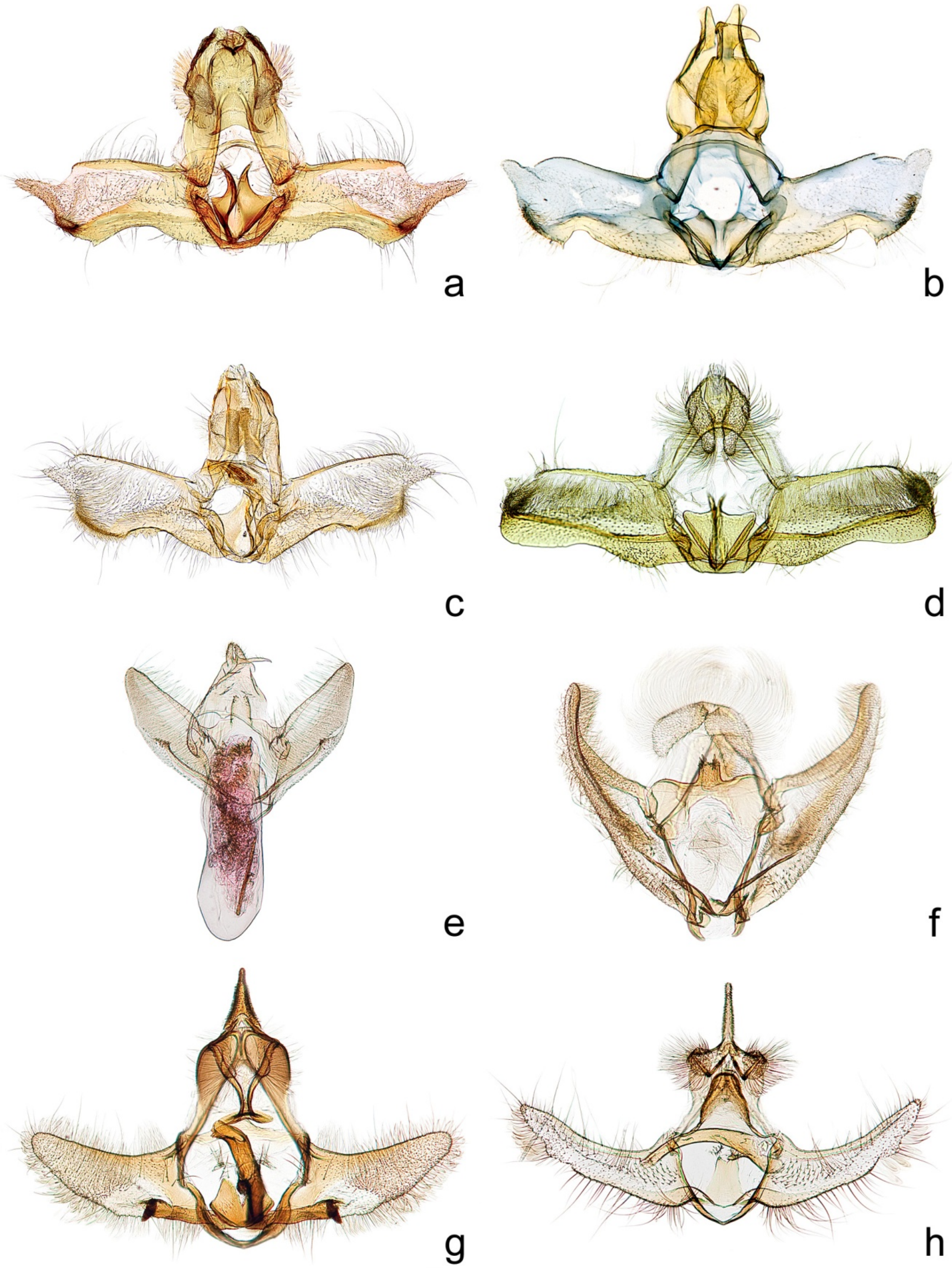


Figure 31. Male genitalia; a: *A. senescens*; b: *A. variana*; c: *A. variegana*; d: *T. viridana*; e: *E. ambiguella*; f: *H. umbrabasana*; g: *C. longana*; h: *D. basiplagana*.

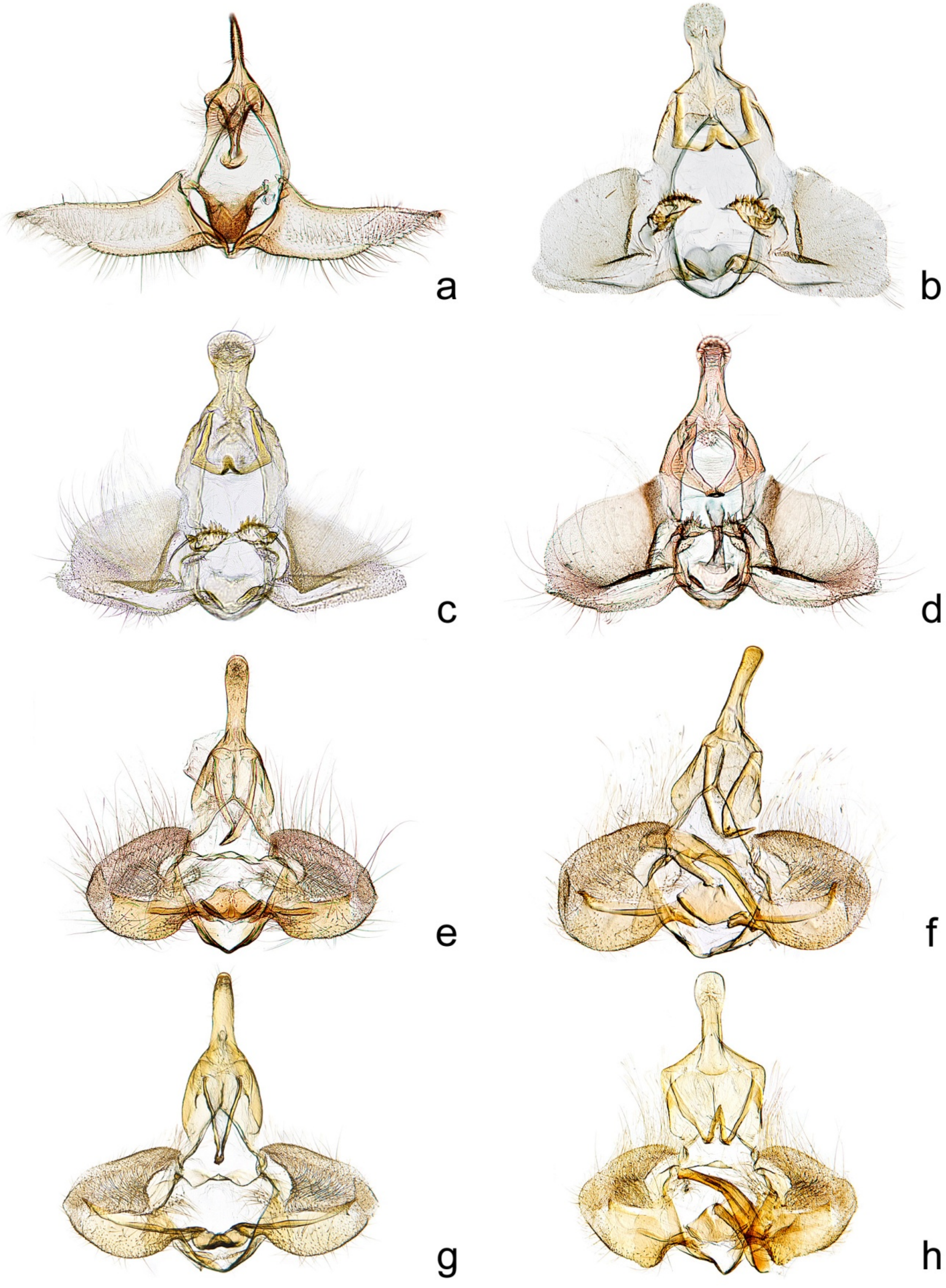


Figure 32. Male genitalia; a: *D. fragariana*; b: *A. furcatana*; c: *A. negundana*; d: *A. orana*; e: *A. argyrospila*; f: *A. cerasivorana*; g: *A. crataegana*; h: *A. fuscocupreanus*.

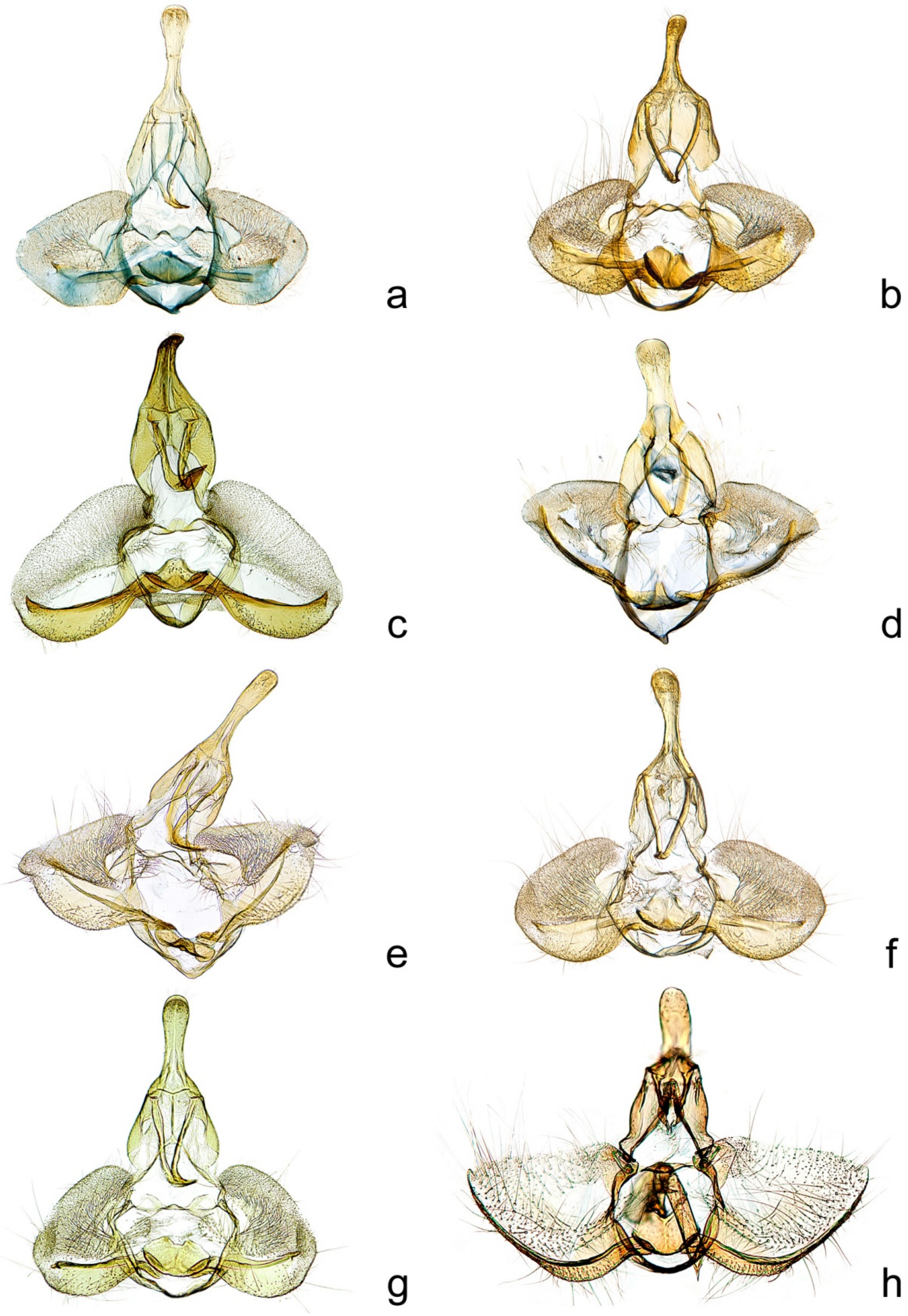


Figure 33. Male genitalia; a: *A. grisea*; b: *A. mortuana*; c: *A. podana*; d: *A. purpurana*; e: *A. rosana*; f: *A. semiferanus*; g: *A. xylosteana*; h: *A. franciscana*.

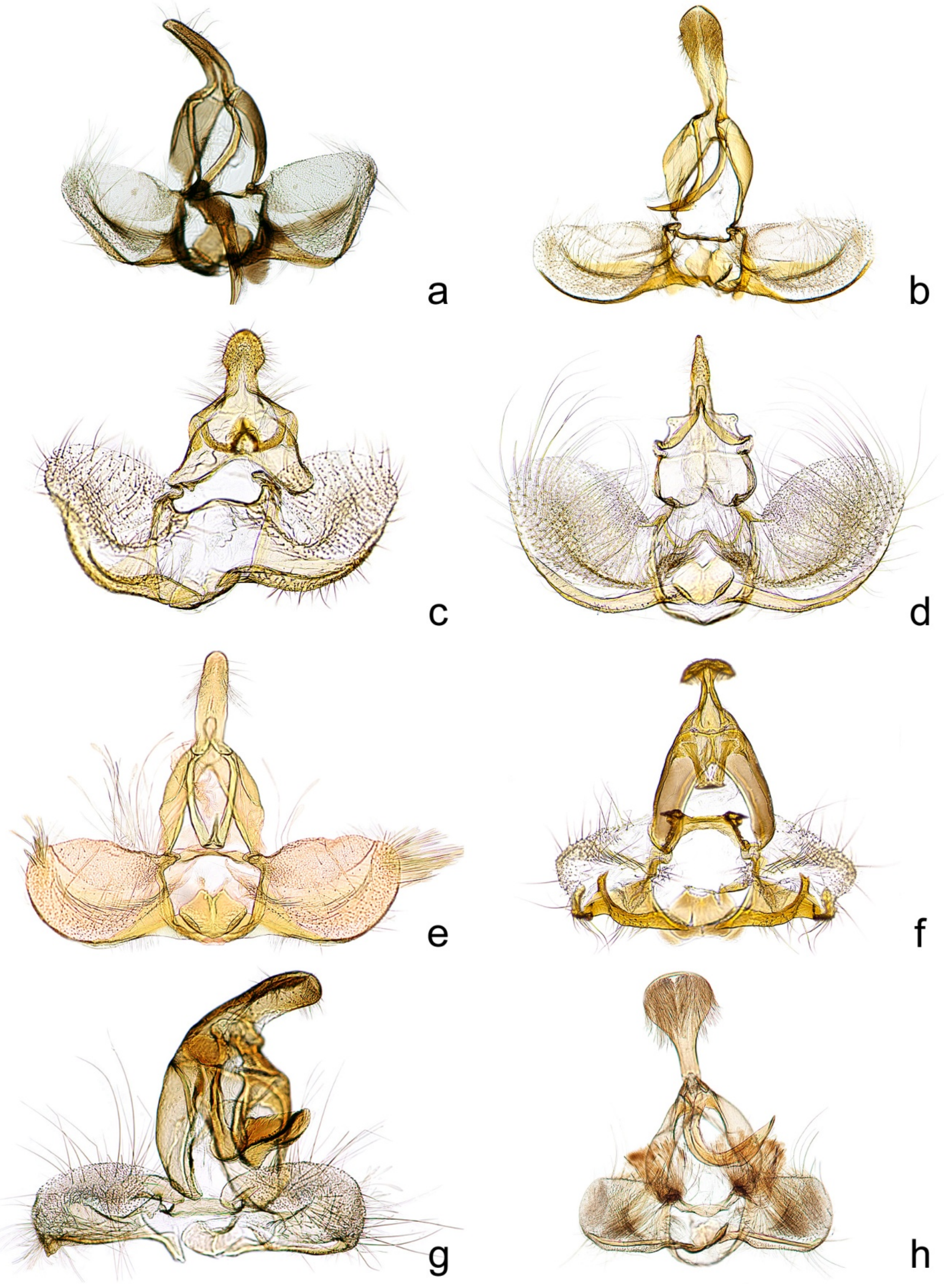


Figure 34. Male genitalia; a: *A. ljungiana*; b: *A. mariana*; c: *A. quadrifasciana*; d: *A. quercifoliana*; e: *A. velutinana*; f: *C. pronubana*; g: *C. occidentalis*; h: *C. rosaceana*.

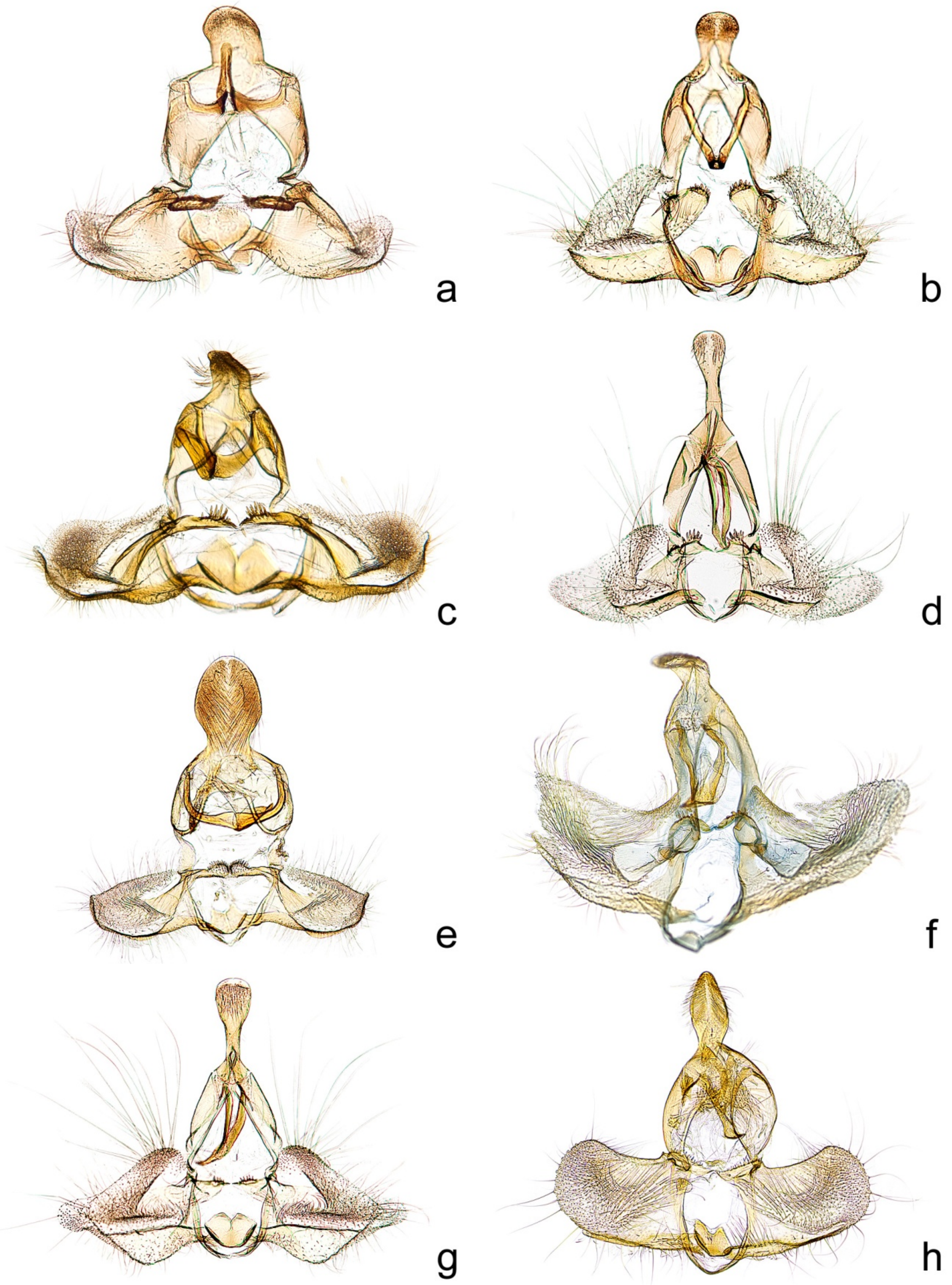


Figure 35. Male genitalia; a: *C. clemensiana*; b: *C. fucana*; c: *C. melaleucanus*; d: *C. peritana*; e: *C. persicana*; f: *C. spectrana*; g: *C. virscana*; h: *C. obliquana*.

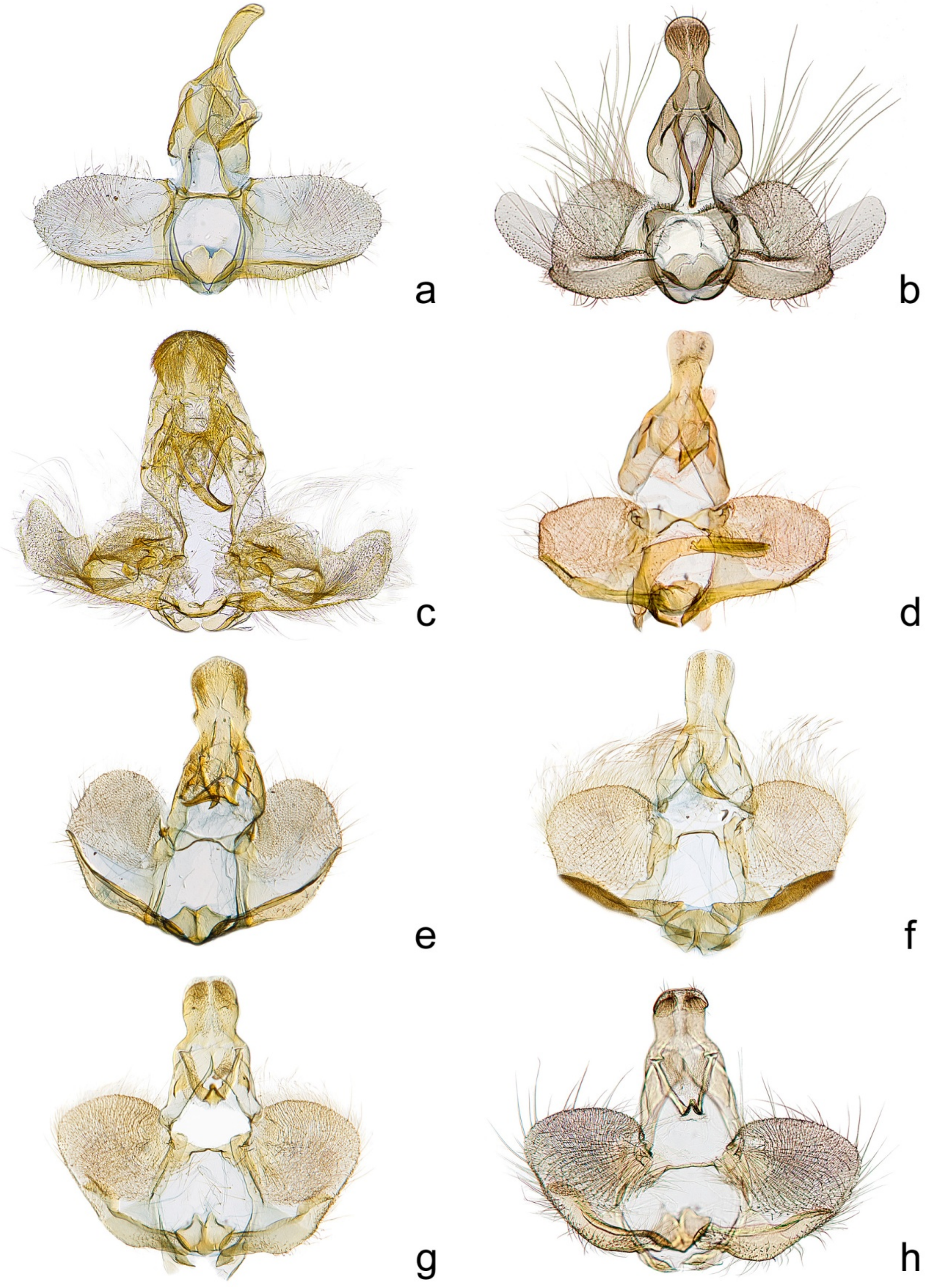


Figure 36. Male genitalia; a: *E. acerbella*; b: *E. postvittana*; c: *I. miserana*; d: *P. cerasana*; e: *P. heparana*; f: *P. lamprosana*; g: *P. limitata*; h: *P. pyrusana*.

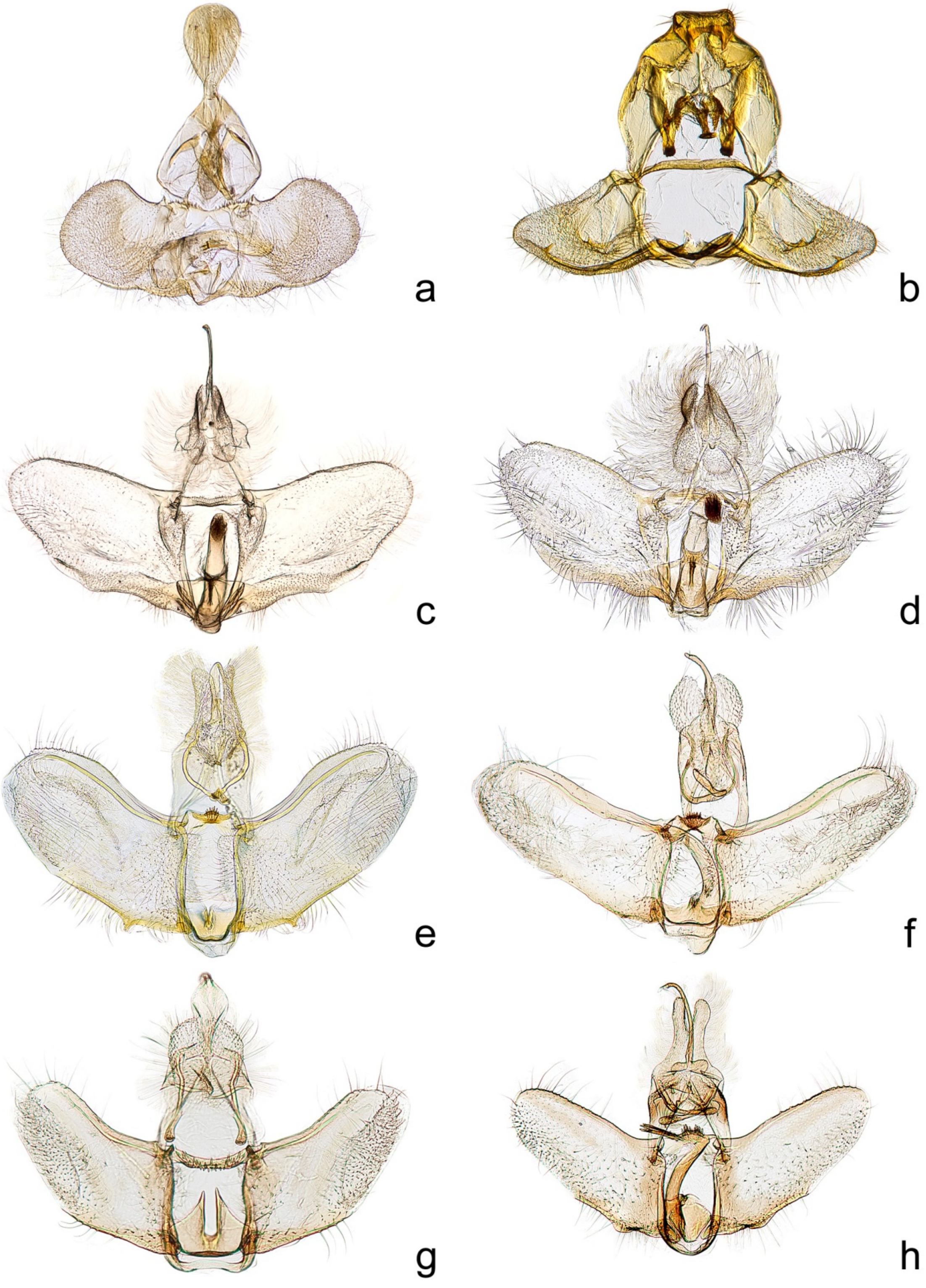


Figure 37. Male genitalia; a: *P. excessana*; b: *X. pallorana*; c: *A. cuneana*; d: *A. emigratella*; e: *P. flavedana*; f: *P. idaeusalis*; g: *P. stultana*; h: *S. senecionana*.

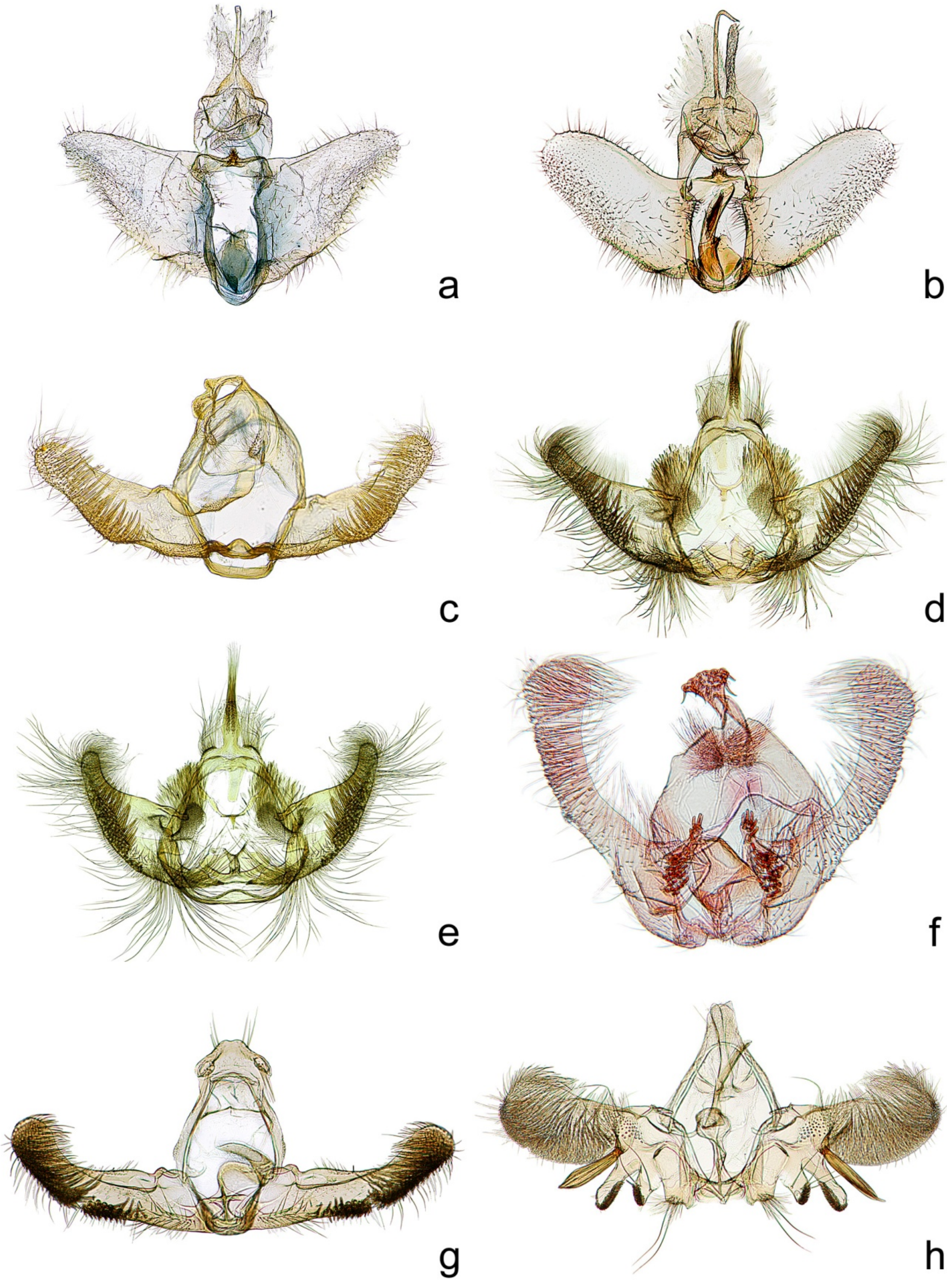


Figure 38. Male genitalia; a: *S. sulfureana*; b: *S. tunicana*; c: *S. rubicola*; d: *C. bipenicilla*; e: *C. perseana*; f: *E. hebesana*; g: *L. botrana*; h: *P. viteana*.

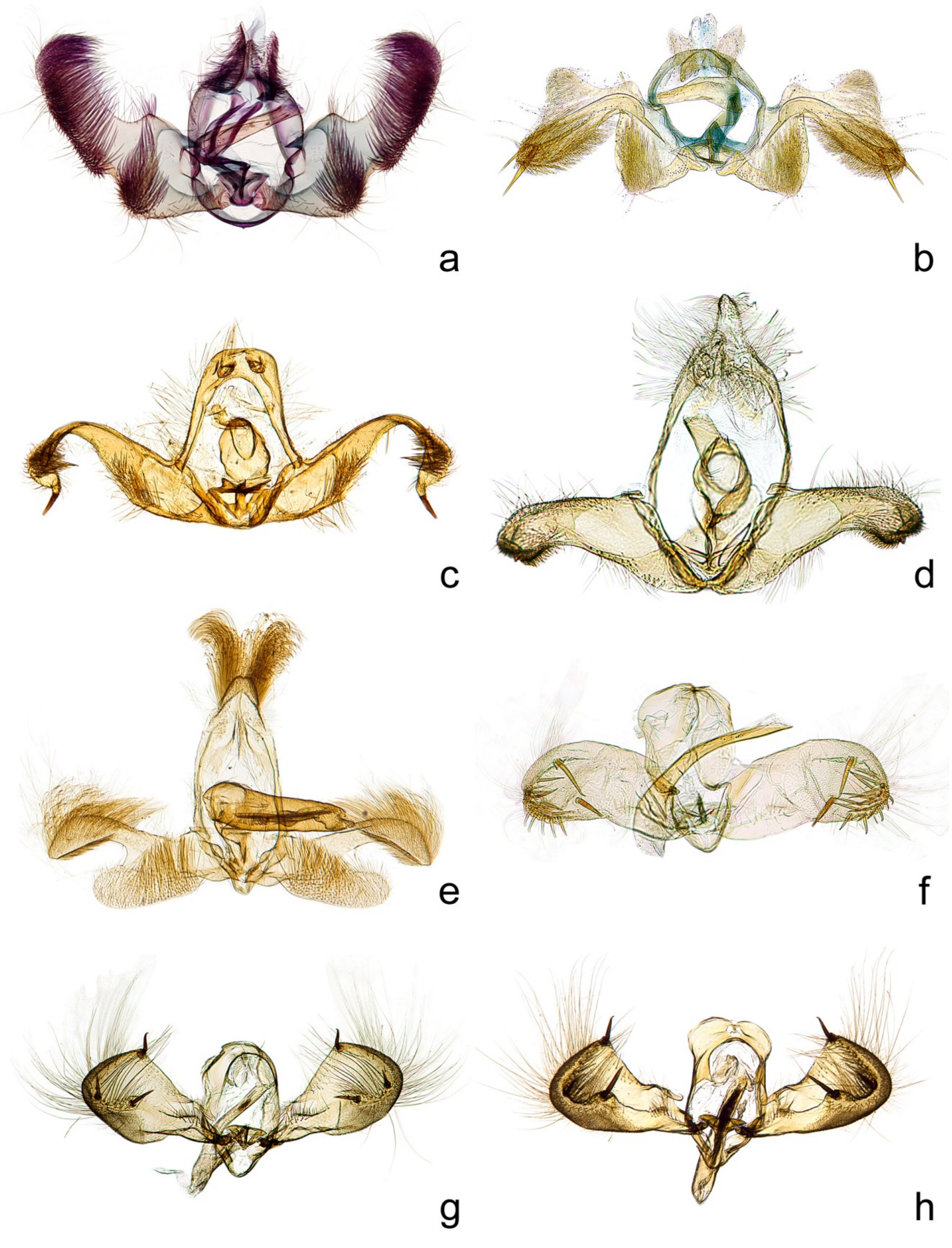


Figure 39. Male genitalia; a: *C. aporema* (photo credit Joaquín Baixeras Almela); b: *C. plebejana*; c: *S. ocellana*; d: *E. formosana*; e: *T. schistaceana*; f: *C. illepida*; g: *C. ombrodelta*; h: *C. peltastica*.

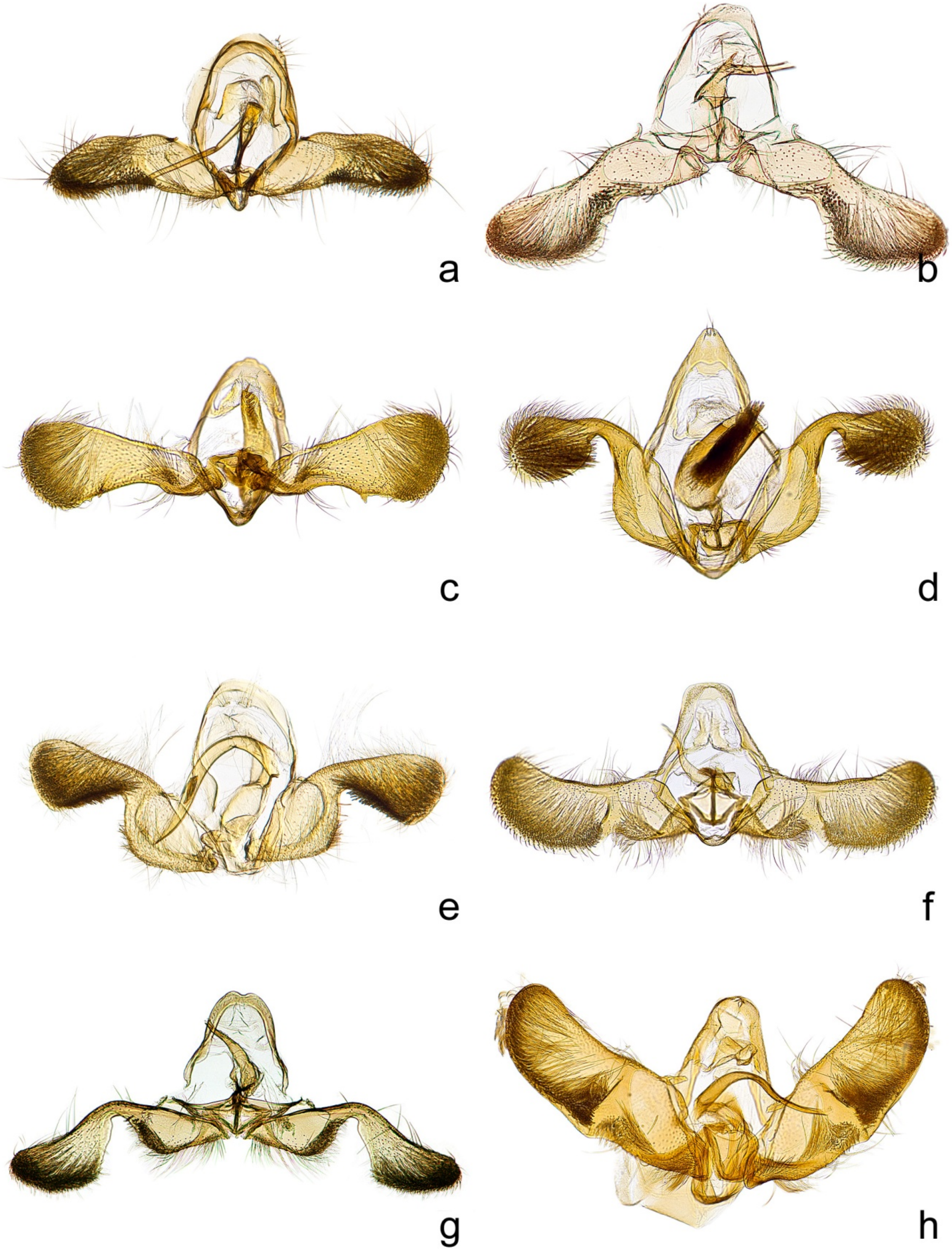


Figure 40. Male genitalia; a: *C. araucariae*; b: *C. caryana*; c: *C. coniferana*; d: *C. deshaisiana*; e: *C. fabivora*; f: *C. fagiglandana*; g: *C. garacana*; h: *C. glandicolana*.

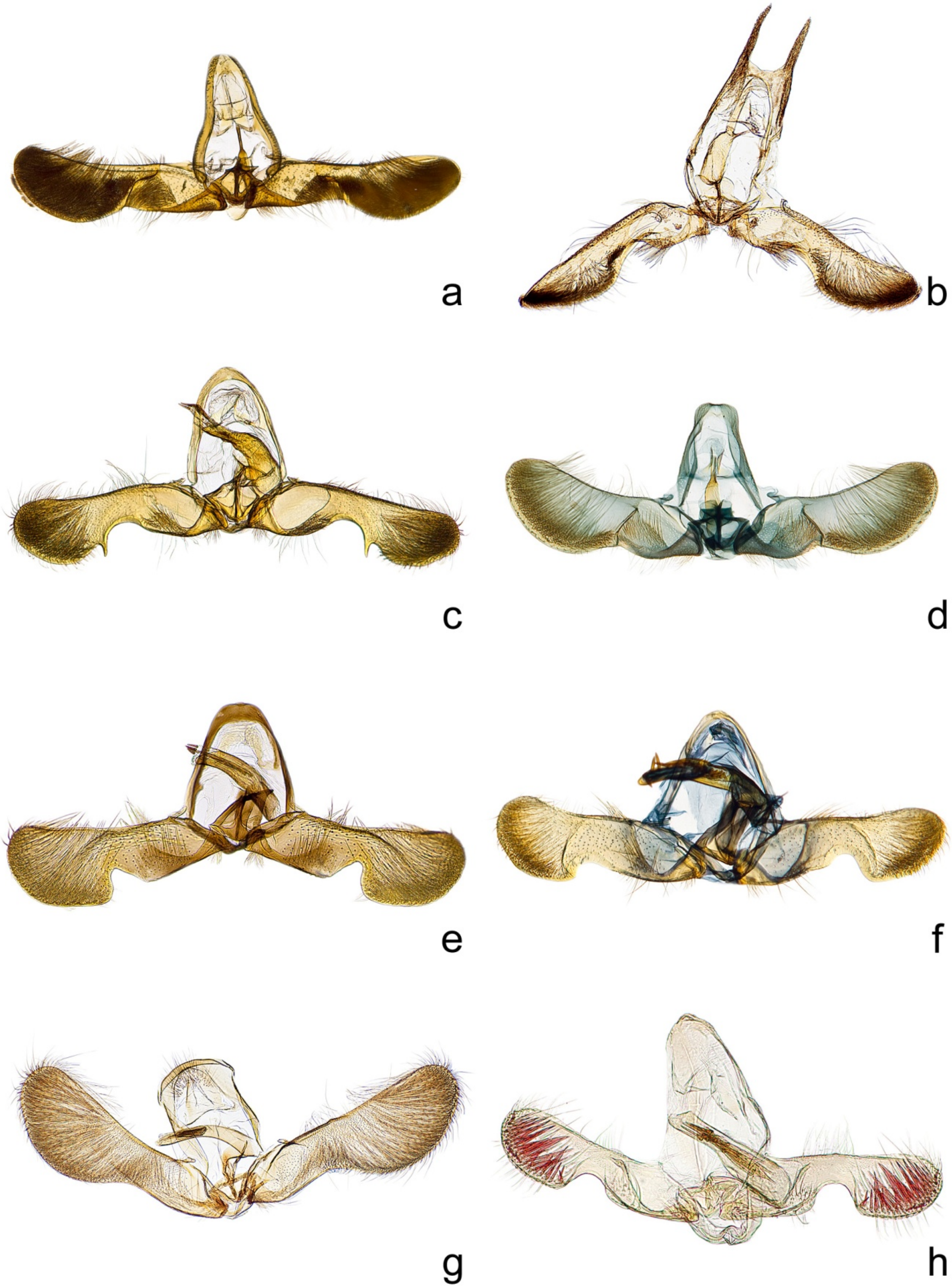


Figure 41. Male genitalia; a: *C. kurokoi*; b: *C. latiferreana*; c: *C. pomonella*; d: *C. splendana*; e: *C. strobilella*; f: *C. toreuta*; g: *E. insiticiana*; h: *G. delineana*.

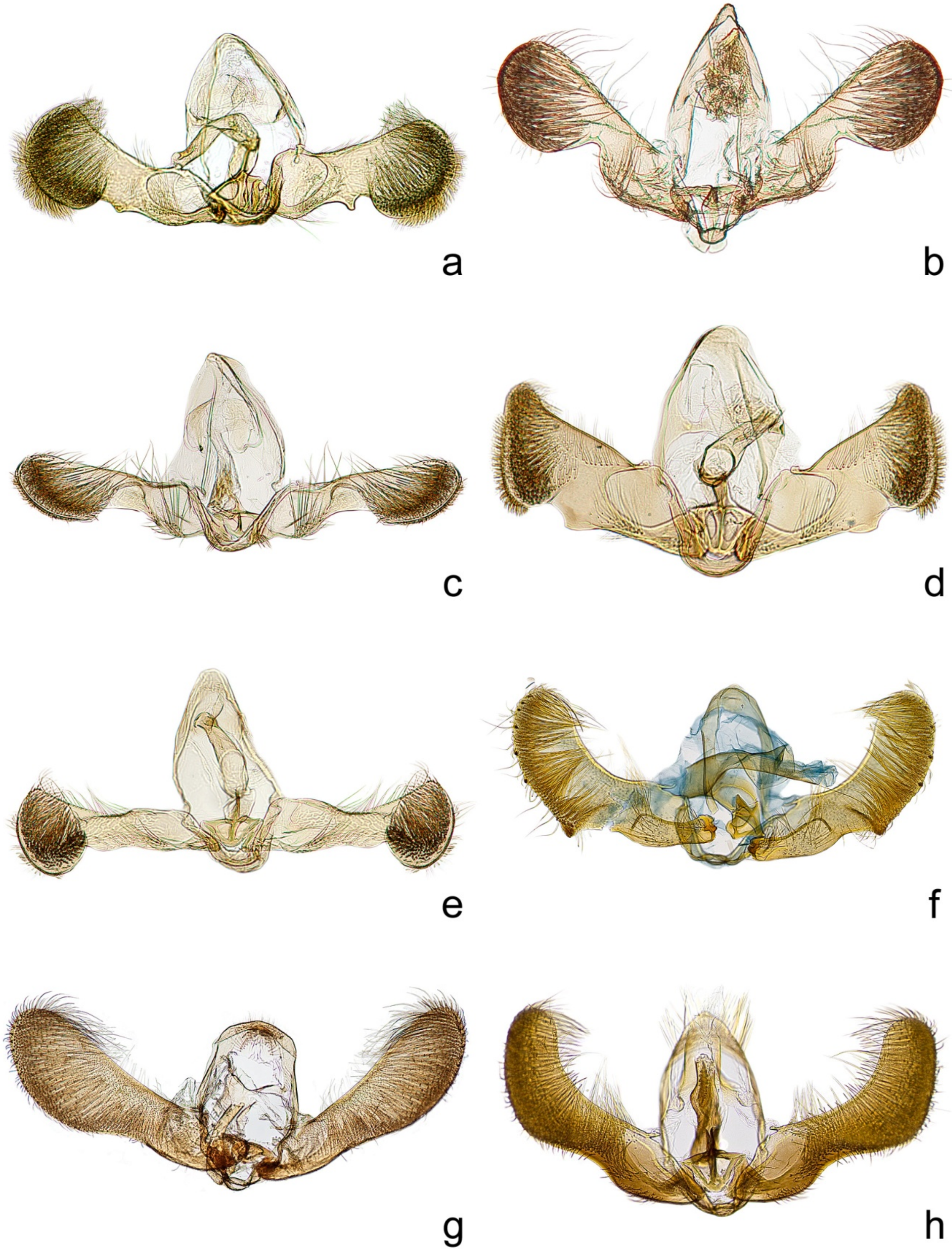


Figure 42. Male genitalia; a: *G. funebrana*; b: *G. interstinctana*; c: *G. molesta*; d: *G. packardi*; e: *G. prunivora*; f: *G. aurantianum*; g: *G. punctidiscanum*; h: *P. fasciana*.

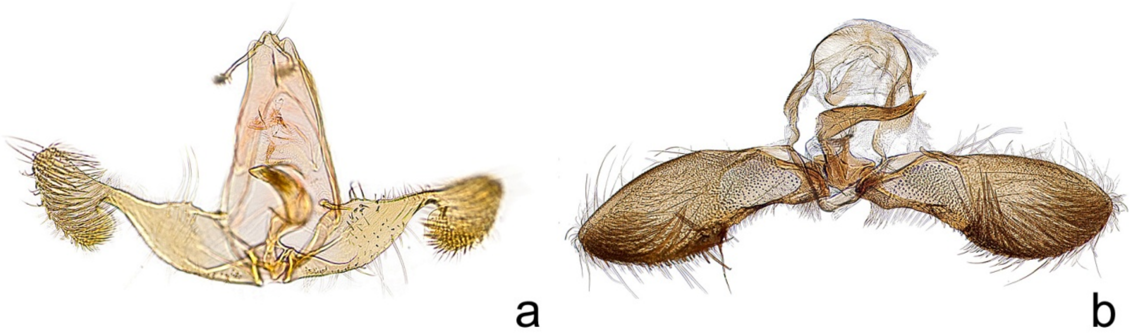


Figure 43. Male genitalia; a: *T. batesi*; b: *T. leucotreta*.

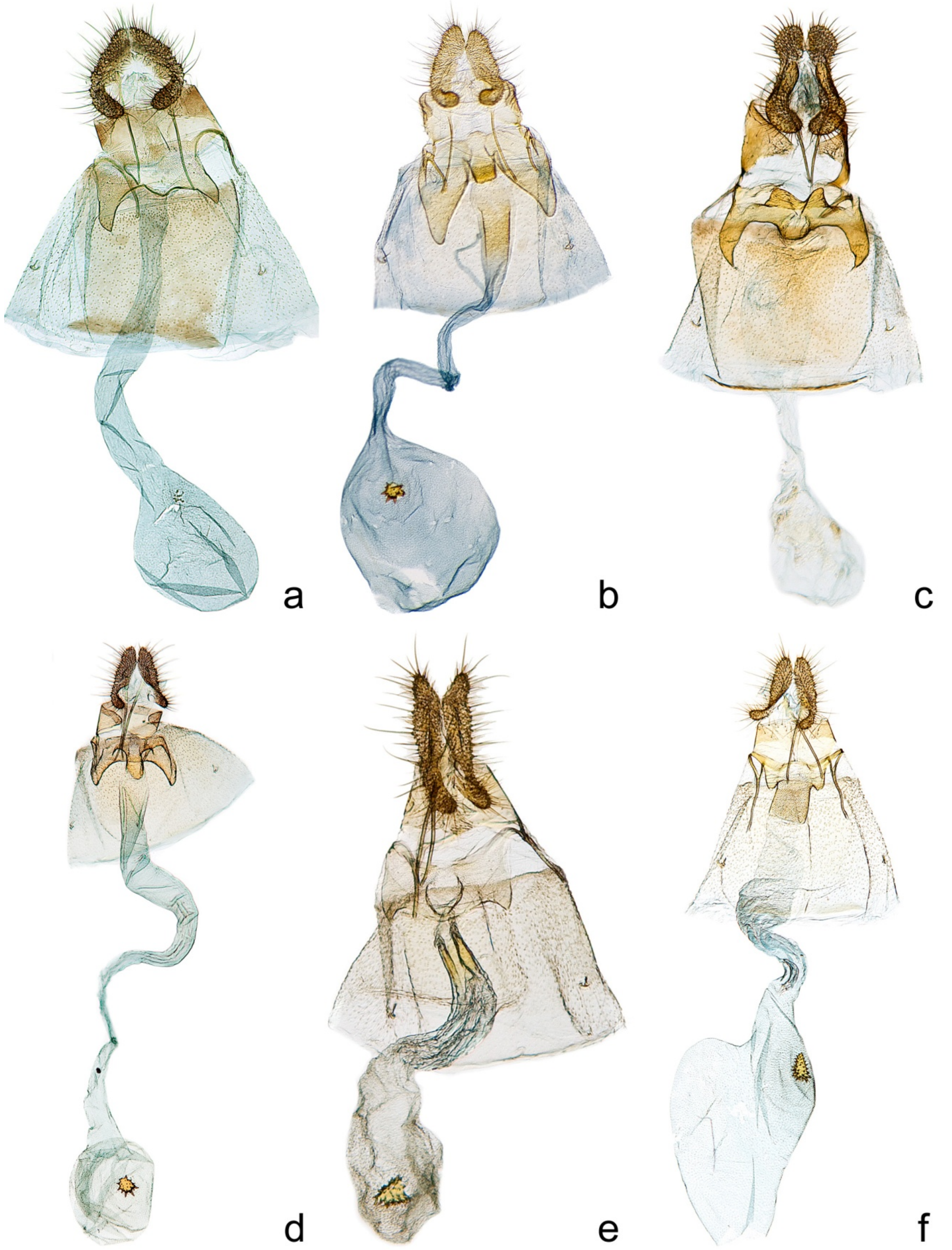


Figure 44. Female genitalia; a: *A. comariana*; b: *A. flavivittana*; c: *A. gloverana*; d: *A. hastiana*; e: *A. holmiana*; f: *A. minuta*.

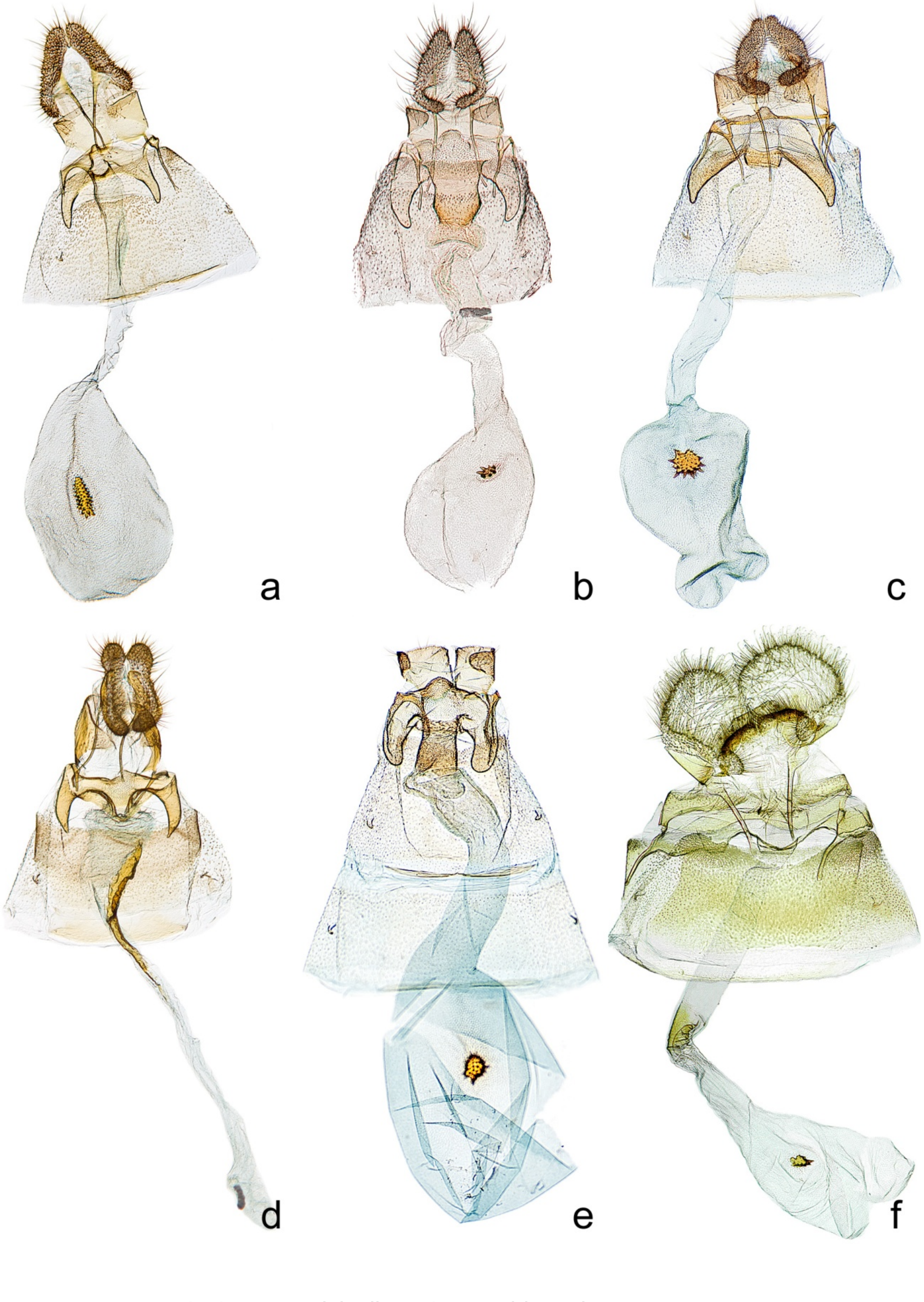


Figure 45. Female genitalia; a: *A. nivisellana*; b: *A. robinsoniana*; c: *A. senescens*; d: *A. variana*; e: *A. variegana*; f: *T. viridana*.

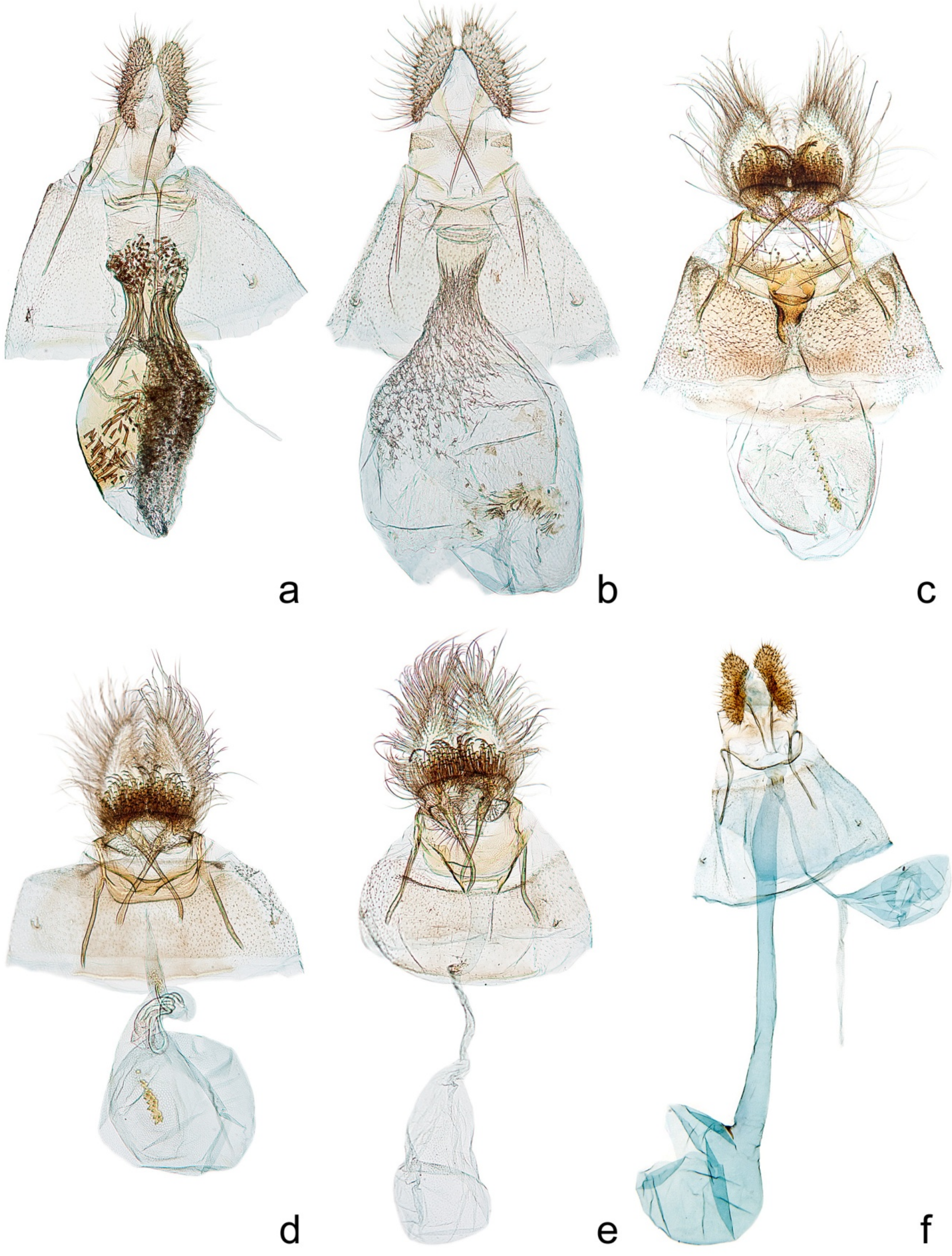


Figure 46. Female genitalia; a: *E. ambiguella*; b: *H. umbrabasana*; c: *C. longana*; d: *D. basiplagana*; e: *D. fragariana*; f: *A. furcatana*.

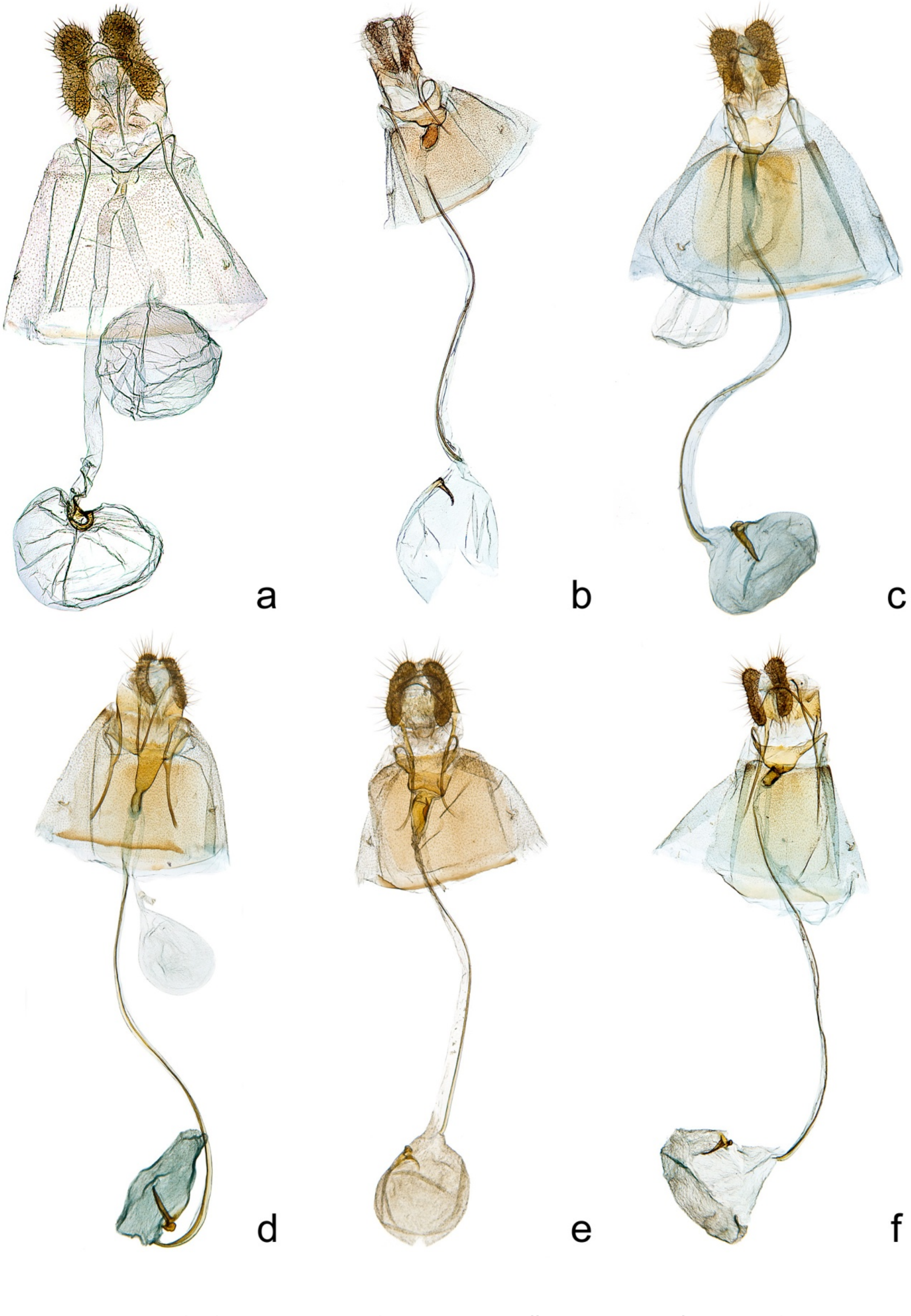


Figure 47. Female genitalia; a: *A. orana*; b: *A. argyrospila*; c: *A. cerasivorana*; d: *A. crataegana*; e: *A. fuscocupreanus*; f: *A. grisea*.

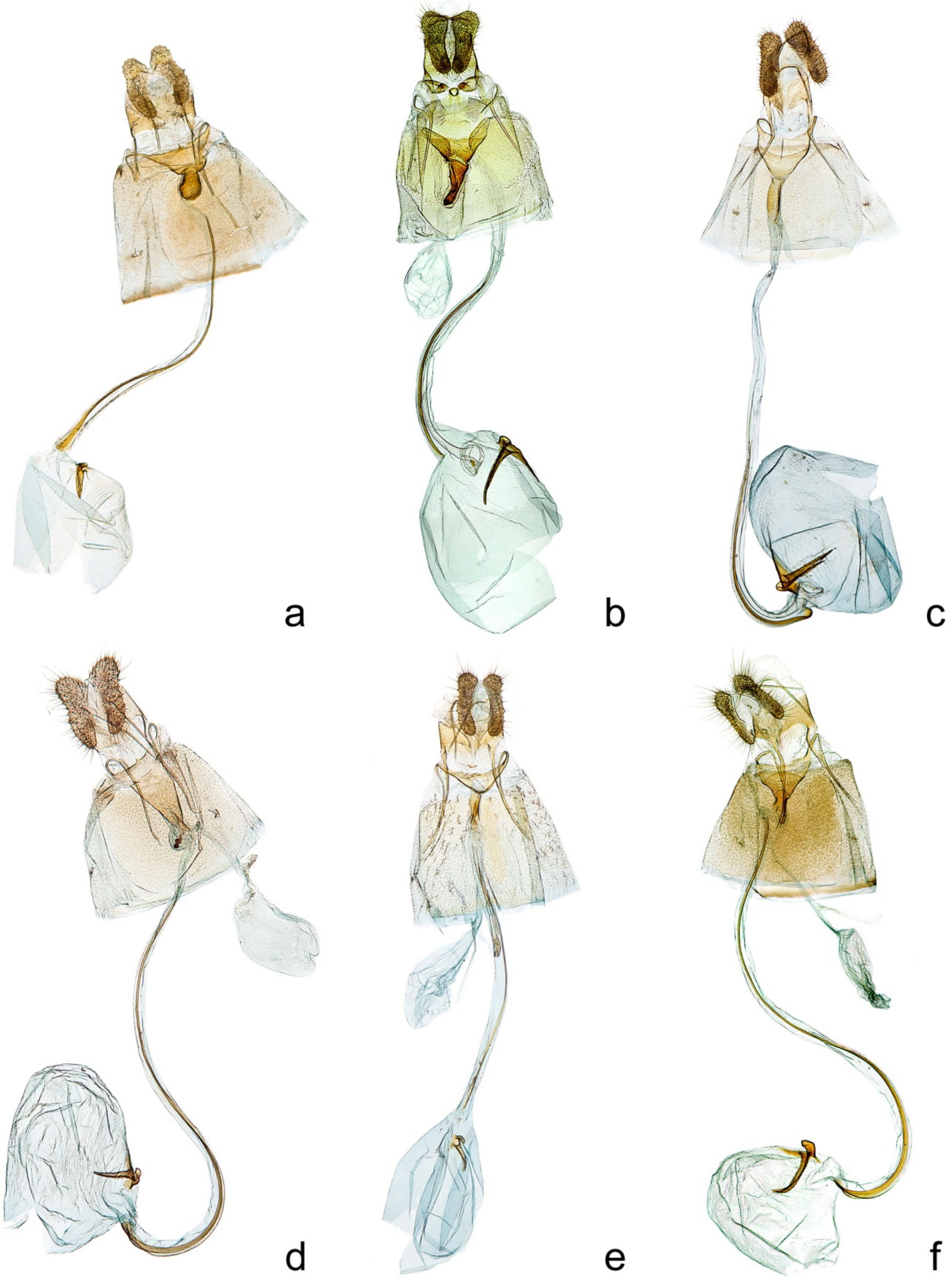


Figure 48. Female genitalia; a: *A. mortuana*; b: *A. podana*; c: *A. purpurana*; d: *A. rosana*; e: *A. semiferanus*; f: *A. xylosteana*.

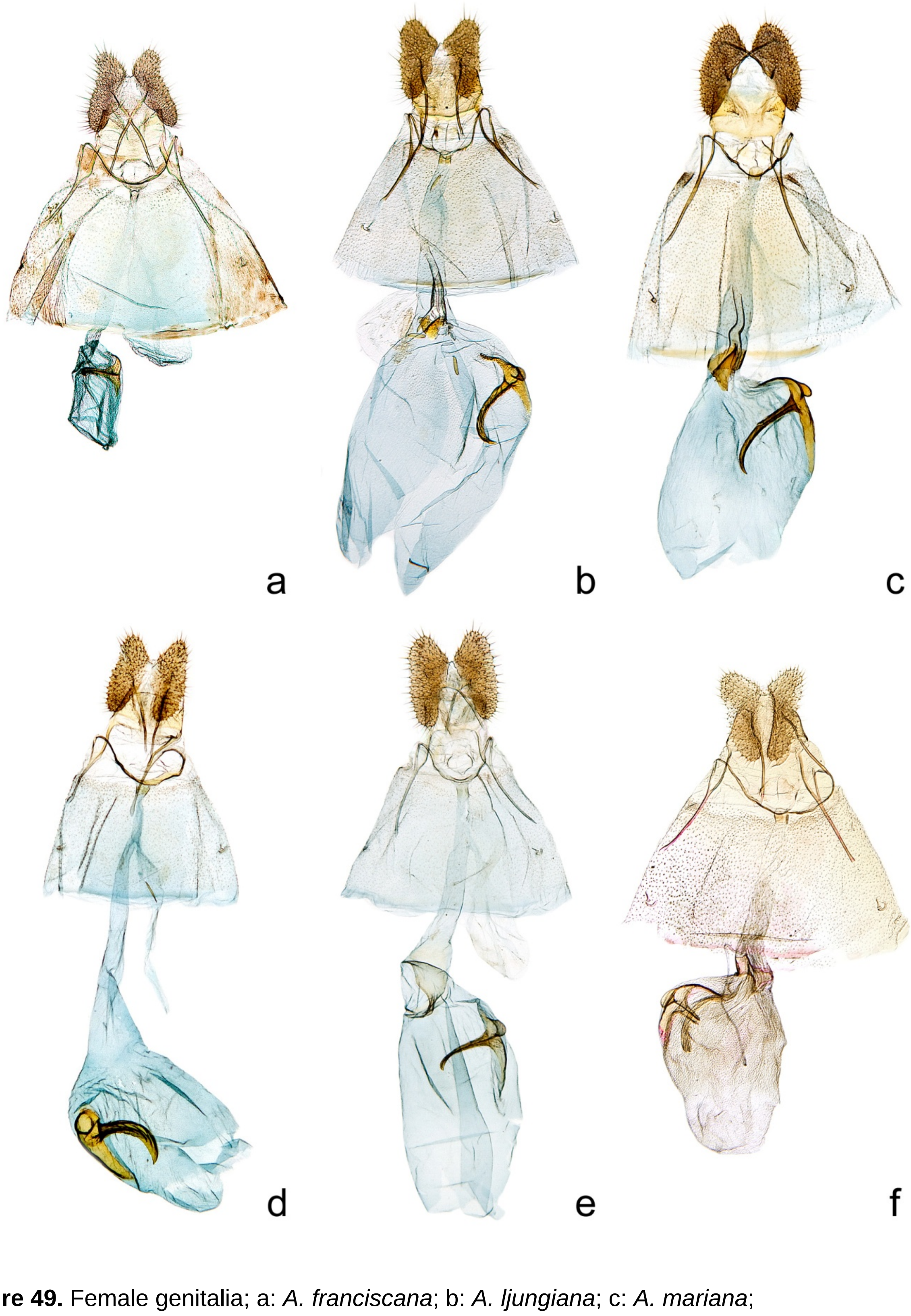


Figure 49. Female genitalia; a: *A. franciscana*; b: *A. ljungiana*; c: *A. mariana*; d: *A. quadrifasciana*; e: *A. quercifoliana*; f: *A. velutinana*.

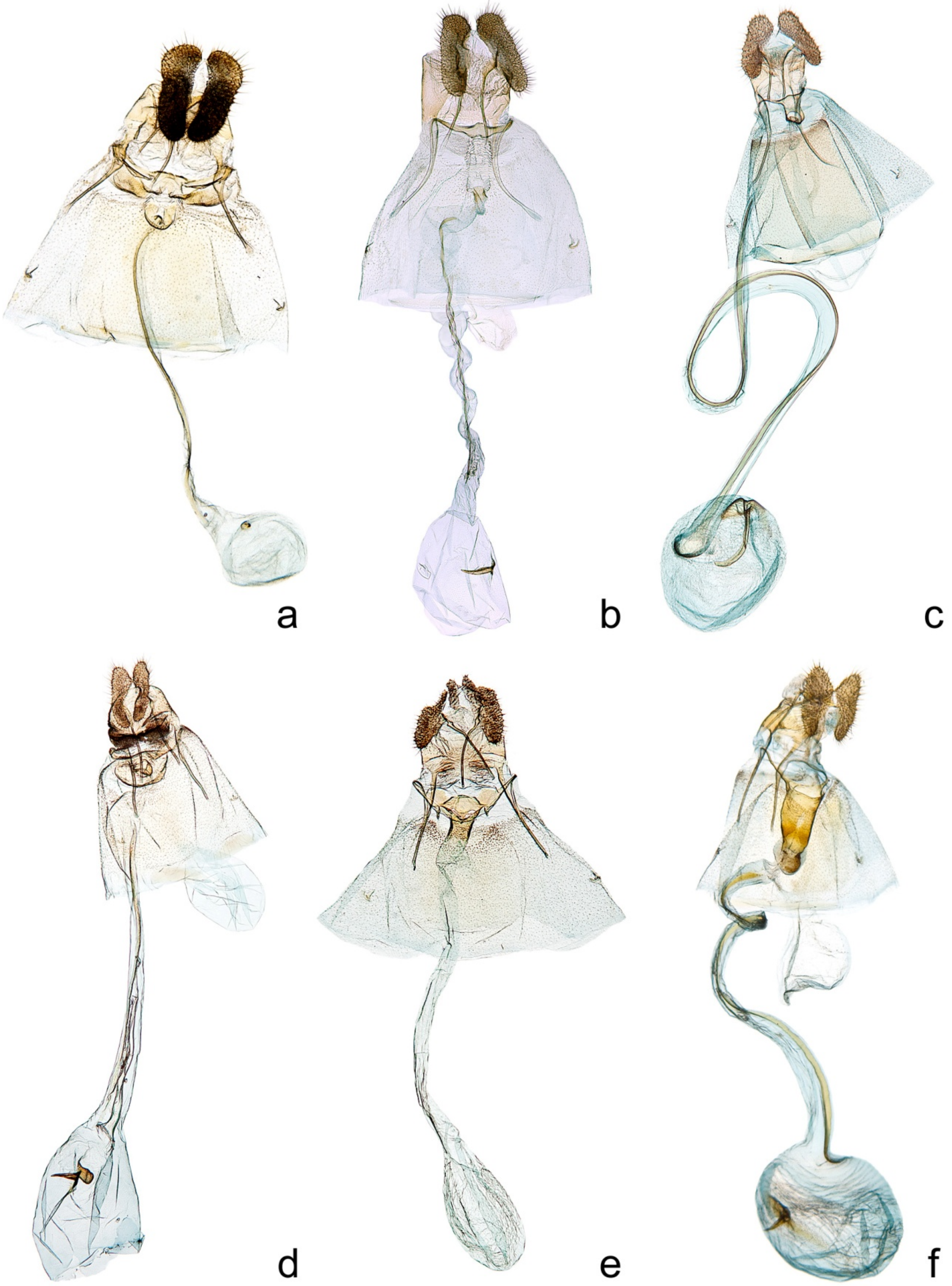


Figure 50. Female genitalia; a: *C. pronubana*; b: *C. occidentalis*; c: *C. rosaceana*; d: *C. clemensiana*; e: *C. fucana*; f: *C. melaleucanus*.

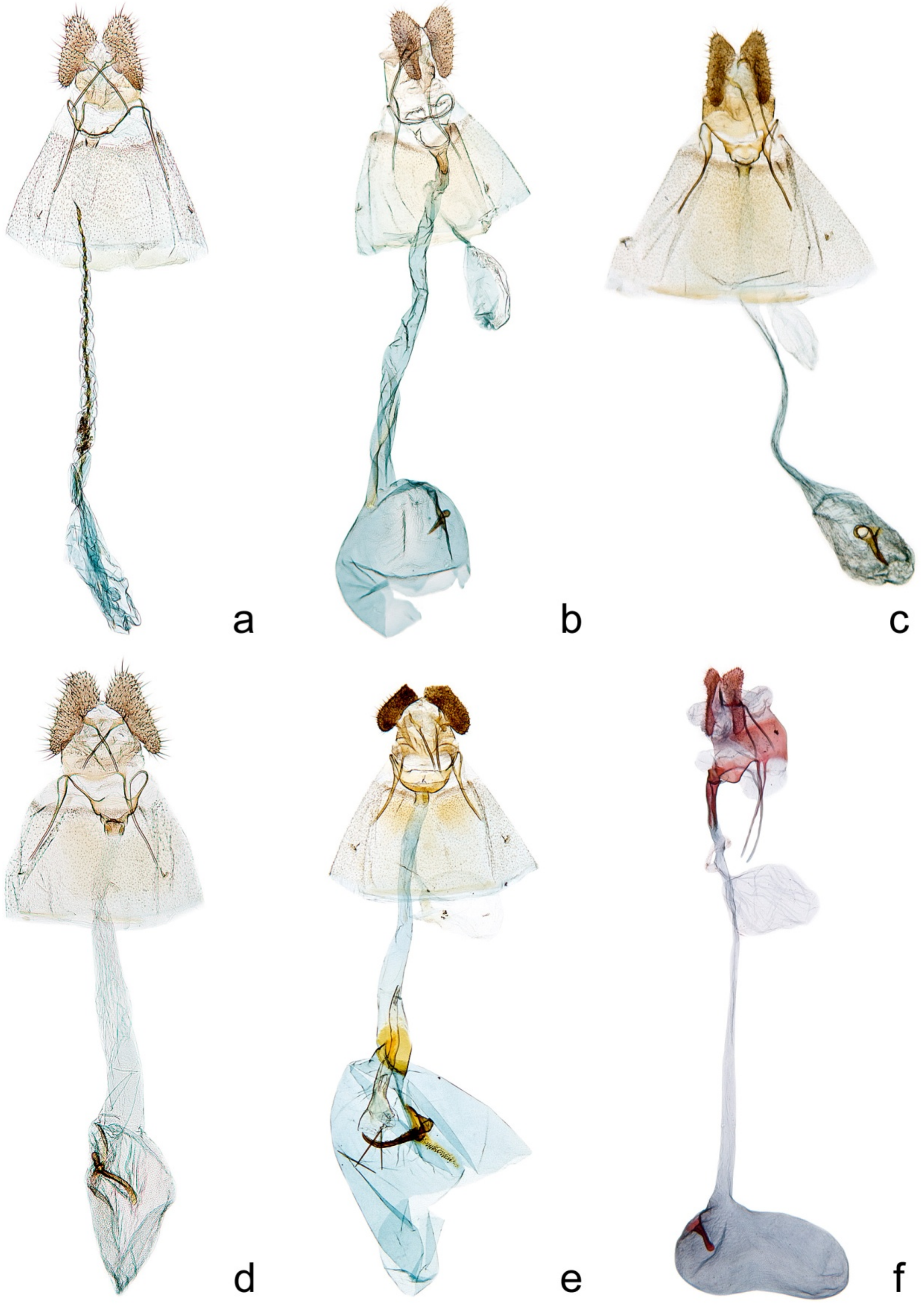


Figure 51. Female genitalia; a: *C. peritana*; b: *C. persicana*; c: *C. spectrana*; d: *C. virescana*; e: *C. obliquana*; f: *E. acerbella*.

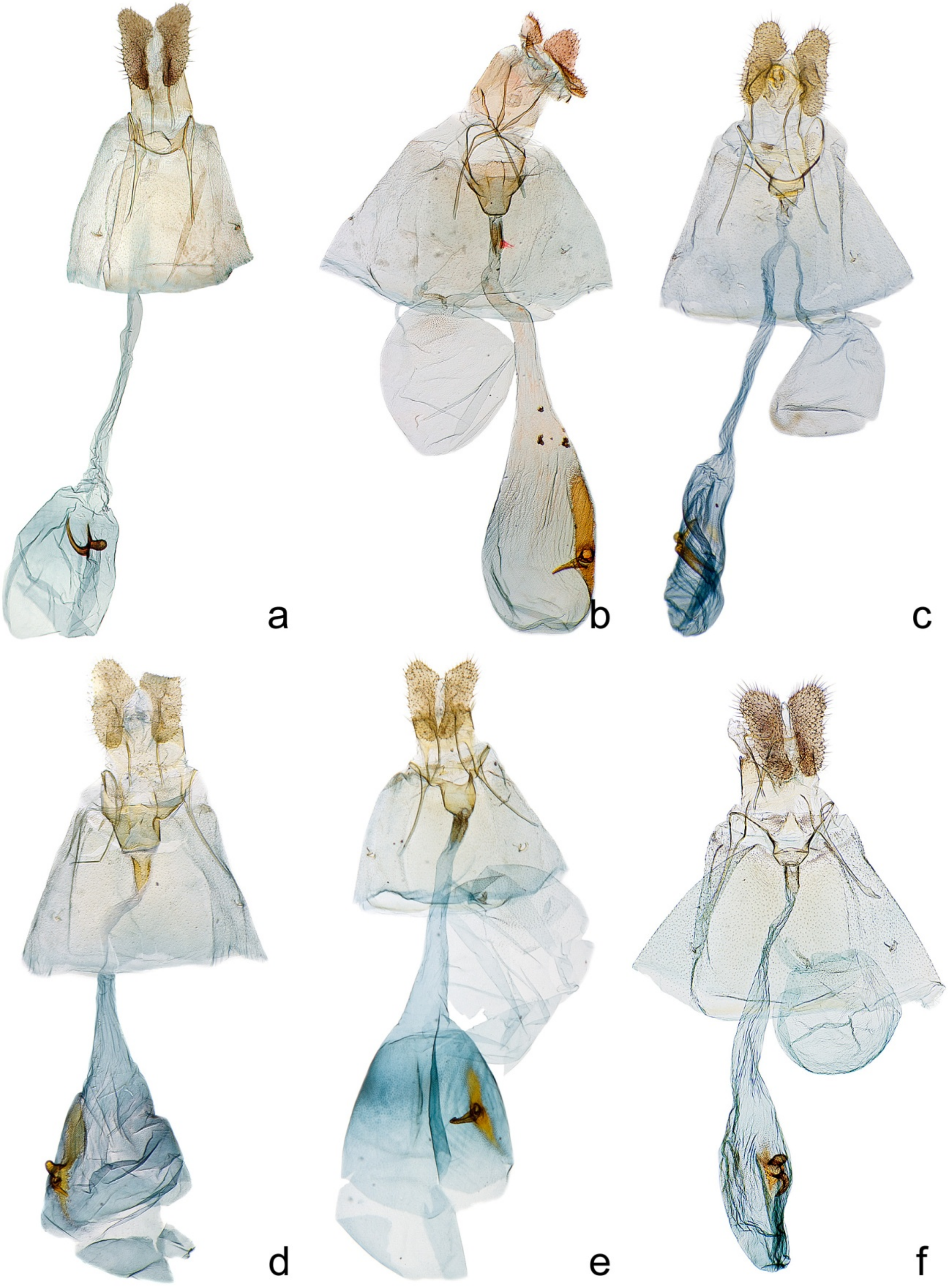


Figure 52. Female genitalia; a: *E. postvittana*; b: *P. cerasana*; c: *P. heparana*; d: *P. lamprosana*; e: *P. limitata*; f: *P. pyrusana*.

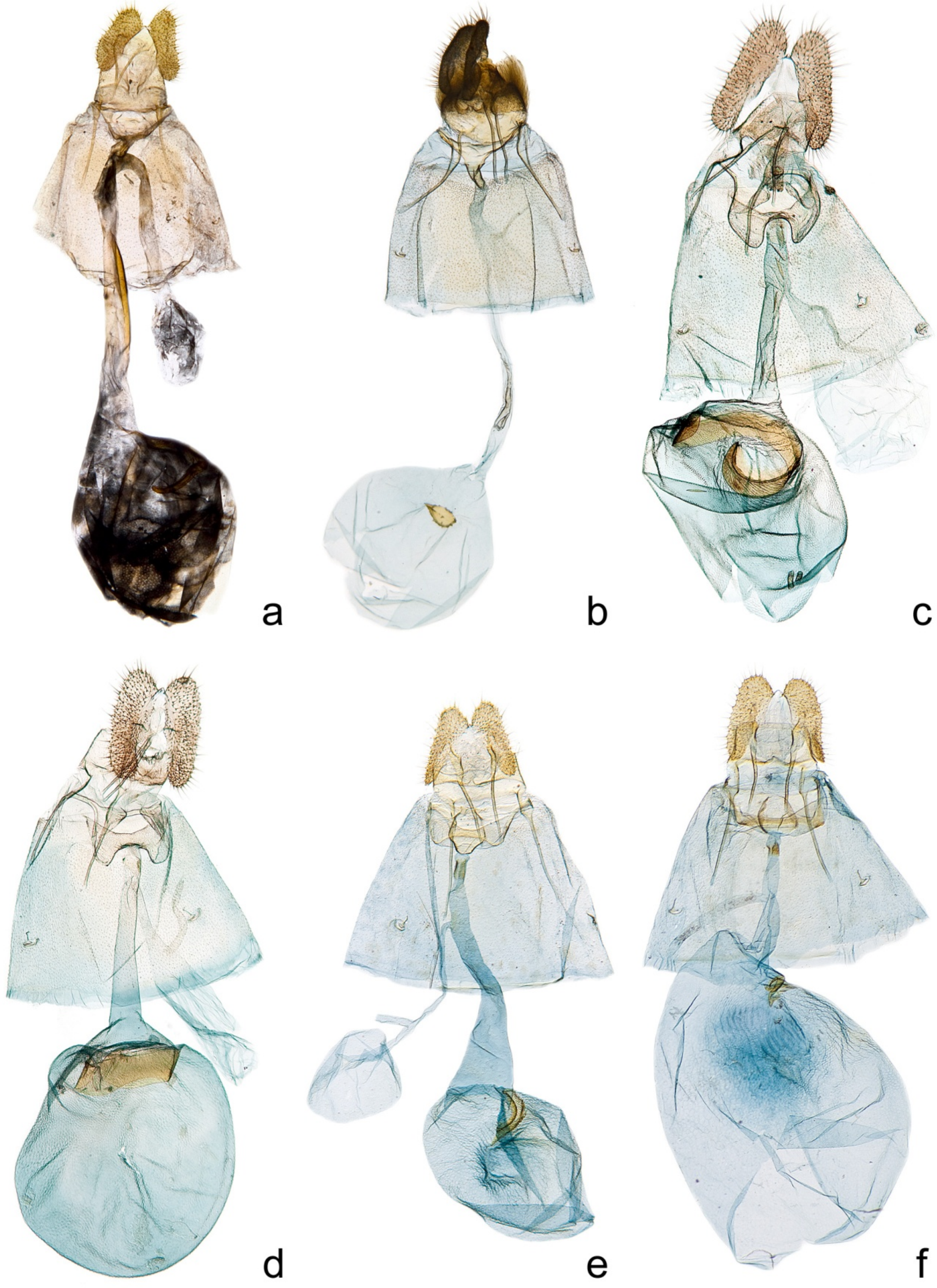


Figure 53. Female genitalia; a: *P. excessana*; b: *X. pallorana*; c: *A. cuneana*; d: *A. emigratella*; e: *P. flavedana*; f: *P. idaeusalis*.

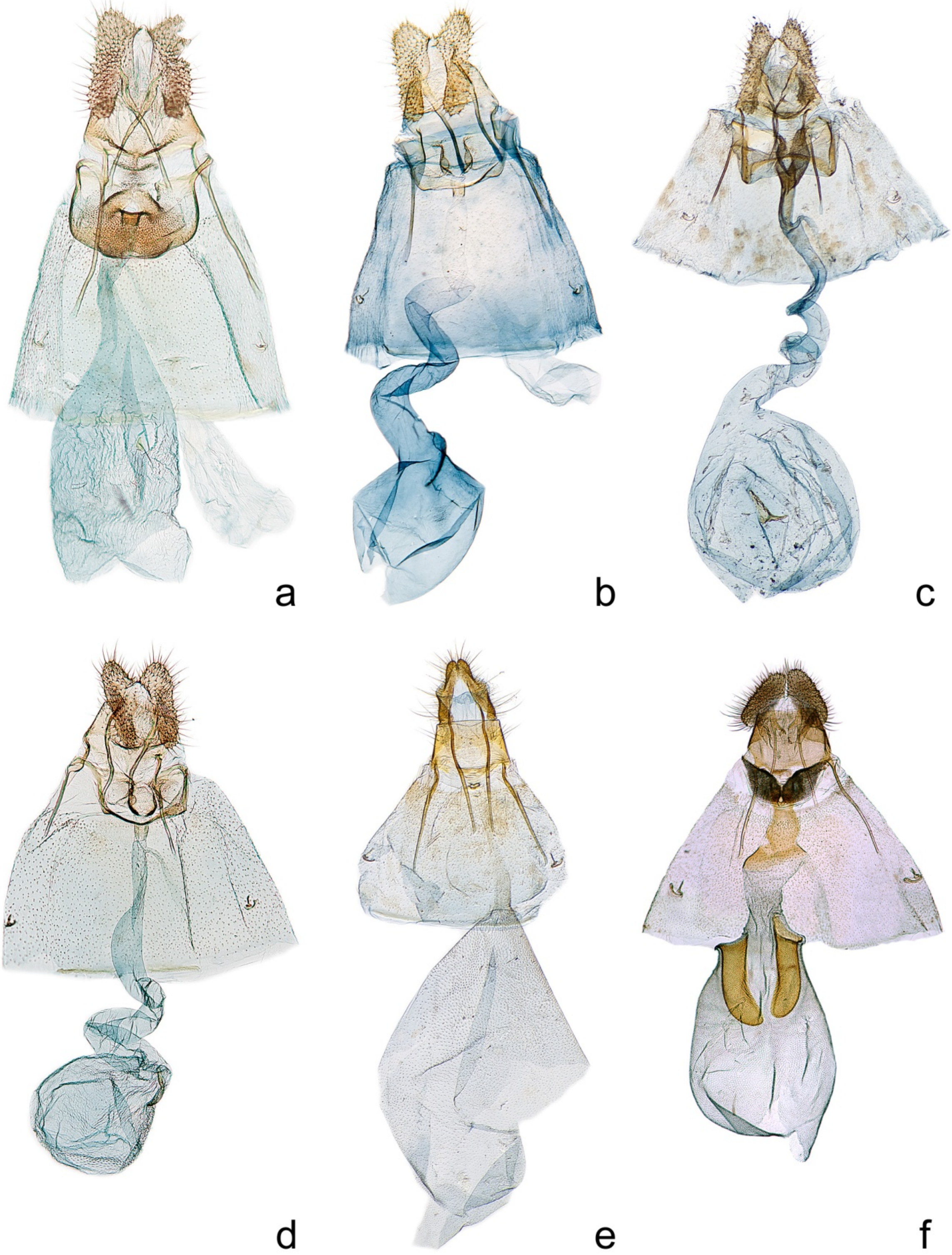


Figure 54. Female genitalia; a: *P. stultana*; b: *S. senecionana*; c: *S. sulfureana*; d: *S. tunicana*; e: *S. rubicola*; f: *C. bipenicilla*.

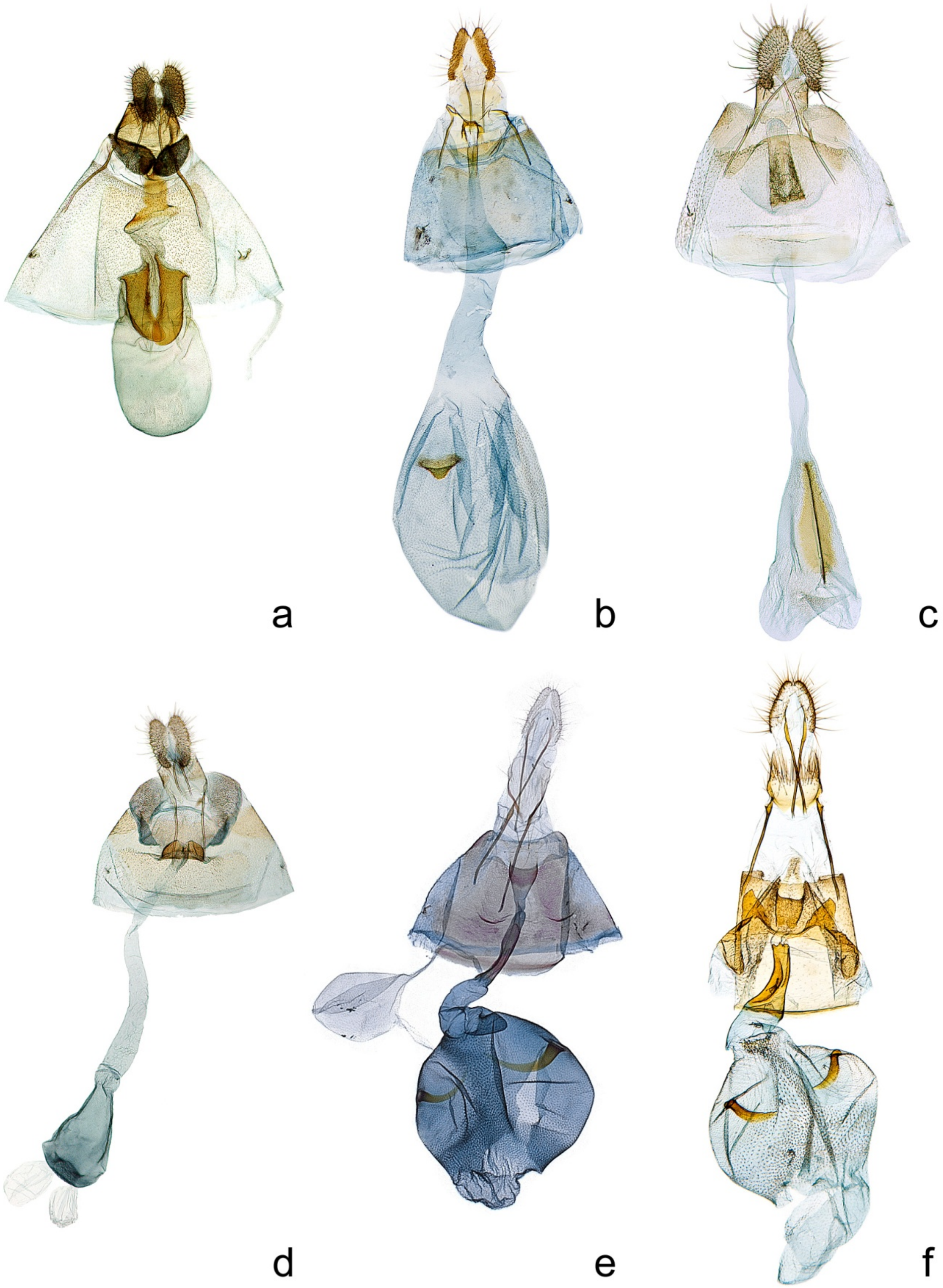


Figure 55. Female genitalia; a: *C. perseana*; b: *E. hebesana*; c: *L. botrana*; d: *P. viteana*; e: *C. aporema* (photo credit Joaquín Baixeras Almela); f: *C. plebejana*.

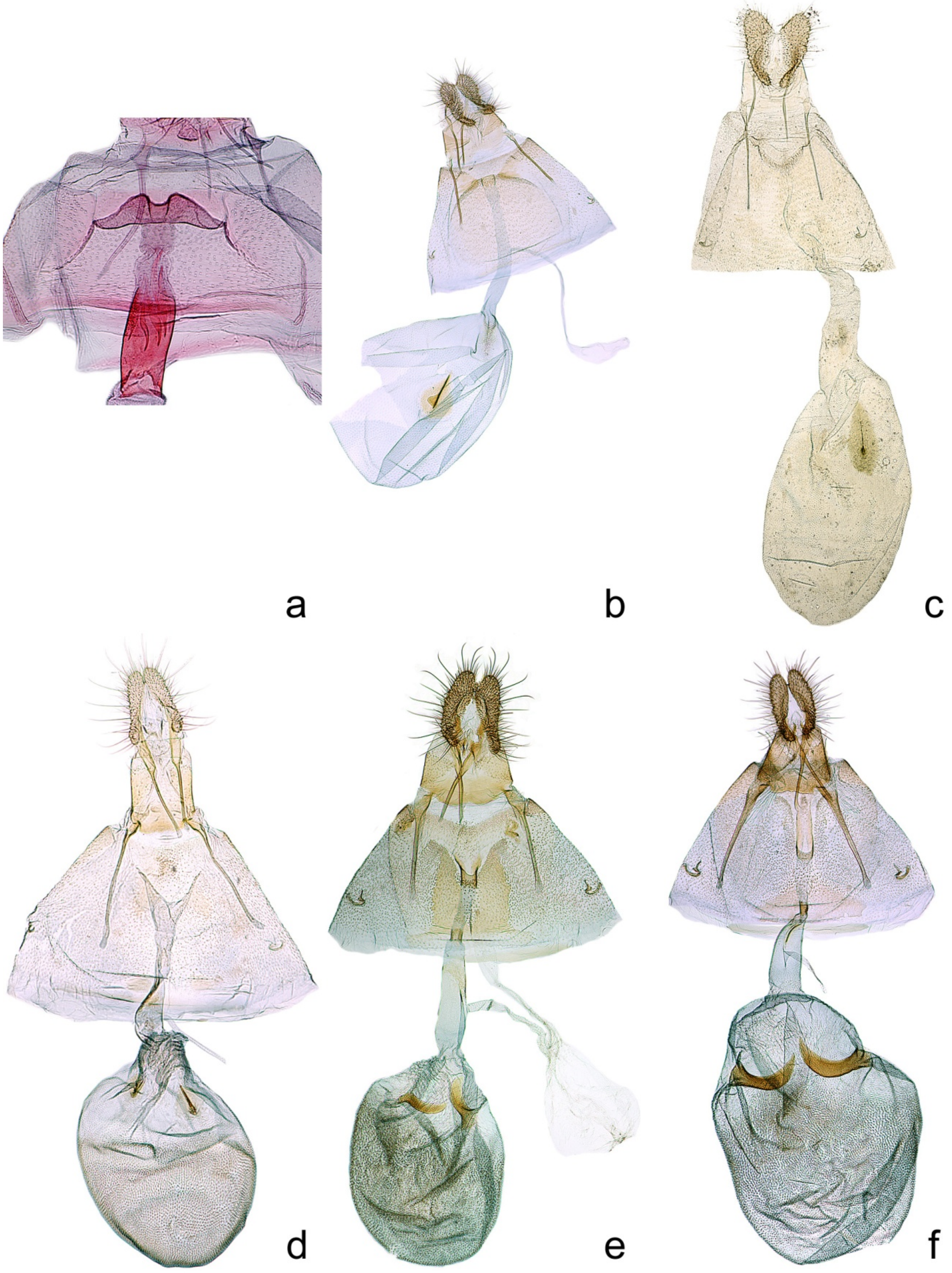


Figure 56. Female genitalia; a: *S. ocellana*; b: *E. formosana*; c: *T. schistaceana*; d: *C. illepida*; e: *C. ombrodelta*; f: *C. peltastica*.

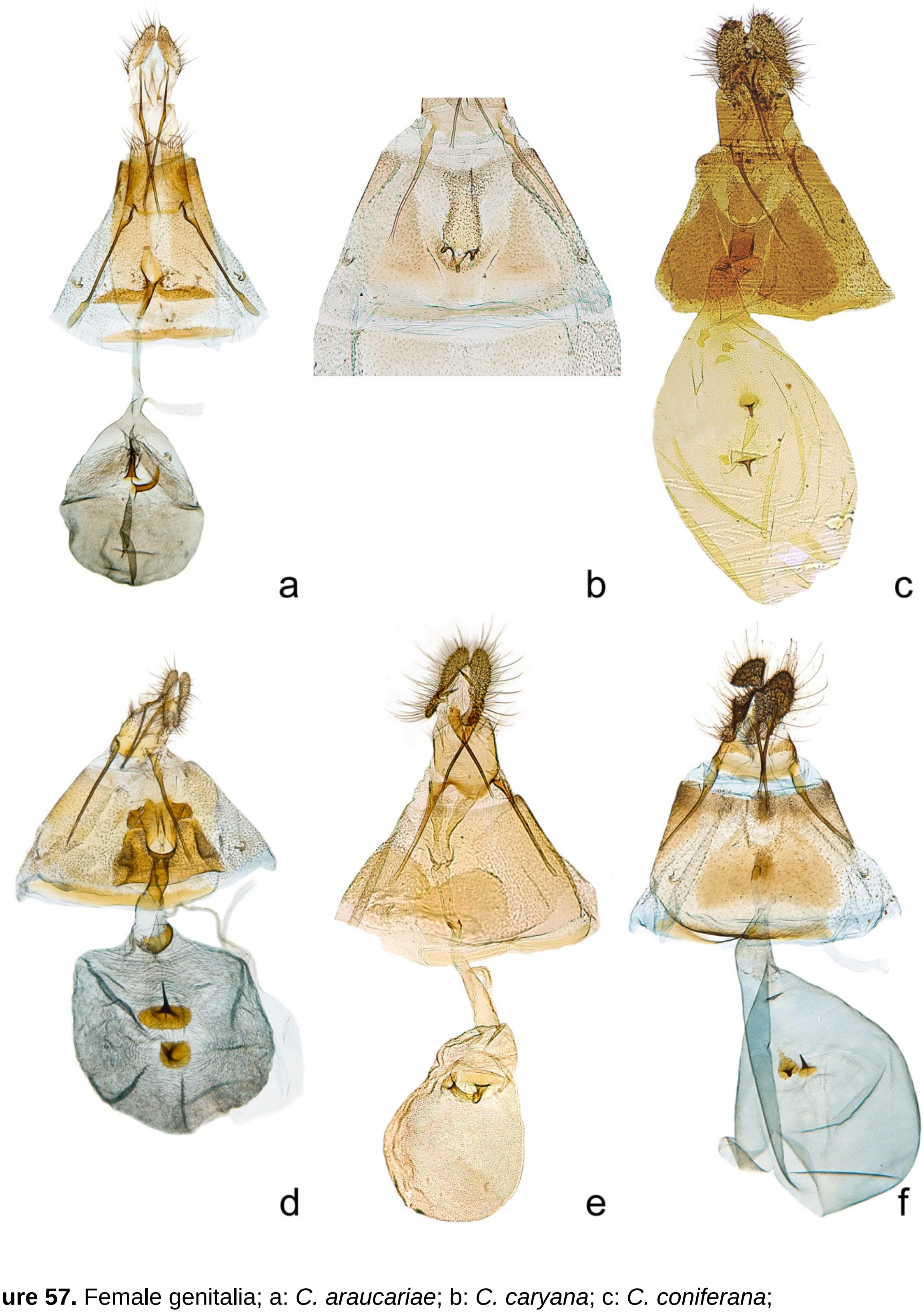


Figure 57. Female genitalia; a: *C. araucariae*; b: *C. caryana*; c: *C. coniferana*; d: *C. deshaisiana*; e: *C. fabivora*; f: *C. fagiglandana*.

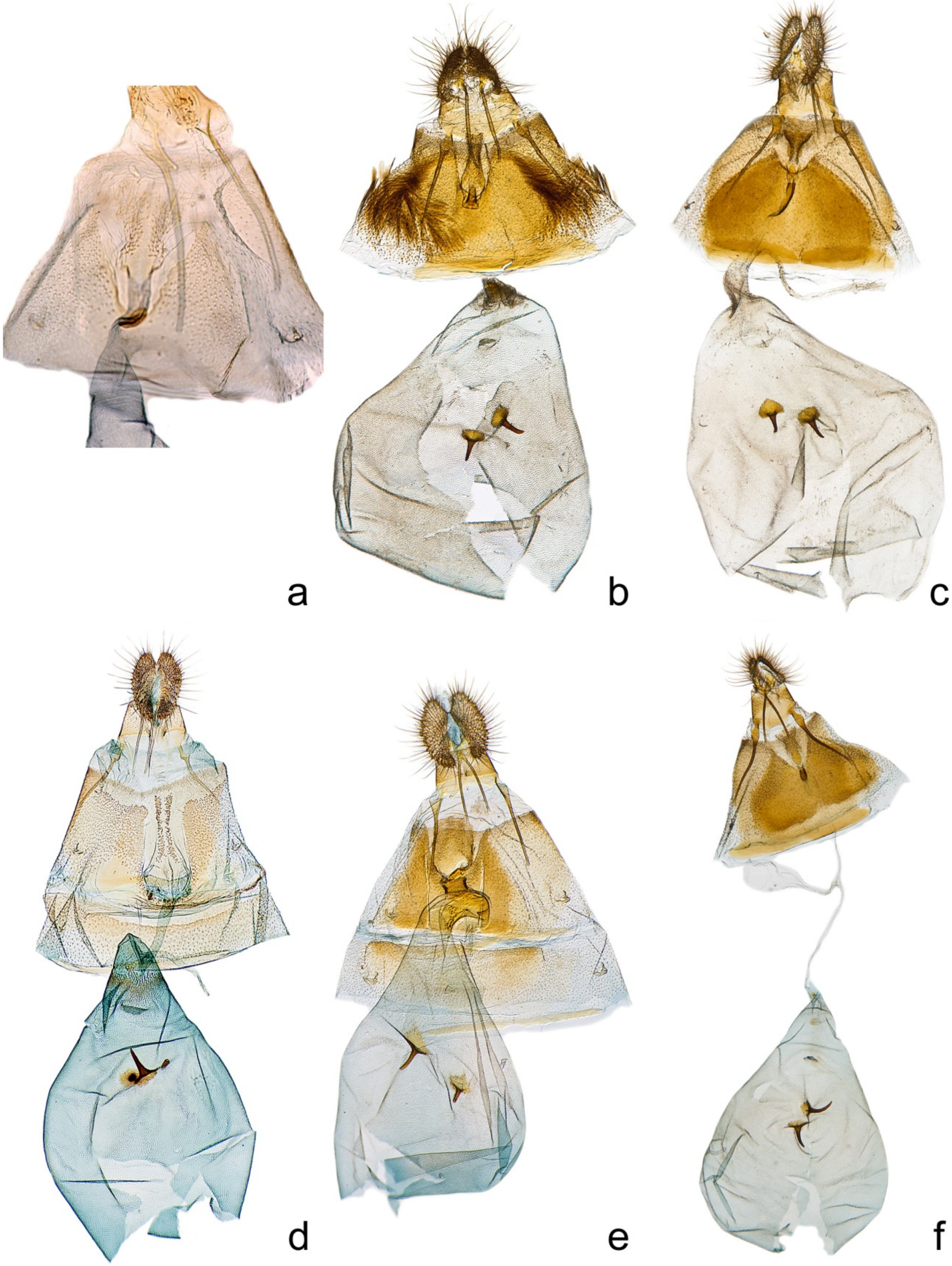


Figure 58. Female genitalia; a: *C. garacana*; b: *C. glandicolana*; c: *C. kurokoi*; d: *C. latiferreana*; e: *C. pomonella*; f: *C. splendana*.

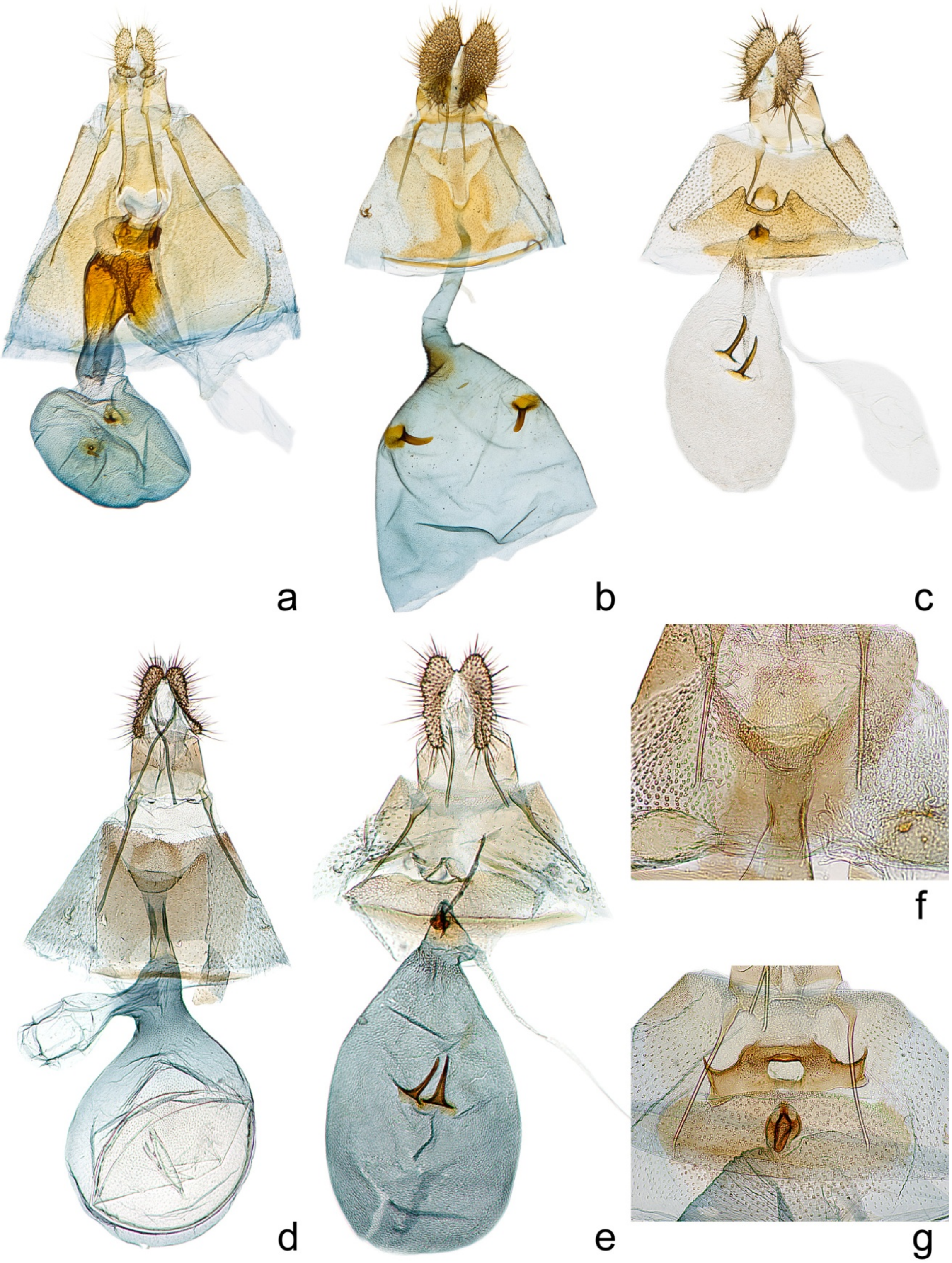


Figure 59. Female genitalia; a: *C. toreuta*; b: *E. insiticiana*; c: *G. funebrana*; d: *G. interstinctana*; e: *G. packardi*; f: *G. delineana*; g: *G. molesta*.

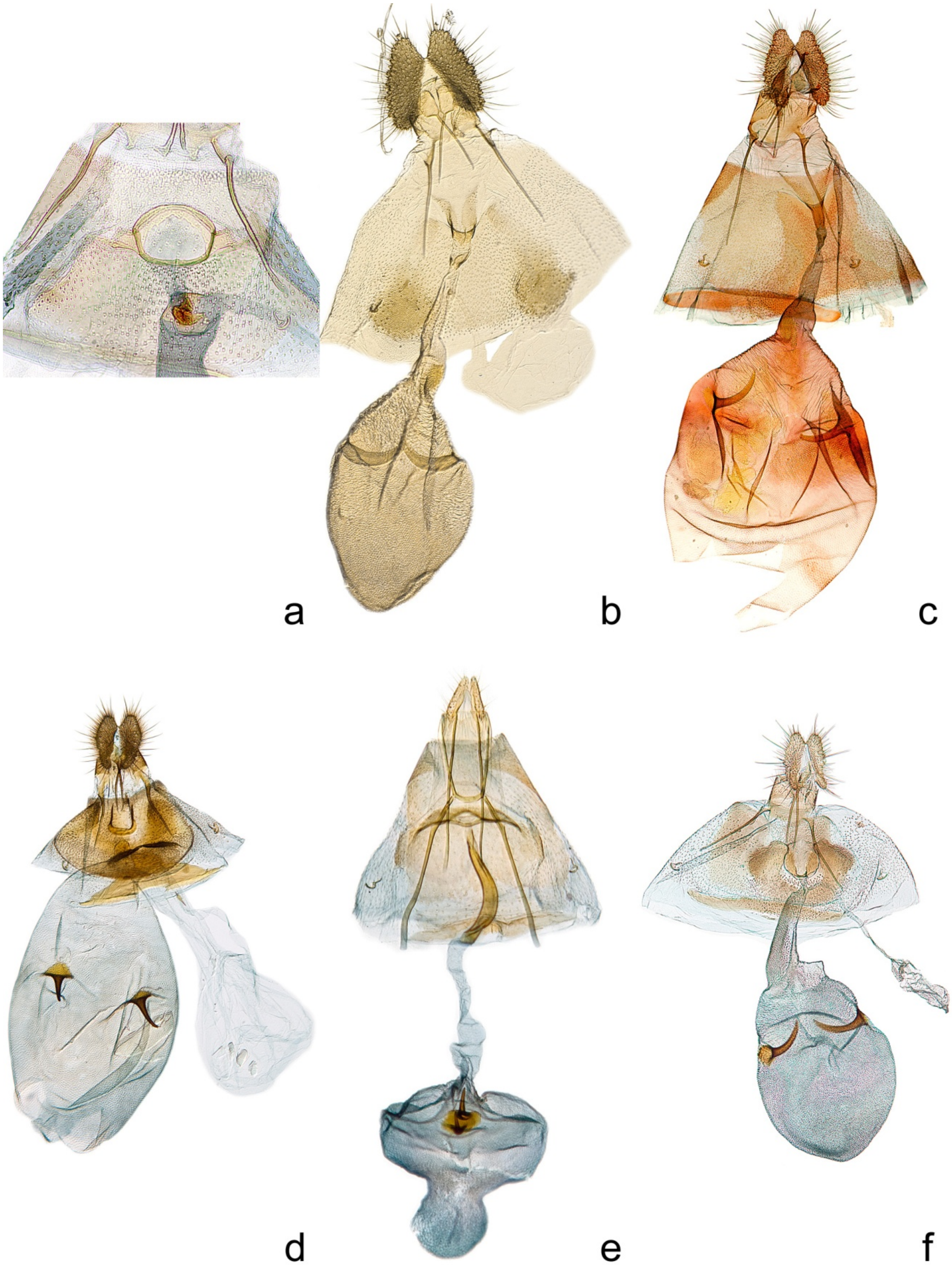


Figure 60. Female genitalia; a: *G. prunivora*; b: *G. aurantianum*; c: *G. punctidiscanum*; d: *P. fasciana*; e: *T. batesi*; f: *T. leucotreta*.

References

Aarvik, L. (2005) Revision of African *Cryptaspasma* Walsingham, 1900 (Lepidoptera: Tortricidae). Norwegian Journal of Entomology. 51: 193–201.

Abrahamson, L. P. and K. J. Kraft. 1965. A population study of the cone moth *Laspeyresia toreuta* Grote in Pinus banksiana stands. Ecology. 46: 561–563.

Adamski, D. and J. W. Brown. 2001. Systematic revision of the *Ecdytolopha* group of genera (Lepidoptera: Tortricidae: Grapholitini) in the New World. Entomologica Scandinavica Supplement 58. 86 pp.

Alford, D. V. 1978. Observations on the specificity of pheromone-baited traps for *Cydia funebrana* (Treitschke) (Lepidoptera: Tortricidae). Bulletin of Entomological Research. 68: 97–103.

Altesor, P., V. R. Horas, M. P. Arcia, C. Rossini, P. H. Zarbin and A. Gonzalez. 2010. Reproductive behaviour of *Crocidosema* (*Epinotia*) *aporema* (Walsingham) (Lepidoptera: Tortricidae): Temporal pattern of female calling and mating. Neotropical Entomology. 39: 324–329.

AliNiazee, M. T. and E. M. Stafford. 1972. Notes on the biology, ecology, and damage of *Platynota stultana* on grapes. Journal of Economic Entomology. 65: 1042–1044.

Arneodo, J., G. Quintana and A. Sciocco-Cap. 2010. Biology and morphometrics of the immature stages of *Epinotia aporema* on artificial diet. Pesquisa Agropecuaria Brasileira. 45: 221–223.

Atkins, E. L., M. H. Frost, L. D. Anderson and A. S. Deal. 1957. The omnivorous leaf roller, *Platynota stultana* Wlshm., on cotton in California: nomenclature, life history, and bionomics (Lepidoptera, Tortricidae). Annals of the Entomological Society of America. 50: 251–259.

Baixeras, J., J. W. Brown & T. M. Gilligan. 2010. T@RTS: Online World Catalogue of the Tortricidae (Version 1.4.0). http://www.tortricidae.com/catalogue.asp.

Baker, R. T. and H. B. Dick. 1981. Leafrollers (Lepidoptera: Tortricidae) in a strawberry crop in Horowhenua: abundance and control. New Zealand Journal of Experimental Agriculture. 9: 377–381.

Barnes, M. M. 1991. Codling moth occurance, host race formation, and damage, pp. 313–327. In L. P. S. van der Geest and H. H. Evenhius [eds.], Tortricid Pests: Their Biology, Natural Enemies, and Control. World Crop Pests, Vol. 5. Elsevier, Amsterdam.

Barr, N. B., L. A. Ledezma, J. D. Vasquez, M. E. Epstein, P. H. Kerr, S. Kinnee, O. Sage and T. M. Gilligan. 2009. Molecular identification of the light brown apple moth (Lepidoptera: Tortricidae) in California using a polymerase chain reaction assay of the internal transcribed spacer 2 locus. Journal of Economic Entomology. 102(6): 2333–2342.

Barr, N. B., L. A. Ledezma, R. E. Farris, M. E. Epstein and T. M. Gilligan. 2011. A multiplex real-time polymerase chain reaction assay to diagnose *Epiphyas postvittana* (Lepidoptera: Tortricidae). Journal of Economic Entomology. 104(5): 1706–1719.

Beckwith, C. S. 1938. *Sparganothis sulfureana* Clem., a cranberry pest in New Jersey. Journal of Economic Entomology. 31: 253–256.

Beeke, H. and D. J. De Jong. 1991. Identification of Larvae and Pupae, pp. 65–76. In L. P. S. van der Geest and H. H. Evenhius [eds.], Tortricid Pests: Their Biology, Natural Enemies, and Control. World Crop Pests, Vol. 5. Elsevier, Amsterdam.

Biosecuity Australia. 2003. Table grapes from Chile, Draft import risk analysis report, Part B. Australian Department of Agriculture, Fisheries and Forestry. http://www.daff.gov.au/__data/assets/pdf_file/0018/11547/ira_tg_chile_b.pdf [accessed 5 Oct 2011].

Bishop, A. L. and P. R. B. Blood. 1978. Temporal distribution, biology and life history of the cotton tipworm, *Crocidosema plebiana* [sic] Zeller, on cotton in the south-eastern Queensland region. Australian Journal of Zoology. 26: 147–152.

Blanco-Metzler, H., A. D. Watt and D. Cosens. 2009. The effect of parasitism on the population dynamics of the macadamia nutborer *Gymnandrosoma aurantianum* (Lepidoptera: Tortricidae). Revista de Biologia Tropical. 57: 1245–1252.

Bogenschutz, H. 1991. Eurasian species in forestry, pp. 673–709. In L. P. S. van der Geest and H. H. Evenhius [eds.], Tortricid Pests: Their Biology, Natural Enemies, and Control. World Crop Pests, Vol. 5. Elsevier, Amsterdam.

Boriss, H., H. Brunke and M. Kreith. 2006. Commodity profile: table grapes. Agricultural Issues Center, University of California. http://aic.ucdavis.edu/profiles/GrapesFreshMarket-2006.pdf [accessed 5 Oct 2011].

Botero-Garces, N. and R. Isaacs. 2003. Distribution of grape berry moth, *Endopiza viteana* (Lepidoptera: Tortricidae) in natural and cultivated habitats. Environmental Entomology. 32: 1187–1195.

Boyne, J. V., G. C. Rock and L. A. Nelson. 1985. Diapause in *Platynota idaeusalis* (Lepidoptera: Tortricidae): Effects of temperature, photoperiod, and time of inoculation in the field on diapause termination. Environmental Entomology. 14: 790–796.

Bradley, J. D. 1953. Some important species of the genus *Cryptophlebia* Walsingham, 1899, with descriptions of three new species (Lepidoptera: Olethreutidae). Bulletin of Entomological Research. 43: 679–689.

Bradley, J. D., W. G. Tremewan and A. Smith. 1973. British Tortricoid Moths - Cochylidae and Tortricidae: Tortricinae. The Ray Society, London, England. 251 pp.

Bradley, J. D., W. G. Tremewan and A. Smith. 1979. British Tortricoid Moths - Tortricidae: Olethreutinae. The Ray Society, London, England. 336 pp.

Brockerhoff, E. G. and M. Kenis. 1996. Parasitoids associated with *Cydia stobilella* (L.) (Lepidoptera: Tortricidae) in Europe, and considerations for their use for biological control in North America. Biological Control. 6: 202–214.

Brown. J. W. 1999. A new genus of tortricid moths (Tortricidae: Euliini) injurious to grapes and stone fruits in Chile. Journal of the Lepidopterists' Society. 53: 60–64.

Brown, J. W. 2005. World Catalogue of Insects. Volume 5, Tortricidae (Lepidoptera). Apollo Books, Stenstrup, Denmark. 741 pp.

Brown, J. W. 2006. Scientific names of pest species in Tortricidae (Lepidoptera) frequently cited erroneously in the entomological literature. American Entomologist. 52: 182–189.

Brown, J. W. and R. L. Brown. 2004. A new species of *Cryptaspasma* Walsingham (Lepidoptera: Tortricidae: Olethreutinae) from Central America, the Caribbean, and southeastern United States, with a catalogue of the world fauna of Microcorsini. Proceedings of the Entomological Society of Washington. 106: 288–297.

Brown, J. W. and Komai, F. 2008. Key to larvae of *Castanea*-feeding Olethreutinae frequently intercepted at U.S. ports-of-entry (Lepidoptera : Tortricidae). Tropical Lepidoptera Research. 18(1): 2–4.

Brown, J. W. and K. Nishida. 2003. First record of larval endophagy in Euliini (Tortricidae): a new species of *Seticosta* from Costa Rica. Journal of the Lepidopterists' Society. 57: 113–120.

Brown, J. W. and S. Passoa. 1998. Larval foodplants of Euliini (Lepidoptera: Tortricidae): from *Abies* to *Vitis*. Pan-Pacific Entomologist. 64: 1–11.

Brown, J. W., G. Robinson & J. A. Powell. 2008. Food plant database of the leafrollers of the world (Lepidoptera: Tortricidae) (Version 1.0.0). http://www.tortricidae.com/foodplants.asp.

Brown, J. W., M. E. Epstein, T. M. Gilligan, S. Passoa and J. A. Powell. 2010. Biology, identification, and history of the light brown apple moth, *Epiphyas postvittana* (Walker) (Lepidoptera: Tortricidae: Archipini) in California: an example of the importance of local faunal surveys to document the establishment of exotic insects. American Entomologist. 56(1): 34–43.

Brown, R. L. 1979. The valid generic and tribal names for the codling moth, *Cydia pomonella* (Olethreutinae: Tortricidae). Annals of the Entomological Society of America. 72: 565–567.

Brown, R. L. 1983. Taxonomic and morphological investigations of Olethreutinae: *Rhopobota*, *Griselda*, *Melissopus*, and *Cydia* (Lepidoptera: Tortricidae). Entomography 2: 97–120.

Carriere, Y., J. P. Deland and D. A. Roff. 1996. Obliquebanded leafroller (Lepidoptera: Tortricidae) resistance to insecticides: among-orchard variation and cross-resistance. Journal of Economic Entomology. 89: 577–582.

Cepeda, D. E. and G. E. Cubillos. 2011. Description of the last larval stage and a compilation of host records, of seven species of tortricids of economic importance in Chile (Lepidoptera: Tortricidae). Gayana. 75: 39–70.

Chapman, P. J. and S. E. Lienk. 1971. Tortricid fauna of apple in New York (Lepidoptera: Tortricidae); including an account of apple's occurrence in the state, especially as a naturalized plant. Spec. Publ. Geneva, NY: New York State Agricultural Experiment Station. 122 pp.

Chapman, P. J., S. E. Lienk and R. W. Dean. 1968. Bionomics of *Choristoneura rosaceana*. Annals of the Entomological Society of America. 61: 285–290.

Chen, M. H. and S. Dorn. 2009. Reliable and efficient discrimination of four internal fruit-feeding *Cydia* and *Grapholita* species (Lepidoptera: Tortricidae) by polymerase chain reaction-restriction fragment length polymorphism. Journal of Economic Entomology. 102: 2209–2216.

Clarke, J. F. G. 1962. A new tortricid genus from South America. Proceedings of the Biological Society of Washington. 75: 293–294.

Clarke, J. F. G. 1972. Two pests of beans from tropical America (Lepidoptera: Olethreutidae). Proceedings of the Entomological Society of Washington. 74: 467–471.

Cranshaw, W. 2010. Colorado insect of interest: Mexican jumping bean moth. http://www.colostate.edu/Dept/bspm/extension%20and%20outreach/Mexican%20jumping%20bean%20moth%2004November2010.pdf

Crop Protection Compendium. 2007 Edition. CAB International, Wallingford, UK, 2007.

Dang, P. T. and D. J. Parker. 1990. First records of *Enarmonia formosana* (Scopoli) in North America (Lepidoptera: Tortricidae). Journal of the Entomological Society of British Columbia. 87: 3–6.

Dang, P., R. Duncan and S. Fitzpatrick. 1996. Occurrence of two palearctic species of *Clepsis* Guenee, *C. spectrana* Trietschke and *C. consimilana* (Hübner) (Tortricidae), in British Columbia, Canada. Journal of the Lepidopterists' Society 50: 321–328.

Danthanarayana, W. 1975. The bionomics, distribution and host range of the light brown apple moth, *Epiphyas postvittana* (Walk.) (Tortricidae). Australian Journal of Zoology 23: 419–437.

Davis, E. E., S. French and R. C. Venette. 2005. Mini risk assessment: summer fruit tortrix moth, *Adoxophyes orana* (Fischer von Roslerstamm, 1834) (Lepidoptera: Tortricidae). USDA - CAPS Pest Risk Assessment.

Davis, E. E., R. C. Venette and E. M. Albrecht. 2006. Oak commodity based survey reference. USDA - CAPS Commodity Reference Guide.

DeLury N. C., G. J. R. Judd and M. G. T. Gardiner. 2006. Attraction of male *Pandemis limitata* (Lepidoptera: Tortricidae) to natural and synthetic pheromone sources: importance for assessing communication disruption. The Canadian Entomologist. 138: 697–711.

Diakonoff, A. 1939. The genera of Indo-Malayan and Papuan Torticidae. Zoologische Mededeelingen (Leiden). 21: 111–240.

Diakonoff, A. 1959. Revision of *Cryptaspasma* Walsingham 1900 (Lepidoptera, Tortricidae). Zoologische Verhandelingen (Leiden). 43: 1–60.

Diakonoff, A. 1967. Microlepidoptera of the Philippine Islands. U.S. National Museum Bulletin 257: 1–484.

Dickler, E. 1991. Tortricid pests of pome and stone fruits, Eurasian species, pp. 435–452. In L. P. S. van der Geest and H. H. Evenhius [eds.], Tortricid Pests: Their Biology, Natural Enemies, and Control. World Crop Pests, Vol. 5. Elsevier, Amsterdam.

Dombroskie, J. J. 2011. Aspects of archipine evolution (Lepidoptera: Tortricidae). University of Alberta, Department of Biological Sciences. Ph.D. dissertation. 488 pp.

Dugdale, J. S., D. Gleeson, L. H. Clunie and P. W. Holder. 2005. A diagnostic guide to Tortricidae encountered in field surveys and quarantine inspections in New Zealand: Morphological and molecular characters. National Plant Pest Reference Laboratory. 161 pp.

EPPO (European and Mediterranean Plant Protection Organization). 1997. Data sheets on quarantine pests: *Acleris variana* and *Acleris gloverana*. http://www.eppo.org/QUARANTINE/insects/Acleris_gloverana/ACLRSP_ds.pdf [accessed 7 March 2011].

EPPO (European and Mediterranean Plant Protection Organization). 2007. PQR, EPPO plant quarantine information retrieval system. Version 4.6 (2007/07) [accessed 28 Feb 2011].

Erichsen, C. and A. S. Schoeman. 1994. Moth pests of avocados. South African Avocado Growers' Association Yearbook. 17: 109–112.

Faber, B. A., J. G. Morse and M. S. Hoddle. 2010. UC IPM Pest Management Guidelines - Avocado. University of California IPM Online. http://www.ipm.ucdavis.edu/PMG/selectnewpest.avocado.html.

Falcon, L. A. and J. Huber. 1991. Biological control of the codling moth, pp. 355–369. In L. P. S. van der Geest and H. H. Evenhius [eds.], Tortricid Pests: Their Biology, Natural Enemies, and Control. World Crop Pests, Vol. 5. Elsevier, Amsterdam.

Folsom, J. W. 1909. Clover seed-caterpillar, pp. 133–137. In: The insect pests of clover and alfalfa. University of Illinois Agricultural Experiment Station Bulletin 134.

Freeman, T. N. 1944. A review of the North American species of the genus *Argyrotaenia* Stephens (Lepidoptera, Tortricidae). Scientific Agriculture. 25: 81–94.

Freeman, T. N. 1958. The Archipinae of North America (Lepidoptera: Tortricidae). The Canadian Entomologist Supplement 7 (Vol. 90): 1–89.

Fullaway, D. T. and N. L. H. Krauss. 1945. 193. *Amorbia emigratella* Busck, pp. 122–123. In Common Insects of Hawaii. Tongg Publishing Company, Honolulu. 228 pp.

Geest and H. H. Evenhius [eds.], Tortricid Pests: Their Biology, Natural Enemies, and Control. World Crop Pests, Vol. 5. Elsevier, Amsterdam.

Geier, P. and D. Briese. 1981. The light-brown apple moth, *Epiphyas postvittana* (Walker): a native leafroller fostered by European settlement., pp. 131–155. In R. Kitching and R. Jones [eds.], The Ecology of Pests. CSIRO, Melbourne, Australia.

Gilligan, T. M. and M. E. Epstein. 2009. LBAM ID: Tools for diagnosing light brown apple moth and related western U.S. leafrollers (Tortricidae: Archipini). CDROM. Center for Plant Health Science and Technology, USDA, APHIS PPQ, Raleigh, NC.

Gilligan, T. M., D. J. Wright and L. D. Gibson. 2008. Olethreutine moths of the midwestern United States, an identification guide. Ohio Biological Survey, Columbus, Ohio. 334 pp.

Gilligan, T. M. and M. E. Epstein. 2011. The European grape vine moth not found in California: *Eupoecilia ambiguella* (Hübner)., pp. 32–34. In Plant Pest Diagnostics Center Annual Report 2010. California Department of Agriculture, Sacaramento, CA.

Gilligan, T. M., J. W. Brown and M. S. Hoddle. 2011. A new avocado pest in Central America (Lepidoptera: Tortricidae) with a key to Lepidoptera larvae threatening avocados in California. Zootaxa. 3137: 31–45.

Gilligan, T. M., M. E. Epstein, S. C. Passoa, J. A. Powell, O. C. Sage and J. W. Brown. 2011b. Discovery of *Lobesia botrana* ([Denis & Schiffermuller]) in California: an invasive species new to North America (Lepidoptera: Tortricidae). Proceedings of the Entomological Society of Washington. 113(1): 14–30.

Glas, M. 1991. Tortricids in cereals, pp. 553–661. In: L. P. S. van der Geest, H. H. Evenhuis (eds.), Tortricid pests, their biology, natural enemies and control. Elsevier, Amsterdam, The Netherlands.

Glavendekic, M. 2006. *Epichoristodes acerbella* (Walker) (Lepidoptera: Tortricidae): a new species in the fauna of Serbia. Acta Entomologica Serbica. 11: 77–81.

Gomez, V. R. C. and J. V. Carrasco. 2008. Morphological characteristics of antennal sensilla in *Talponia batesi* (Lepidoptera: Tortricidae). Annals of the Entomological Society of America. 101: 181–188.

Gu, H. and W. Danthanarayana. 1992. Influence of larval rearing conditions on the body size and flight capacity of Epiphyas postvittana moths. Australian Journal of Zoology 40: 573–581.

Harbo, J. R. and K. J. Kraft. 1969. A study of *Phanerotoma toreutae*, a parasite of the pine cone moth *Laspeyresia toreuta*. Annals of the Entomological Society of America. 62: 214–220.

Harman, D. M. and C. W. Berisford. 1979. Host relationships and determination of larval instars of the locust twig borer *Ecdytolopha insiticiana*. Environmental Entomology. 8: 19–23.

Heinrich, C. 1923. Revision of the North American moths of the subfamily Eucosminae of the family Olethreutidae. U.S. National Museum Bulletin. 123: 1–298.

Heinrich, C. 1926. Revision of the North American moths of the subfamilies Laspeyresiinae and Olethreutinae. Bulletin of the U.S. National Museum. 132: 1–216.

Heinrich, C. 1932. A new species infesting *Annona* (Lepidoptera: Olethreutidae). Proceedings of the Entomological Society of Washington. 34: 20–23.

Heinrich, C. 1943. A new species of *Laspeyresia*, a bean pest from tropical America (Lepidoptera: Olethreutidae). Proceedings of the Entomological Society of Washington. 45: 71–73.

Hoddle, M. S. and C. D. Hoddle. 2008. Lepidoptera and associated parasitoids attacking Hass and non-Hass avocadoes in Guatemala. Journal of Economic Entomology. 101: 1310–1316.

Hoebeke, E. R., A. G. Wheeler, Jr. and J. W. Brown. 2008. *Archips xylosteana* (L.) (Lepidoptera: Tortricidae), a Palearctic leafroller new to North America. Proceedings of the Entomological Society of Washington. 110: 789–795.

Hoffman, M. P., L. M. McDonough and J. B. Bailey. 1983. Field test of the sex pheromone of *Amorbia cuneana* (Walsingham) (Lepidoptera: Tortricidae). Environmental Entomology. 12: 1387–1390.

Hogmire, H. W. and A. J. Howitt. 1979. The bionomics of the tufted apple budmoth, *Platynota idaeusalis* in Michigan. Annals of the Entomological Society of America. 72: 121–126.

Horak, M. 2006. Olethreutine moths of Australia (Lepidoptera: Tortricidae). Monographs on Australian Lepidoptera, Vol. 10. 522 pp.

Horak, M. and R. L. Brown. 1991. Taxonomy and phylogeny, pp. 23–50. In: L. P. S. van der Geest, H. H. Evenhuis (eds.), Tortricid Pests: Their Biology, Natural Enemies and Control. Elsevier, Amsterdam, The Netherlands.

IPNI. 2008. International Plant Names Index. Published on the Internet http://www.ipni.org.

Isaacs, R., K. S. Mason and E. Maxwell. 2005. Stage-specific control of grape berry moth, *Endopiza viteana* (Clemens) (Lepidoptera: Tortricidae), by selective and broad-spectrum insecticides. Journal of Economic Entomology. 98: 415–422.

Jeppson, L. R. 1989. Biology of citrus insects, mites, and mollusks., pp. 2–81. In Reuther, W., E. C. Calavan and G. E. Carman [eds.], Citrus Industry, Volume 5. Division of Agriculture and Natural Resources, University of California.

Jones, V. P. 1994. Feeding by *Cryptophlebia illepida* and *C. ombrodelta* (Lepidoptera: Tortricidae) on macadamia nut abortion. Journal of Economic Entomology. 87: 781–786.

Jones, V. P. 1995. Sampling plans for *Cryptophlebia* spp. (Lepidoptera: Tortricidae) attacking macadamia and litchi in Hawaii. Journal of Economic Entomology. 88: 1337–1342.

Kay, M. K. 1979. *Ctenopseustis obliquana* (Walker) (Lepidoptera: Tortricidae). Brownheaded leafroller. New Zealand Forest Service, Forest and Timber Insects in New Zealand. No. 40.

Komai, F. 1999. A taxonomic review of the genus *Grapholita* and allied genera (Lepidoptera: Tortricidae) in the Palaearctic region. Entomologica Scandinavica Supplement 55. 226 pp.

Komai, F. and K. Ishikawa. 1987. Infestation of chestnut fruits in China with two species of the genus *Cydia* (Lepidoptera, Tortricidae). Japanese Journal of Applied Entomology and Zoology. 31: 55–62. [in Japanese]

LaGasa, E. 1999. 1998 western Washington pheromone-trap delimiting survey for *Clepsis spectrana* (Treitschke), the straw-colored tortrix moth. 1998 Entomology Project Report, Laboratory Services Division, Washington State Department of Agriculture. 3 pp.

LaGasa, E. H., P. Hertzog, D. Bowden and B. Fair. 2003. *Archips podana* (Scopoli) (Lepidoptera: Tortricidae) host survey 2002 - Northwest Washington occurrence of an Old World tree-fruit pest new to the United States. Plant Protection Division, Washington State Department of Agriculture. WSDA PUB 080.

LaGasa, E. and S. Passoa. 2007. First report of the Palearctic species *Cydia coniferana* (Tortricidae) in the western United States. Journal of the Lepidopterists' Society. 61: 172–175.

Lambert, R. L. 1950. Revision of the moths in the subfamily Sparganothidinae (Lepidoptera, Tortricidae). Ph. D. thesis, Cornell University, Ithaca, NY. 502 pp.

Landry, B., J. A. Powell and F. A. H. Sperling. 1999. Systematics of the *Argyrotaenia franciscana* (Lepidoptera: Tortricidae) species group: evidence from mitochondrial DNA. Annals of the Entomological Society of America. 92: 40–46.

Landry, J-F., M. Roy and C. Turcotte. 2002. Cranberry pests of Quebec, an identification guide. Agriculture and Agri-Food Canada, Ottawa, Ontario. 117 pp.

Langhoff, P., A. Authier, T. R. Buckley, J. S. Dugdale, A. Rodrigo and R. D. Newcomb. 2009. DNA barcoding of the endemic New Zealand leafroller moth genera, *Ctenopseustis* and *Planotortrix*. Molecular Ecology Resources. 9: 691–698.

Lee, S. Y., H. Park, K. S. Boo, K.-T. Park and S. Cho. 2005. Molecular identification of *Adoxophyes honmai* (Yasuda) (Lepidoptera: Tortricidae) based on mitochondrial COI gene sequences. Molecules and cells. 19: 391–397.

MacKay, M. R. 1959. Larvae of the North American Olethreutidae (Lepidoptera). Canadian Entomologist Supplement 10: 1–338.

MacKay, M. R. 1962. Larvae of the North American Tortricinae (Lepidoptera: Tortricidae). The Canadian Entomologist Supplement 28: 1–182.

Maier, C. T. 2003. Distribution, hosts, abundance, and seasonal flight activity of the exotic leafroller, *Archips fuscocupreanus* Walsingham (Lepidoptera: Tortricidae), in the northeastern United States. Annals of the Entomological Society of America. 96: 660–666.

Maier, C. T. and V. C. Mastro. 1998. Discovery, abundance, and distribution of the exotic apple tortrix, *Archips fuscocupreanus* Walsingham (Lepidoptera: Tortricidae), in the northeastern United States. Proceedings of the Entomological Society of Washington. 100: 545–552.

Manrakhan, A., D. Abeeluck and A. Gokool. 2008. Assessment of damage by *Cryptophlebia peltastica* (Meyrick) (Lepidoptera: Tortricidae) in litchi orchards in Mauritius. African Entomology. 16: 203–208.

McDunnough, J. 1923. New Canadian Lepidoptera. The Canadian Entomologist. 55: 163–168.

Mecke, R. 2002. Insects of the Brazilian Pine. Attempto Service GmbH, Tubingen, Germany. 79 pp.

Meijerman, L. and S. A. Ulenberg. 2000. Arthropods of Economic Importance: Eurasian Tortricidae. Arthropods of Economic Importance series. ETI/ZMA.

Miller, W. E. 1982. *Grapholita delineana* (Walker), a Eurasian hemp moth, discovered in North America. Annals of the Entomological Society of America. 75: 184–186.

Miller, W. E. 1983. Nearctic *Endothenia* species: a new synonymy, a misidentification, and a revised status (Lepidoptera: Tortricidae). Great Lakes Entomologist. 16: 5–12.

Mutuura, A. 1980. Two *Pandemis* species introduced into British Columbia, with a comparison of native North American species (Lepidoptera: Tortricidae). The Canadian Entomologist. 112: 549–554.

Mutuura, A. 1982. *Cnephasia stephensiana*, a species newly recorded from Canada and compared with the previously recorded C. interjectana (Lepidoptera: Tortricidae). The Canadian Entomologist. 114: 667–671.

Mutuura, A. and E. Munroe. 1978. An old world leaf roller potentially injurious to Rosaceae, newly discovered in British Columbia, *Croesia holmiana* (Lepidoptera: Tortricidae). The Canadian Entomologist. 110: 641–646.

Nakai, M. 2009. Biological control of Tortricidae in tea fields in Japan using insect viruses and parasitoids. Virologica Sinica. 24: 323–332.

Namba, R. 1957. *Cryptophlebia illepida* (Butler) (Lepidoptera: Eucosmidae) and other insect pests of the macadamia nut in Hawaii. Proceedings of the Hawaiian Entomological Society. 16: 284–297.

Newcomer, E. J., and F. W. Carlson. 1952. The leaf roller moth *Pandemis pyrusana*. Journal of Economic Entomology. 45: 1079–1081.

Obraztsov, N. S. 1959. Characters separating *Archips rileyanus* and *cerasivoranus* as two species (Lepidoptera: Tortricidae). Entomological News. 70: 263–267.

Obraztsov, N. S. 1963. Some North American moths of the genus Acleris. Proceedings of the United States National Museum. 114: 213–270.

Opler, P. A. 1974. Biology, ecology, and host specificity of microlepidoptera associated with *Quercus agrifolia* (Fagaceae). University of California Press. vol. 75. 83 pp.

Otvos, I. S. 1991. North American species in forestry, pp. 719–756. In L. P. S. van der Geest and H. H. Evenhius [eds.], Tortricid Pests: Their Biology, Natural Enemies, and Control. World Crop Pests, Vol. 5. Elsevier, Amsterdam.

Ovsyannikova, E. I. and I. Ya. Grichanov. 2009. Pests: *Argyrotaenia ljungiana* (Thunberg) - grape tortrix. Interactive Agricultural Ecological Atlas of Russian and Neighboring Countries: Economic Plants and their Diseases, Pests and Weeds. http://www.agroatlas.ru/en/content/pests/Argyrotaenia_ljungiana/ [accessed 2 April 2011].

Packard, A. S. 1890. Insects injurious to forest and shade trees. USDA, Fifth Report of the U.S. Entomological Commission, Bulletin No. 7. 957 pp.

Pastrana, J. A. 1950. Una nueva peste en Araucaria de Misiones Republic Argentina "Lepidoptera - Grapholitidae." De la Revista de Invest Agricultura. 4: 243–244.

Payne, J. A. and E. K. Heaton. 1975. The hickory shuckworm: its biology, effect upon nut quality, and control. Annual Report of the Northern Nut Growers Association. 66: 19–25.

Passoa, S. 2008. Immature stages, pp. 297–314. In Gilligan, T. M., D. J. Wright and L. D. Gibson. Olethreutine moths of the midwestern United States, an identification guide. Ohio Biological Survey, Columbus, Ohio.

Pereyra P. C. and N. E. Sanchez. 1998. Effects of different host-plant species on growth, development and feeding of the bud borer, *Epinotia aporema* (Lepidoptera: Tortricidae) in La Plata, Argentina. Revista Chilena de Historia Natural. 71: 269–275.

Phillips-Rodriguez, E. and J. A. Powell. 2007. Phylogenetic relationships, systematics, and biology of the species of *Amorbia* Clemens (Lepidoptera: Tortricidae: Sparganothini). Zootaxa. 1670. 109 pp.

Pinhey, E. C. G. 1975. Moths of southern Africa: descriptions and colour illustrations of 1183 species. Tafelberg. 273 pp.

Powell, J. A. 1962. Taxonomic studies on the *Acleris gloverana-variana* complex, the black-headed budworms (Lepidoptera: Tortricidae). Canadian Entomologist. 94: 833–840.

Powell, J. A. 1964. Biological and taxonomic studies on tortricine moths, with reference to the species in California. University of California Publications in Entomology. Vol. 32. 317 pp.

Powell, J. A. 1983. Expanding geographical and ecological range of *Platynota stultana* in California (Lepidoptera: Tortricidae). Pan-Pacific Entomologist. 59: 233–239.

Powell, J. A. 1986. Synopsis of the classification of Neotropical Tortricidae, with descriptions of new genera and species (Lepidoptera: Tortricidae). Pan-Pacific Entomologist. 62: 372–398.

Powell, J. A. and P. A. Opler. 2009. Moths of western North America. University of California Press, Berkeley. 369 pp.

Prentice, R. M. 1966. Forest Lepidoptera of Canada recorded by the Forest Insect Survey. Vol. 4. Microlepidoptera. Publication 1142, Department of Forestry, Canada, Ottawa. 543–840.

Razowski, J. 1977. Monograph of the genus *Archips* Hübner (Lepidoptera, Tortricidae). Acta Zoologica Cracoviensia. 22: 55–206.

Razwoski, J. 1979. Revision of the genus *Clepsis* Guenee (Lepidoptera, Tortricidae). Part I. Acta Zoologica Cracoviensia. 23: 101–198.

Razwoski, J. 1979. Revision of the genus *Clepsis* Guenee (Lepidoptera, Tortricidae). Part II. Acta Zoologica Cracoviensia. 24: 113–152.

Razowski, J. 1987. The genera of Tortricidae (Lepidoptera). Part 1. Palearctic Chlidanotinae and Tortricinae. Acta Zoologica Cracoviensia. 30: 141–355.

Razowski, J. 2002. Tortricidae of Europe, Vol. 1, Tortricinae and Chlidanotinae. Frantisek Slamka, Slovakia. 247 pp.

Razowski, J. 2003. Tortricidae of Europe, Vol. 2, Olethreutinae. Frantisek Slamka, Slovakia. 301 pp.

Razowski, J. 2008. Tortricidae (Lepidoptera) from South Africa. 6: *Choristoneura* Hübner and *Procrica* Diakonoff. Polskie Pismo Entomologiczne. 77: 245–254.

Razowski, J. 2009. Tortricidae (Lepidoptera) of the Palaearctic Region, Vol. 2, Cochylini. Frantisek Slamka, Slovakia. 195 pp.

Razowski, J. and V. Pelz. 2010. Tortricidae from Chile (Lepidoptera: Tortricidae). SHILAP Revista de Lepidopterologia. 38: 5–55.

Roehrich, R. and E. Boller. 1991. Tortricids in vineyards, pp. 507–514. In L. P. S. van der Geest and H. H. Evenhius [eds.], Tortricid Pests: Their Biology, Natural Enemies, and Control. World Crop Pests, Vol. 5. Elsevier, Amsterdam.

Roelofs, W. L. and R. L. Brown. 1982. Pheromones and evolutionary relationships of Tortricidae. Annual Review of Ecology and Systematics. 13: 395–422.

Rothschild, G. H. L. and R. A. Vickers. 1991. Biology, ecology and control of the oriental fruit moth, pp. 389–412. In: L. P. S. van der Geest, H. H. Evenhuis (eds.), Tortricid pests, their biology, natural enemies and control. Elsevier, Amsterdam, The Netherlands.

Sakamaki, Y. and T. Hayakawa. 2004. Specific differences in larval and pupal characters of Japanese species of *Adoxophyes* (Lepidoptera, Tortricidae). Applied Entomology and Zoology. 39: 443–453.

Sandberg, S. and S. Passoa. 1989. New host records and morphological notes on four tortricines (Tortricidae). Journal of Research on the Lepidoptera. 27: 104–108.

Sheldon, W. G. 1925. *Peronea comariana* Zeller, and its variation. Entomologist. 58: 281–285.

Shirai, Y. and Y. Kosugi. 2000. Flight activity of the smaller tea tortrix, *Adoxophyes honmai* (Lepidoptera: Tortricidae). Applied Entomology and Zoology. 35: 459–466.

Solomon, J. D. 1995. Guide to insect borers in North American broadleaf trees and shrubs. Forest Service Agriculture Handbook AH-706, USDA, Washington, D. C. 735 pp.

Stansly, P. A. and W. Sanchez. 1990. Biology and oviposition behavior of *Cydia fabivora* (Lepidoptera: Tortricidae) in soybean on Ecuador's coastal plain. The Florida Entomologist. 73: 219–225.

Stevens, P., K. Froud and L. Jamieson. 2002. Effects of adult feeding on longevity and fecundity of *Ctenopseustis obliquana* (Lepidoptera: Tortricidae). New Zealand Journal of Crop and Horticultural Science. 30: 229–234.

Suckling D. M., J. G. I. Khoo and D. J. Rogers. 1990. Disruption of lightbrown apple moth *Epiphyas postvittana* (Lepidoptera: Tortricidae) trapping in Nelson, New Zealand. Australian Journal of Zoology 38: 363–373.

Suckling, D. M. and E. G. Brockerhoff. 2010. Invasion biology, ecology, and management of the light brown apple moth (Tortricidae). Annual Review of Entomology. 55: 285–306.

Summerland, S. A. and D. W. Hamilton. 1955. Biology of the red-banded leaf roller in southern Indiana. Journal of Economic Entomology. 48: 51–53.

Tamaki, Y. 1991. Tortricids in tea, pp. 541–551. In L. P. S. van der Geest and H. H. Evenhius [eds.], Tortricid Pests: Their Biology, Natural Enemies, and Control. World Crop Pests, Vol. 5. Elsevier, Amsterdam.

Tanigoshi, L. K. & P. Stary. 2003. Distribution, habitats and host plants of the cherry bark tortrix, *Enarmonia formosana* (Scopoli) in the Czech Republic (Lepidoptera, Tortricidae). Journal of Pest Science. 76: 41–43.

Thomas, W. P. 1979. Greenheaded leafroller, *Planotortrix excessana* (Walker). HortFACT. The Horticulture and Food Research Institute of New Zealand Ltd. http://www.hortnet.co.nz/publications/hortfacts/hf401026.htm [accessed 4 Oct 2011].

Timm, A. E., H. Geertsema and L. Warnich. 2006. Analysis of population genetic structure of two closely related tortricid species of economic importance on macadamias and litchis in South Africa. Agricultural and Forest Entomology. 8: 113–119.

Timm, A. E., L. Warnich and H. Geertsema. 2008. Morphological and molecular identification of economically important Tortricidae (Lepidoptera) on deciduous fruit tree crops in South Africa. African Entomology. 16: 209–219.

Tooman, L. K., C. J. Rose, C. Carraher, D. M. Suckling, S. R. Paquette, L. A. Ledezma, T. M. Gilligan, M. Epstein, N. B. Barr and R. D. Newcomb. 2011. Patterns of mitochondrial haplotypes diversity in the invasive pest *Epiphyas postvittana* (Lepidoptera: Tortricidae). Journal of Economic Entomology. 104(3): 920–932.

Turner, J. R. G. 1968. The ecological genetics of Acleris comariana (Zeller) (Lepidoptera: Tortricidae), a pest of strawberry. Journal of Animal Ecology. 37: 489–520.

USDA, NRCS. 2012. The PLANTS Database (http://plants.usda.gov, 7 January 2012). National Plant Data Team, Greensboro, NC 27401-4901 USA.

van der Geest, L. P. S., C. H. Wearing and J. S. Dugdale. 1991. Tortricids in miscellaneous crops, pp. 563–577. In: L. P. S. van der Geest, H. H. Evenhuis (eds.), Tortricid pests, their biology, natural enemies and control. Elsevier, Amsterdam, The Netherlands.

Van de Vrie, M. 1991. Tortricids in ornamental crops in greenhouses, pp. 515–539. In: L. P. S. van der Geest, H. H. Evenhuis (eds.), Tortricid pests, their biology, natural enemies and control. Elsevier, Amsterdam, The Netherlands.

Wagner, D. L. 2005. Caterpillars of eastern North America: A guide to identification and natural history. Princeton University Press, Princeton, New Jersey. 512 pp.

Wearing C. H., W. P. Thomas, J. W. Dugdale and W. Danthanarayana. 1991. Tortricid pests of pome and stonefruits, Australian and New Zealand species., pp. 453–472. In L. P. S. van der Geest and H. H. Evenhius [eds.], Tortricid Pests: Their Biology, Natural Enemies, and Control. World Crop Pests, Vol. 5. Elsevier, Amsterdam.

Wearing, C. H., J. D. Hansen, C. Whyte, C. E. Miller and J. Brown. 2001. The potential for spread of codling moth (Lepidoptera: Tortricidae) via commercial sweet cherry fruit: a critical review and risk assessment. Crop Protection. 20: 465–488.

Wehrle, L. P. 1924. The clover-seed caterpillar. Cornell University Agricultural Experiment Station Bulletin 428. 34 pp.

Weires, R. and H. Riedel. 1991. Other tortricids on pome and stone fruits, North American species, pp. 413–434. In: L. P. S. van der Geest, H. H. Evenhuis (eds.), Tortricid pests, their biology, natural enemies and control. Elsevier, Amsterdam, The Netherlands.

Wilde, G. and M. Semel. 1966. The life history of *Platynota flavedana*, a leaf roller of strawberry. Journal of Economic Entomology. 59: 1037–1041.

Williams, J. R. 1953. The larvae and pupae of some important Lepidoptera. Bulletin of Entomological Research. 43: 691–701.

Witzgall, P., M. Bengtsson and R. M. Trimble. 2000. Sex pheromone of grape berry moth (Lepidoptera: Tortricidae). Environmental Entomology. 29: 433–436.

Wyoski, M. and Y. Izhar. 1976. The carnation leaf-roller *Cacoecimorpha* (*Cacoecia*) *pronubana* Huebner (Lepidoptera, Tortricidae) on avocado trees in Israel. California Avocado Society Yearbook. 60: 92–95.

Yasuda, T. 1975. The Tortricinae and Sparganothinae of Japan (Lepidoptera: Tortricidae). Part II. Bulletin of the University of Osaka Prefecture, Series B. 27: 79–251.

Yasuda, T. 1998. The Japanese species of the genus *Adoxophyes* Meyrick (Lepidoptera, Tortricidae). Transactions of the Lepidopterological Society of Japan. 49: 159–173.

Zhou, J.-H., H.-G. Qui and W.-J. Fu. 1997. Summer fruit tortrix *Adoxophyes orana* should be classified as two subspecies (Lepidoptera: Tortricoidea: Tortricidae). Entomotaxonomia. 19: 130–135.

Zimmerman, E. C. 1978. Insects of Hawaii, Volume 9, Microlepidoptera, Part 1. University of Hawaii Press, Honolulu, Hawaii, 881 pp.

Chapter 3

The discovery of light brown apple moth, *Epiphyas posvittana* (Walker) in California (Lepidoptera: Tortricidae)*

Introduction

Epiphyas postvittana (Walker), the light brown apple moth (LBAM), is a polyphagous species that is an important pest of apple, citrus, and grapes in Australia and New Zealand (Bradley 1956). Because its introduction into North America could have a significant economic impact on U.S. agriculture, sporadic efforts over the past two or three decades have focused on its detection and exclusion from the United States. In 2006, a retired University of California professor collected two male LBAM at blacklight in an urban area of Berkeley, CA. This discovery triggered a flurry of activities that documented the occurrence of this species over a broad latitudinal range in coastal central California.

---

*Portions of this chapter are published in:

Gilligan, T. M. & M. E. Epstein. 2009. LBAM ID: Tools for diagnosing light brown apple moth and related western U.S. leafrollers (Tortricidae: Archipini). CD-ROM. Center for Plant Health Science and Technology, USDA/APHIS/PPQ, Raleigh, NC.

Brown, J. W., M. E. Epstein, T. M. Gilligan, S. Passoa & J. A. Powell. 2010. Biology, identification, and history of the light brown apple moth, *Epiphyas postvittana* (Walker) (Lepidoptera: Tortricidae: Archipini) in California: an example of the importance of local faunal surveys to document the establishment of exotic insects. American Entomologist. 56(1): 34-43.

This chapter summarizes the geographic distribution, hosts, life cycle, and morphology of LBAM to make this information available for persons involved in the exclusion, detection, and identification of this species. It also chronicles the discovery of LBAM in California and summarizes the events that followed. This is not an attempt to document pathways of LBAM entry into California, but rather presents a chronological overview of when specimens were first encountered in counties throughout the state.

Nomenclature

Epiphyas postvittana (Walker) was originally described in the genus *Teras* and subsequently was treated in *Tortrix*, *Archips*, *Cacoecia*, and *Eulia* by various authors. It was designated as the type species of *Austrotortrix* Bradley (1956), which was synonymized with *Epiphyas* Turner (1927) by Common (1961). Currently, *Epiphyas* includes 40 species, all described from Australia (Gilligan et al. 2012).

Geographic distribution

Epiphyas postvittana is indigenous to Australia. It occurs in all apple-growing areas in the southeastern part of that country, mainly along the coast and extending inland up to 200 miles. LBAM was introduced inadvertently into Tasmania and New Zealand (Bradley et al. 1973). It was first collected in the Hawaiian Islands in 1896 (Zimmerman 1978) and is now found at higher elevations on the islands, common only around Kula on Maui (D. Rubinoff, University of Hawaii, pers. comm.).

Meyrick (1937) recorded its discovery in the United Kingdom. Bradley et al. (1973) found that it was first established in Cornwall and subsequently spread throughout much of England. Bond (1998) first reported it from Ireland. An account of its history and distribution in the United

Kingdom was presented by Baker (1968). It has been reported intermittently elsewhere in Europe, including in the Netherlands (Wolschrijn and Kuchlein 2006) and southern Sweden (Svensson 2009). Although it was reported from New Caledonia (Bradley et al. 1973), its presence in that country could not be verified by Suckling and Brockerhoff (2010).

Larval hosts and damage

Epiphyas postvittana is a polyphagous pest on pome and stone fruits and other horticultural crops. It has been recorded from >500 plant species in 121 families and 363 genera (Suckling and Brockerhoff 2010), although larvae prefer herbaceous plants over woody ones. A partial host list compiled from Danthanarayana (1975), Wearing et al. (1991), Venette et al. (2003), and CAB International (2007) is presented in Table 109. Larvae feed on the leaves, buds, flowers, and fruits of its hosts, but most economic damage is caused by injury on the surface of fruits under webbed leaves, causing scarring as well as providing a site for infection and rot. When left uncontrolled, larval damage to fruit crops in Australia and New Zealand during years of high population densities reached 70% (with a range of 5–70%) on crops such as apple, citrus, and grape (Danthanarayana 1975, Buchanan 1977, Wearing et al. 1991).

Life cycle and biology

Epiphyas postvittana completes 2–4 generations annually over much of its range, depending on temperature and latitude. Populations in California appear to undergo at least four generations and adults are active throughout the year. The upper and lower temperature thresholds for development in laboratory studies are 7.5 and 31˚C; 20˚C is the optimum for development, leading to a life cycle of 25 days (Danthanarayana 1975).

Eggs are deposited on smooth surfaces of host plant foliage, including leaves, stems, and fruit (Danthanarayana 1975); and females often select the depression along the upper side midrib of leaves (Powell and Common 1985). Egg masses vary from 4 to 150 eggs. Under laboratory conditions, the largest mass usually is deposited first, with successively smaller numbers per oviposition bout over several days (Dumbleton 1932, Powell and Common 1985). In the field, females on average deposit fewer eggs per oviposition event; Geier and Briese (1980) tallied 23–26 per mass over four seasons, and Wearing et al. (1991) recorded a mean of 35 (range 4–96). Larvae pass through five or six instars; they can overwinter, but development during cooler months is slower.

Like most Archipini, early instars feed on the undersides of leaves within a silk chamber. Late instars may fold individual leaves, create a nest of several leaves webbed together, or web leaves to fruit and feed on the surface of the fruit. Caterpillars on deciduous trees and shrubs feed as long as leaves remain on the host plant and then drop to the ground, where they may feed on understory vegetation or survive in leaf litter. Pupation occurs in the larval nest, and metamorphosis takes about 10 days at 20˚C (Danthanarayana 1975).

Table 109: Partial list of *E. postvittana* host plants

Family	**Genus/species**	**Common name**
Actinidaceae	*Actinidia* Lindl.	kiwi
Aizoaceae	*Mesembryanthemum* L.	iceplant
Amaranthaceae	*Amaranthus* L.	pigweed
Anacardiaceae	*Mangifera indica* L.	mango
Apiaceae	*Daucus* L.	wild carrot
Apiaceae	*Petroselinum* J. Hill	parsley
Apiaceae	*Platysace* Bunge	
Apocynaceae	*Vinca* L.	periwinkle
Araliaceae	*Hedera* L.	ivy
Asteraceae	*Arctotheca calendula* (L.) Levyns	capeweed
Asteraceae	*Artemisia* L.	sagebrush
Asteraceae	*Aster* L.	aster

Table 109: Partial list of *E. postvittana* host plants

Family	**Genus/species**	**Common name**
Asteraceae	*Baccharis* L.	baccharis
Asteraceae	*Calendula* L.	marigold
Asteraceae	*Chrysanthemum* L.	daisy
Asteraceae	*Dahlia* Cav.	dahlia
Asteraceae	*Erigeron* L.	fleabane
Asteraceae	*Gerbera* J. F. Gmel.	Transvaal daisy
Asteraceae	*Helichrysum* Mill.	strawflower
Asteraceae	*Senecio* L.	ragwort
Asteraceae	*Tithonia* Desf. ex Juss.	tithonia
Betulaceae	*Alnus glutinosa* (L.) Gaertn.	European alder
Bignoniaceae	*Campsis* Lour.	trumpet-vine
Brassicaceae	*Brassica* L.	mustard
Brassicaceae	*Raphanus* L.	radish
Brassicaceae	*Sisymbrium* L.	hedgemustard
Buddlejaceae	*Buddleja* L.	butterflybush
Cannabaceae	*Humulus lupulus* L.	common hop
Caprifoliaceae	*Lonicera* L.	honeysuckle
Caprifoliaceae	*Viburnum* L.	viburnum
Celastraceae	*Euonymus* L.	spindletree
Chenopodiaceae	*Chenopodium* L.	goosefoot
Clusiaceae	*Hypericum perforatum* L.	common St. Johnswort
Cupressaceae	*Cupressus* L.	cypress
Ebenaceae	*Diospyros kaki* L. f.	Japanese persimmon
Ebenaceae	*Diospyros* L.	diospyros
Epacridaceae	*Monotoca* R. Br.	
Ericaceae	*Arbutus* L.	madrone
Ericaceae	*Vaccinium* L.	blueberry
Escalloniaceae	*Escallonia* Mutis ex L.f.	escallonia
Euphorbiaceae	*Breynia* J.R. Forst. & G. Forst.	breynia
Euphorbiaceae	*Phyllanthus* L.	leafflower
Fabaceae	*Acacia* Mill.	acacia
Fabaceae	*Cassia* L.	cassia
Fabaceae	*Cytisus scoparius* (L.) Link	Scotch broom
Fabaceae	*Genista* L.	broom
Fabaceae	*Hardenbergia* Benth.	
Fabaceae	*Lathyrus* L.	pea
Fabaceae	*Lupinus* L.	lupine
Fabaceae	*Medicago sativa* L.	alfalfa
Fabaceae	*Trifolium* L.	clover

Table 109: Partial list of *E. postvittana* host plants

Family	**Genus/species**	**Common name**
Fabaceae	*Ulex europaeus* L.	common gorse
Fabaceae	*Vicia faba* L.	horsebean
Fagaceae	*Quercus* L.	oak
Geraniaceae	*Pelargonium* L'Her. ex Aiton	geranium
Grossulariaceae	*Ribes* L.	currant
Hydrangeaceae	*Philadelphus* L.	mock orange
Iridaceae	*Crocosmia* Planch.	crocosmia
Juglandaceae	*Juglans* L.	walnut
Juncaginaceae	*Triglochin* L.	arrowgrass
Lamiaceae	*Lavandula* L.	lavender
Lamiaceae	*Mentha* L.	mint
Lamiaceae	*Salvia* L.	sage
Lauraceae	*Persea americana* Mill.	avocado
Loganaceae	*Gelsemium* Juss.	trumpetflower
Magnoliaceae	*Michelia* L.	michelia
Malvaceae	*Sida* L.	fanpetals
Myoporaceae	*Myoporum* Sol. ex G. Forst.	myoporum
Myrtaceae	*Acca sellowiana* (O. Berg.) Burret	feijoa
Myrtaceae	*Astartea* DC.	
Myrtaceae	*Callistemon* R. Br.	bottlebrush
Myrtaceae	*Eucalyptus* L'Her.	gum
Myrtaceae	*Leptospermum* J.R. Forst. & G. Forst.	teatree
Myrtaceae	*Melaleuca* L.	bottlebrush
Oleaceae	*Forsythia* Vahl	fosythia
Oleaceae	*Jasminum* L.	jasmine
Oleaceae	*Ligustrum* L.	privet
Oxalidaceae	*Oxalis* L.	woodsorrel
Pinaceae	*Pinus* L.	pine
Pittosporaceae	*Billardiera* Sm.	
Pittosporaceae	*Bursaria* Cav.	
Pittosporaceae	*Pittosporum* Banks ex Sol.	cheesewood
Plantaginaceae	*Plantago lanceolata* L.	narrowleaf plantain
Polygonaceae	*Polygala* L.	milkwort
Polygonaceae	*Polygonum* L.	knotweed
Polygonaceae	*Rumex* L.	dock
Proteaceae	*Grevillea* R. Br. ex Knight	grevillea
Proteaceae	*Leucadendron* L.	
Proteaceae	*Macadamia integrifolia* Maiden & Betche	macadamia nut
Proteaceae	*Persoonia* Sm.	

Table 109: Partial list of *E. postvittana* host plants

Family	**Genus/species**	**Common name**
Pteridaceae	*Adiantum* L.	maidenhair fern
Pteridaceae	*Pteris* L.	brake fern
Ranunculaceae	*Aquilegia* L.	columbine
Ranunculaceae	*Clematis* L.	leather flower
Ranunculaceae	*Ranunculus* L.	buttercup
Resedaceae	*Reseda* L.	mignonette
Rhamnaceae	*Ceanothus* L.	ceanothus
Rosaceae	*Cotoneaster* Medik.	cotoneaster
Rosaceae	*Crataegus* L.	hawthorn
Rosaceae	*Cydonia oblonga* Mill.	quince
Rosaceae	*Eriobotrya japonica* (Thunb.) Lindl.	loquat
Rosaceae	*Fragaria* L.	strawberry
Rosaceae	*Malus* Mill.	apple
Rosaceae	*Photinia* Lindl.	chokeberry
Rosaceae	*Prunus armeniaca* L.	apricot
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Pyracantha* M. Roem.	firethorn
Rosaceae	*Pyrus* L.	pear
Rosaceae	*Rosa* L.	rose
Rosaceae	*Rubus* L.	[various]
Rutaceae	*Boronia* Sm.	boronia
Rutaceae	*Choisya* Kunth	Mexican orange
Rutaceae	*Citrus* L.	citrus
Rutaceae	*Correa* Andrews	Australian fuschia
Rutaceae	*Eriostemon* Sm.	
Rutaceae	*Fortunella* Swingle	kumquat
Salicaceae	*Populus* L.	cottonwood
Salicaceae	*Salix* L.	willow
Sapindaceae	*Dodonaea* Mill.	dodonaea
Sapindaceae	*Litchi chinensis* Sonn.	lychee
Scrophulariaceae	*Hebe* Comm. ex Juss.	hebe
Smilacaceae	*Smilax* L.	greenbrier
Solanaceae	*Datura* L.	jimsonweed
Solanaceae	*Solanum lycopersicum* L. var. *lycopersicum*	garden tomato
Solanaceae	*Solanum tuberosum* L.	Irish potato
Theaceae	*Camellia japonica* L.	
Ulmaceae	*Trema* Lour.	trema
Urticaceae	*Urtica* L.	nettle
Valerianaceae	*Centranthus* Neck. ex Lam. & DC.	fox-brush

Table 109: Partial list of *E. postvittana* host plants

Family	**Genus/species**	**Common name**
Verbenaceae	*Clerodendron* Burm.	
Vitaceae	*Parthenocissus* Planch.	creeper
Vitaceae	*Vitis* L.	grape

Morphology

Egg. Eggs of *E. postvittana* (Fig. 61.1), like those of most Archipini and Sparganothini, are flat and broadly oval. The eggs are regularly overlapped, shingle-like, in rows or oval patches, without scaling or debris applied by the female; there is no colleterial secretion visible on the eggs or surrounding substrate (Powell and Common 1985, Wearing et al. 1991). When newly laid, the eggs are pale yellow to white and translucent; the embryos become visible as incubation proceeds. The chorion is reticulated, which separates eggs of this species from some, but not all, tortricids in North America (Peterson 1965).

Larva. Mature larvae (Figs. 61.5–6) range from 10–20 mm long and are generally yellowish green with paler subdorsal (SD), subventral (SV) and ventral lines. First instars are ~1.5 mm long with a dark head and light-colored body; succeeding instars (Figs. 61.3–4) have a darker body than fully grown larvae. The head, prothoracic shield, legs, and anal plate are pale brown, the genal dash is present or absent, and the prothoracic shield is only slightly darker than the rest of the integument. These structures lack contrasting darker markings that are characteristic of larvae in many archipine genera and other Tortricinae. All instars are darker dorsally, and the pinacula of later instars are slightly paler than the surrounding integument. Danthanarayana (1975) tallied head capsule (HC) widths for each instar.

Figure 61. *Epiphyas postvittana* immature stages; 1: egg mass; 2: pupa; 3–6: larvae.

Larval chaetotaxy of *E. postvittana* (Fig. 62.7) is typical of most Archipini with the small SD2 pinaculum fused to the anterior edge of the larger SD1 pinaculum on abdominal segments 1–7. LBAM larvae can be distinguished from other North American Archipini and Sparganothini studied by MacKay (1962) by the following combination of characters (Figs. 62.7–9): adfrontal area sharply pointed; distance between AF2 and P1 approximately equal to the distance between P1 and P2; a horizontal line connecting the AF2 setae midway between P1 and P2; SD pinacula usually rounded (not posteriorly elongated) and SV group unisetose on T2 and T3; tarsal coloration pale; spiracles of A1–7 smaller than the SD1 pinaculum but larger than the SD1 setal base; distance between the D1 setae greater than the distance separating D1 from SD1 laterally on the rounded or very slightly tapered anal shield; V1 setae on abdominal segment 9 no farther apart than the V1 on abdominal segments 7 and 8; and anal fork well developed with 7–9 straight pointed teeth, not minutely bifurcated at their tip. MacKay (1962) did not study

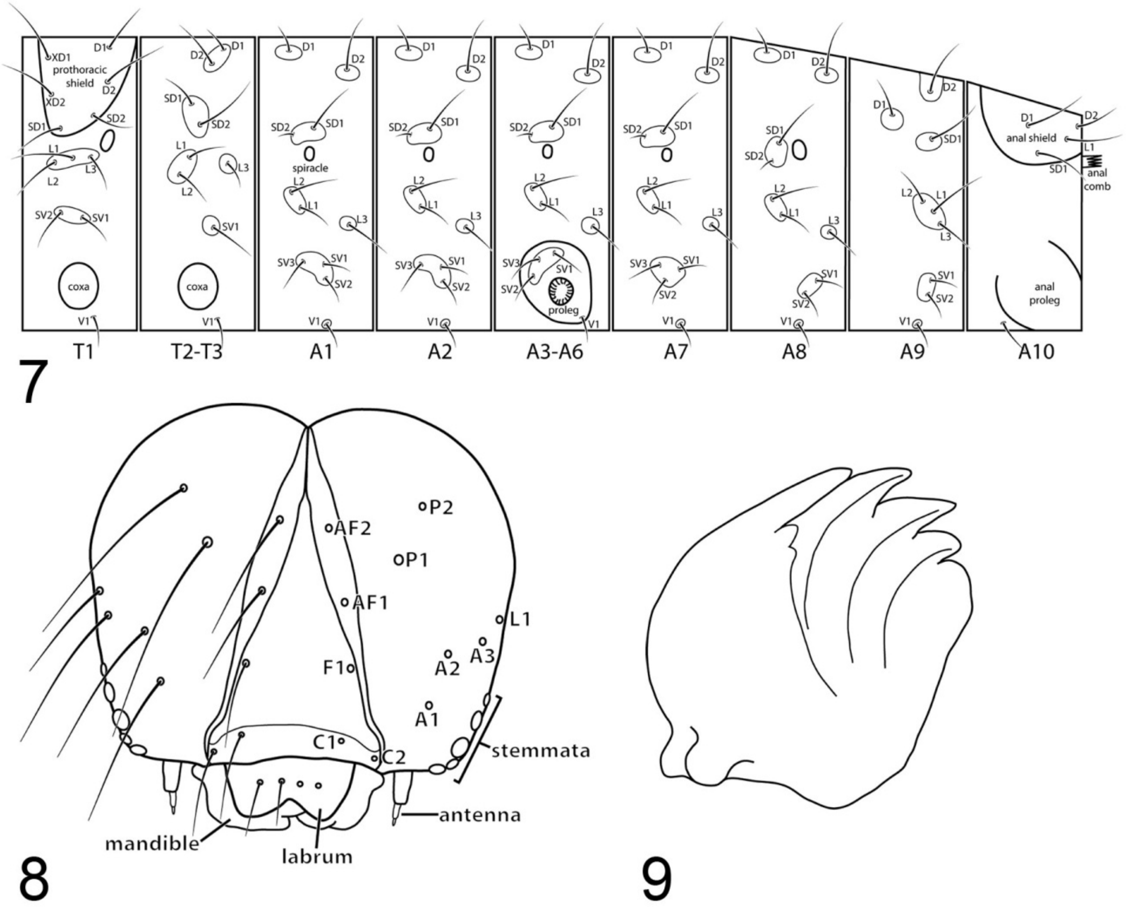


Figure 62. *Epiphyas postvittana* larval morphology; 7: setal map; 8: head; 9: mandible.

mandibles, but LBAM usually has two “inner teeth” (sensu Passoa 1985) on the mandible (Fig. 62.9) in contrast to many common species in California that lack inner teeth or have a very large retinaculum. However, in some specimens of LBAM, the mandible has only one inner tooth or both teeth are worn smooth, leaving only a thin ridge. Characters to separate LBAM from California species of Tortricidae were given by Gilligan and Epstein (2009, 2012).

Pupa. The pupa of *E. postvittana* (Fig. 61.2), like most tortricids, is greenish-brown initially and turns reddish-brown to dark brown when fully hardened. The average lengths of male and female pupae under laboratory conditions are 7.6 and 9.8 mm, respectively (Danthanarayana 1975).

Using Adler’s (1991) study of pupae of eastern U.S. apple-feeding Tortricidae, loss or reduction of the following characters is helpful for recognizing the pupa of *E. postvittana*: no vertex projections (present in many Tortricinae) and no dorsal abdominal pits (present in *Amorbia*, *Coelostathma*, and some introduced *Archips*). The vertex (“front” of Adler 1991) has two pairs of setae (some Archipini have only one pair), and the maxillae/labial palpi index is 2.4–2.7 in *E. postvittana*. The pupa of *E. postvittana* is most similar to that of *Pandemis,* but it differs in the relative length of the appendages; the prothoracic leg of *E. postvittana* extends past the prothoracic femur (“coxae” of Adler 1991) by more than half the height of the mesothoracic coxae. This distance is half the length of the mesothoracic coxae or less in *Pandemis.*

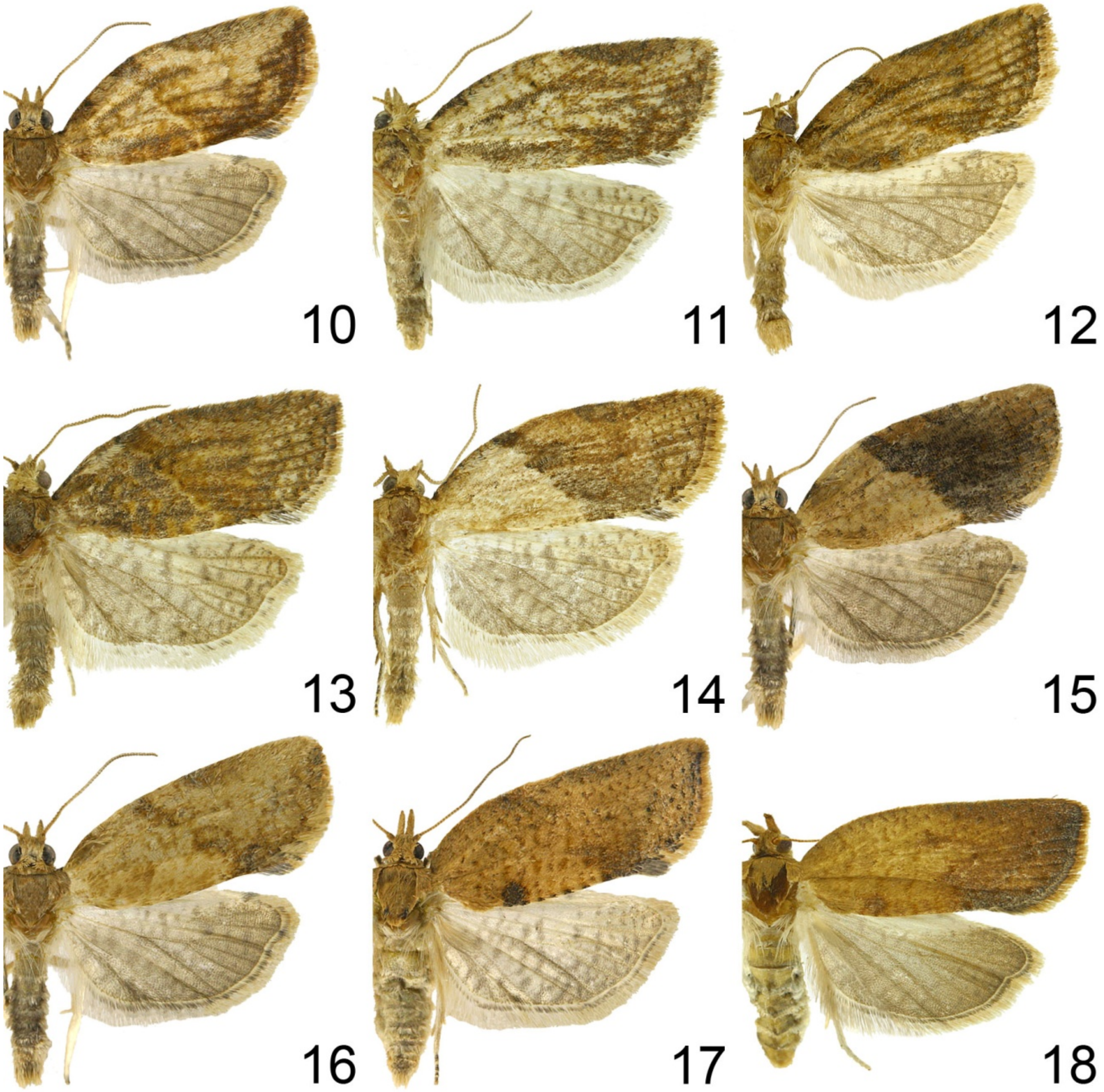


Figure 63. *Epiphyas postvittana* adults; 10–16: males; 17–18: females.

Adult. The forewing (FW) pattern of *E. postvittana* (Figs. 63.10–18) is sexually dimorphic and variable within each sex. The color varies from rust-brown to pale yellow with brown to dark brown markings. Males are more variable than females; although the basal half of the FW is slightly to markedly paler, the median fascia usually is well defined, and there is a dark mark on the costa distal to the median fascia. In California, the FW pattern includes a form with pale basal half and solid dark distal half (Figs. 63.14–15). The female FW color is more uniform, with a poorly defined median fascia and more speckled appearance than in males. Males have a FW costal fold, which is lacking in females, and this is an important diagnostic superficial feature in comparison with other tortricine moths in California. The fold extends from the base to ~0.3 the FW length and bears a flared scale fringe along its distal half. The strongly speckled underside, especially the hindwing, is diagnostic compared with that of *Argyrotaenia citrana* (Fernald) and other species that have similar, reduced FW pattern in the females. Most female *E. postvittana* have a dark mark on the dorsal margin of the forewing and two dark spots on the posterior of the thorax (Figs. 62.17–18). The hindwing is variably mottled with dark speckles, especially the underside, usually more evident in females (Passoa et al. 2008). Forewing length ranges from 5.3 to 11.1 mm in males and 5.4 to 12.5 mm in females (Danthanarayana 1975). Several *Epiphyas* species in Australia resemble *E. postvittana*, which makes identification of specimens from that continent difficult (Bradley 1956).

Male genitalia (Figs. 64.19–20) are distinctive, and examination of these structures is essential for reliable identification. Males have a combination of the following characters: spatulate (spoon-shaped) uncus; reduced socii; short valva with a broad sacculus; membranous lobe on the apex of the valve (the most diagnostic feature); and an aedeagus with 2–4 deciduous cornuti (Zimmerman 1978).

Female genitalia (Fig. 64.21) are typical of many Archipini, and females may be difficult to identify based on dissection alone. *E. postvittana* females have a combination of the following characters: simple sterigma; long, straight ductus bursae which is 2/3 or more the length of the abdomen; and corpus bursae with a single, hook-shaped signum (Gilligan and Epstein 2009).

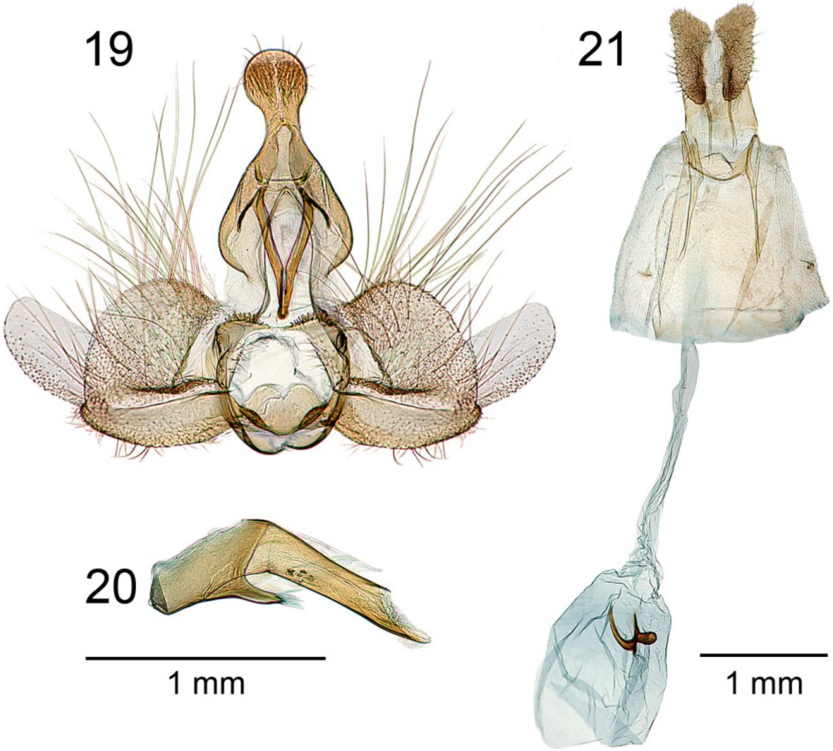


Figure 64. *Epiphyas postvittana* genitalia; 19: male, aedeagus removed; 20: aedeagus; 21: female.

Discovery of LBAM in California

The first specimen to be recognized as *Epiphyas* in California was attracted to blacklight at Berkeley (Yosemite Rd.), California, on July 19, 2006, and the second on November 19, 2006. This site had been surveyed for all moth species for the past 20 years by Jerry A. Powell. In 2006, the blacklight was monitored on approximately 140 dates through June, and 127 dates July–December (J. A. Powell, pers. comm.).

Marianne Horak of the Australian National Collection, Canberra (CSIRO) confirmed identification of the original specimens as *Epiphyas postvittana* in late January 2007, and that information was relayed to CDFA and USDA, who initiated pheromone trap surveys in early February. Captures at Richmond, approximately six miles from the Berkeley site, within 1 week confirmed that LBAM was established in the East Bay area, and captures in San Francisco soon thereafter revealed widespread establishment. Subsequently LBAM males were recorded at the Berkeley site on five dates in 2007 (July to September); on 15 dates in 2008 (21 specimens in April through mid-November); and in 2009, LBAM numbers increased dramatically, by 25× on >100 dates (January through August).

More than 257,000 individual specimens of adult LBAM were trapped using pheromone lures over the next 24 months (2007–2009) in 18 California counties. The following locations and dates are a chronological account and do not reflect colonization; they represent the continual discovery of the range of the species in Central California as more traps were deployed. The earliest LBAM collection dates were from Berkeley, Alameda Co. (February 27, 2007) and Richmond, Contra Costa Co. (March 6). Adult specimens of LBAM from pheromone traps were first received by the CDFA Plant Pest Identification Laboratory on March 9 (M. Epstein, pers. comm.). During the next 2 weeks, additional specimens were identified from Albany (Alameda Co.) and El Cerrito (Contra Costa Co.). The first specimens from San Francisco Co. were extracted from traps in Golden Gate Park on March 22, followed by samples from Sausalito and San Rafael (Marin Co.) on March 27.

On April 2, 2007 the first samples of *E. postvittana* were found in Palo Alto (Santa Clara Co.) and on April 13 in Belmont (San Mateo Co.). The first specimens from Santa Cruz Co. (Soquel) were discovered in trap samples on April 12, followed by many samples from the same general area, as well as Santa Cruz and Capitola during the remainder of the month. Also on April 12, the first samples were found in Monterey Co. (at Prunedale), followed shortly by captures in Pajaro, Royal Oaks, and Seaside.

The first sample from Napa Co. came from the city of Napa on May 9, 2007, followed by Solano Co. on June 27 from the city of Vallejo. A single LBAM was found in a trap in Los Angeles Co. (Sherman Oaks) on June 28. San Luis Obispo Co. was the last new county reported in 2007 with a sample collected in Cambria on September 25.

In 2008, LBAM was reported from three additional counties: Santa Barbara (Carpinteria) on January 15, Sonoma (Sonoma) on February 15, and San Benito (Aromas) on April 17. In 2009, LBAM was documented from Ventura Co. in February, Yolo Co. in April, and San Joaquin Co. in June. LBAM was rediscovered in Los Angeles and San Luis Obispo Counties in July, 2009, after going undetected in 2008.

Conclusions

The potential introduction of LBAM into the United States has been a concern to USDA for decades. Its presence would not only represent a new pest that could inflict considerable damage to ornamental plants and agricultural crops, but could result in quarantines that would adversely affect agricultural exports to trading partners. Hence, the discovery of LBAM in California in 2007 was met with swift and broad action to document its range and understand the scope of the problem.

The San Francisco Bay Area has been subject to fairly intensive survey efforts in the past, both focused LBAM pheromone trapping and general Lepidoptera inventories. Hence, if LBAM had been present, or at least widespread and/or abundant, in the region before about 2006, it probably would have been detected. Despite this, focused surveys using pheromone traps in 2007–2009 resulted in >257,000 individuals from 18 California counties: Alameda, Contra Costa, Los Angeles, Marin, Monterey, Napa, San Benito, San Francisco, San Joaquin, San Luis Obispo, San Mateo, Santa Barbara, Santa Clara, Santa Cruz, Solano, Sonoma, Ventura, and Yolo. Although the ultimate impact of LBAM in California is yet to be realized, this moth has not become established in the agriculturally important Central Valley; and armed with accurate information on its distribution, morphology, detection, and control, prospects for its management appear optimistic.

References

Adler, C. R. L. 1991. Identification of pupae on apple in eastern North America, pp. 51–64. *In* L. P. S. van der Geest and H. H. Evenhuis [Eds.]. Tortricid pests: their biology, natural enemies and control. Elsevier, Amsterdam.

Baker, C. R. B. 1968. Notes on *Epiphyas* (=*Austrotortrix*) *postvittana* (Walker) (Lep., Tortricidae). Entomologist's Gazette. 19: 167–172.

Bond, K. G. M. 1998. *Epiphyas postvittana* (Walker) (Lep., Tortricidae) new to Ireland. Entomologist's Record and Journal of Variation. 110: 250.

Bradley, J. D. 1956. A new genus for *Tortrix postvittana* Walker and certain other Australian and New Zealand species (Lepidoptera; Tortricidae). Bulletin of Entomological Research. 47: 104–105.

Bradley, J. D., W. G. Tremewan, and A. Smith. 1973. British Tortricoid moths. Cochylidae and Tortricidae: Tortricinae. Ray Society, London. 251 pp.

Buchanan, G. A. 1977. The seasonal abundance and control of light brown apple moth, *Epiphyas postvittana* (Walker) (Lepidoptera: Tortricidae), on grapevines in Victoria. Australian Journal of Agricultural Research. 28: 125–132.

CAB International. 2007. Crop protection compendium, 2007 Edition. CABI, Wallingford, UK.

Chapman, P. J., and S. E. Lienk. 1971. Tortricid fauna of apple in New York (Lepidoptera: Tortricidae); including an account of apple's occurrence in the state, especially as a naturalized plant. N.Y. State Agricultural Experiment Station, Special Publications, Geneva. 122 pp.

Common, I. F. B. 1961. The generic position of the Australian light-brown apple moth (Lepidoptera: Tortricidae). Proceedings of The Linnean Society of New South Wales. 86: 177–182.

Danthanarayana, W. 1975. The bionomics, distribution and host range of the light brown apple moth, *Epiphyas postvittana* (Walk.) (Tortricidae). Australian Journal of Zoology. 23: 419–437.

Dugdale, J. S., D. Gleeson, L. H. Clunie, and P. W. Holder. 2005. A diagnostic guide to Tortricidae encountered in field surveys and quarantine inspections in New Zealand: Morphological and molecular characters. National Plant Pest Reference Laboratory, Auckland, NZ.

Dumbleton, L. J. 1932. The apple leaf roller (*Tortrix postvittana* Walker). New Zealand Journal of Science and Technology. 14: 83–92.

Geier, P. W. and D. T. Briese 1980. The light-brown apple moth, *Epiphyas postvittana* (Walker): 4. Studies on population dynamics and injuriousness to apples in the Australian Capital Territory. Australian Journal of Ecology. 5: 63–93.

Gilligan, T. M. and M. E. Epstein. 2009. LBAM ID: Tools for diagnosing light brown apple moth and related western U.S. leafrollers (Tortricidae: Archipini). CD-ROM. Center for Plant Health Science and Technology, USDA/APHIS/PPQ, Raleigh, NC.

Gilligan, T. M. and M. E. Epstein. 2012. TortAI, Tortricids of Agricultural Importance to the United States (Lepidoptera: Tortricidae). CD-ROM. Identification Technology Program (ITP), USDA/APHIS/PPQ/CPHST, Fort Collins, CO.

Gilligan, T. M., J. Baixeras, J. W. Brown and K. R. Tuck. 2012. T@RTS: Online World Catalogue of the Tortricidae (Ver. 2.0). http://www.tortricid.net/catalogue.asp.

MacKay, M. R. 1962. Larvae of the North American Tortricinae (Lepidoptera: Tortricidae). Canadian Entomologist Supplement. 28: 1–182.

Meyrick, E. 1937. *Tortrix postvittana* Walk. (Microlepidoptera), a species new to Britain. Entomologist. 70: 256.

Passoa, S. 1985. Taxonomy of the larvae and pupae of economically important Pyralidae in Honduras. M.S. thesis, University of Florida, Gainesville.

Passoa, S., M. Epstein, T. M. Gilligan, J. Brambila and M. O'Donnell. 2008. Light brown apple moth (LBAM) *Epiphyas postvittana* (Walker) screening and identification aid, pp. 28–32. *In* 2008 light brown apple moth national survey guidelines. *Epiphyas postvittana* (Walk.) Tortricidae. USDA, APHIS, PPQ.

Peterson, A. 1965. Some eggs of moths among the Olethreutidae and Tortricidae (Lepidoptera). Florida Entomologist. 48:1–8.

Powell, J. A., and I. F. B. Common 1985. Oviposition patterns and egg characteristics of Australian tortricine moths (Lepidoptera: Tortricidae). Australian Journal of Zoology. 33: 179–216.

Suckling, D. M., and E. G. Brockerhoff. 2010. Invasion biology, ecology, and management of the light brown apple moth (Tortricidae). Annual Review of Entomology. 55:285–306.

Svensson, I. 2009. Anmärkningsvärda fynd av småfjärilar (Microlepidoptera) I Sverige 2008. Entomologisk Tidskrift. 130:61–72.

Turner, A. J. 1927. New and little-known Tasmanian Lepidoptera. Part II. Papers and Proceedings of the Royal Society of Tasmania. 1926: 119–164.

Venette, R. C., E. E. Davis, M. DaCosta, H. Heisler, and M. Larson. 2003. Mini risk assessment. Light brown apple moth, *Epiphyas postvittana* (Walker) [Lepidoptera: Tortricidae]. USDA/APHIS/PPQ Pest Risk Assessment. 38 pp.

Wearing, C. H., W. P. Thomas, J. W. Dugdale, and W. Danthanarayana. 1991. Tortricid pests of pome and stonefruits, Australian and New Zealand species., pp. 453–472. *In* L. P. S. van der Geest and H. H. Evenhius [Eds.]. Tortricid pests: their biology, natural enemies, and control; World Crop Pests, vol. 5. Elsevier, Amsterdam.

Wolschrijn, J. B., and J. H. Kuchlein. 2006. *Epiphyas postvittana,* nieuw voor Nederland en het Europese continent (Lepidoptera: Tortricidae). Tinea Nederland. 1(5–6): 37–39.

Zimmerman, E. C. 1978. Insects of Hawaii, vol. 9, Microlepidoptera, Part 1. University of Hawaii Press, Honolulu.

Chapter 4

Discovery of *Lobesia botrana* ([Denis & Schiffermüller]) in California: an invasive species new to North America (Lepidoptera: Tortricidae)*

Introduction

Lobesia botrana ([Denis and Schiffermüller]) is an economically important pest of vineyards throughout the Palearctic Region (e.g., Bovey 1966, Voigt 1972, Thiéry and Moreau 2005), but is particularly destructive in southern Europe and several Mediterranean countries (Roehrich and Boller 1991). The larvae cause damage to grapes by feeding directly on fruit, although the most serious economic losses are due to secondary infection of feeding sites by *Botrytis cinerea* Persoon (botrytis bunch rot or gray mold) (e.g., Fermaud and Le Menn 1992, Roehrich and Boller 1991). The threat that this moth poses to American agriculture has not gone unnoticed (Pierce 1918), and there have been sporadic efforts focused on its exclusion and/or detection in North America over the last few decades through a series of pest alerts, risk assessments, and surveys (Whittle 1985, Fowler and Larkin 2002, Venette et al. 2003). On September 30, 2009, tortricid larvae found in grape (*Vitis vinifera* L.: Vitaceae) from the Napa Valley of California were identified as *L. botrana*, representing the first North American records of this species. Because more than 800,000 acres of grapes are cultivated throughout the state, *L. botrana* could have a significant impact on California agriculture.

---

*This chapter is published as:

Gilligan, T. M., M. E. Epstein, S. C. Passoa, J. A. Powell, O. C. Sage & J. W. Brown. 2011. Discovery of *Lobesia botrana* ([Denis & Schiffermüller]) in California: an invasive species new to North America (Lepidoptera: Tortricidae). Proceedings of the Entomological Society of Washington. 113(1): 14-30.

The purpose of this chapter is to summarize the literature on the geographic distribution, hosts, life cycle, and morphology of *L. botrana* and to make this information available to those involved in exclusion, detection, and identification of this species. The discovery of *L. botrana* in California is also documented.

Materials and methods

Specimens of *L. botrana* from Europe, Chile, and California were examined and dissected using a Wild M5A stereomicroscope (Wild Heerbrugg AG, Switzerland). Dissection methodology follows Brown and Powell (1991) except that some preparations were not transferred to xylene and instead were mounted in Euparal (Bioquip Products, Rancho Dominguez, CA). Adults were photographed with a Canon EOS 40D digital SLR camera (Canon U.S.A., Lake Success, NY) mounted on a Visionary Digital BK Lab System (Visionary Digital, Palmyra, VA). Photographs of genitalia and larval cuticles were taken using a Nikon DXM 1200 digital camera mounted on a Nikon Labophot2 compound microscope (Nikon Instruments, Melville, NY). The photograph of the pupal abdominal segments and cremaster was taken with a Nikon DXM 1200 camera mounted on an upright Zeiss Axiomat microscope (Carl Zeiss Inc., Thornwood, NY). Scanning electron micrographs (SEMs) were prepared using a JEOL JSM-6500 microscope. Some photographs are a combination of several layers produced with Helicon Focus 4.80 software (Helicon Soft Ltd., Kharkov, Ukraine). All photographs were edited using Adobe Photoshop CS3 Extended (Adobe Systems Inc., San Jose, CA). Morphological terminology and wing pattern descriptions follow Gilligan et al. (2008). Natural history and host information is compiled from Ben-Yehuda et al. (1993), Bradley et al. (1979), CPC (2007), Thiéry and Moreau (2005), and Venette et al. (2003).

Authorship of the species name *botrana* is unequivocally attributed to Denis & Schiffermüller even though their names do not appear on the original work. Hence, per

Recommendation 51D of the International Code of Zoological Nomenclature (International Commission on Zoological Nomenclature 1999), the authors' names …"should be enclosed in square brackets to show the original anonymity." Unfortunately, this convention frequently is ignored.

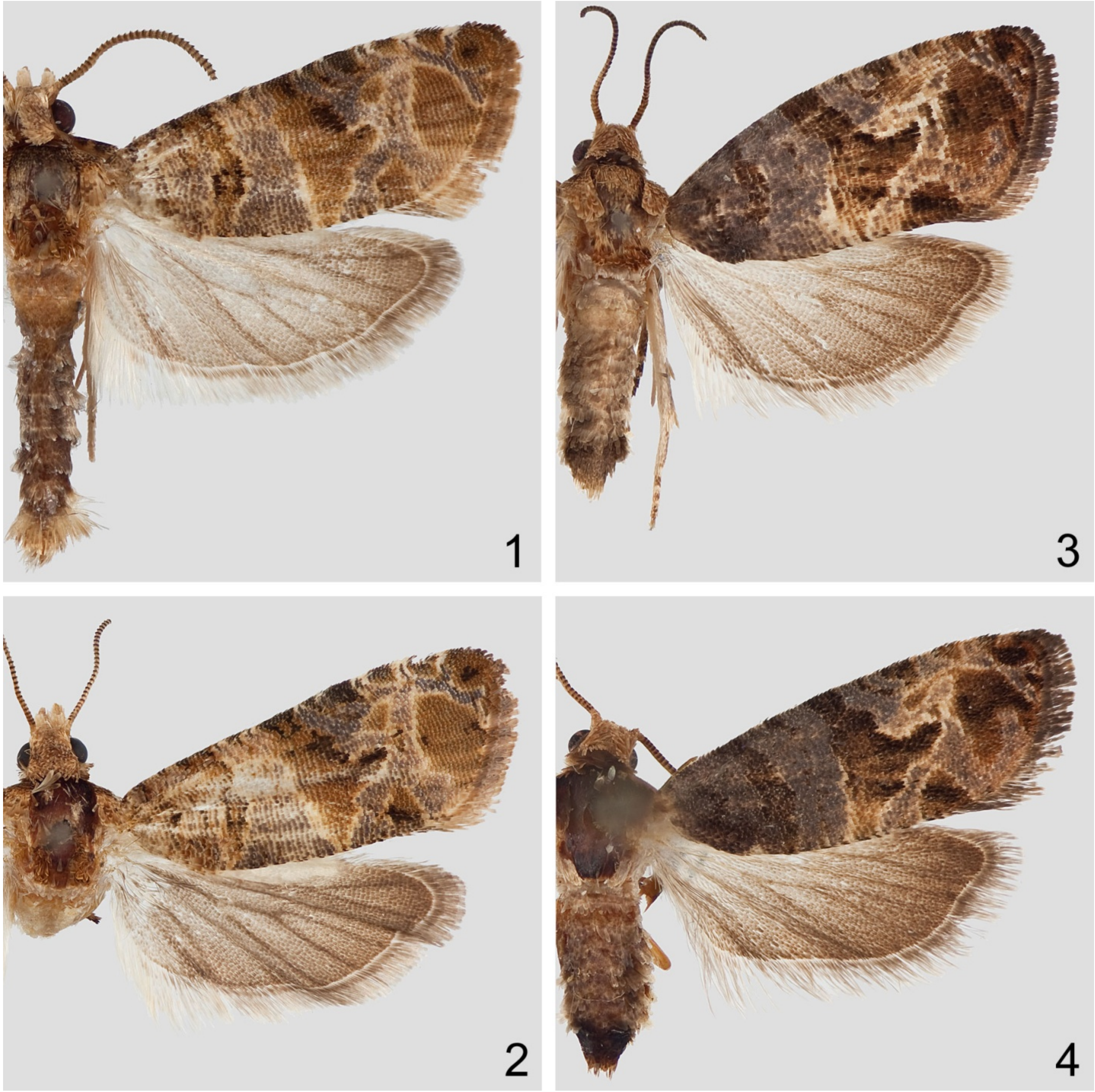


Figure 65. Adult wing patterns; 1–2: *Lobesia botrana*; 3–4: *Paralobesia viteana*.

Results and discussion

Nomenclature

Tortrix botrana [Denis and Schiffermüller] was described from Vienna, Austria. Ragonot (1894) proposed the genus *Polychrosis* with *T. botrana* as the type species. Over half a century later, Obraztsov (1953) relegated *Polychrosis* to a subgenus of *Lobesia* Guenée, with *L. botrana* as its only member. Synonyms of *L. botrana* include *Phalaena vitisana* Jacquin and *Olindia rosmarinana* Millière (Brown 2005). Dufrane (1960) described "flavosquamella" as a form of *L. botrana* from France. Older literature often used the combination *Eudemis botrana* (González 2008).

Lobesia botrana has a wide variety of common names in several languages (see list in González 2008). English common names for *L. botrana* include: European grape vine moth (EGVM), European grape berry moth, grape berry moth, grapevine moth, vine moth, European vine moth, grape leaf roller, and grape fruit moth (CPC 2007, Meijerman and Ulenberg 2000); several of these names are shared with with *Eupoecilia ambiguella* (Hübner), another European tortricid grape pest. When common names are necessary, "European grape vine moth" is recommended for *L. botrana* and "European grape berry moth" for *E. ambiguella*.

Geographic distribution

Lobesia botrana is native to the Palearctic Region and is widely distributed in Western Europe, Central Asia, and northern Africa (CPC 2007, Razowski 2003). Records from Japan (Bae and Komai 1991) and eastern Africa (Kenya, Ethiopia, and Eritrea) are likely due to inadvertent human introductions (CPC 2007).

North American records of *L. botrana* from the mid- to late-1800s are misidentifications of *Paralobesia viteana* (Clemens) (Kearfott 1904), a native North American grape-feeding tortricid that is extremely similar morphologically to *L. botrana*.

Lobesia botrana was first discovered in the Americas from Chile in April 2008, where it was reported feeding on grape by the Chilean Department of Agriculture (González 2008). Subsequently, it spread to all grape growing regions of that country (SAG 2010), and in March 2010, the Argentinean National Service for Agrifood Health and Quality reported *L. botrana* in Argentina at two locations in the Maipú Department, Mendoza Province, close to the Chilean border (SENASA 2010).

Larval hosts and damage

Lobesia botrana is one of the most important pests of grape (*Vitis vinifera*) in the Palearctic (Bovey 1966, Roehrich and Boller 1991). Larvae are polyphagous and have been recorded from over 40 species of plants in the following families: Actinidiaceae, Araliaceae, Asteraceae, Berberidaceae, Caprifoliaceae, Caryophyllaceae, Cornaceae, Ebenaceae, Ericaceae, Grossulariaceae, Lamiaceae, Liliaceae, Menispermaceae, Oleaceae, Punicaceae, Ranunculaceae, Rhamnaceae, Rosaceae, Thymeleaceae, and Vitaceae. A list of host plants is presented in Table 110 (summarized from Brown et al. 2008).

Table 110: Documented host plants for *L. botrana*

Family	**Genus/species**	**Common name**
Actinidiaceae	*Actinidia chinensis* Planch.	kiwi
Araliaceae	*Hedera helix* L.	English ivy
Asteraceae	*Tanacetum vulgare* L.	common tansy
Berberidaceae	*Berberis vulgaris* L.	common barberry
Caprifoliaceae	*Lonicera tatarica* L.	Tatarian honeysuckle
Caprifoliaceae	*Viburnum lantana* L.	wayfaringtree
Caryophyllaceae	*Dianthus* L.	carnation
Cornaceae	*Cornus mas* L.	Cornelian cherry
Cornaceae	*Cornus sanguinea* L.	bloodtwig dogwood

Table 110: Documented host plants for *L. botrana*

Family	**Genus/species**	**Common name**
Cornaceae	*Cornus* L.	dogwood
Ebenaceae	*Diospyros kaki* L. f.	Japanese persimmon
Ebenaceae	*Diospyros virginiana* L.	common persimmon
Ericaceae	*Arbutus unedo* L.	strawberry tree
Grossulariaceae	*Ribes nigrum* L.	European black currant
Grossulariaceae	*Ribes rubrum* L.	cultivated currant
Grossulariaceae	*Ribes uva-crispa* L.	European gooseberry
Lamiaceae	*Rosmarinus officinalis* L.	rosemary
Liliaceae	*Urginea maritima* (L.) Baker	red squill
Menispermaceae	*Menispermum canadense* L.	common moonseed
Oleaceae	*Ligustrum vulgare* L.	European privet
Oleaceae	*Olea europaea* L.	olive
Oleaceae	*Syringa vulgaris* L.	common lilac
Punicaceae	*Punica granatum* L.	pomegranate
Ranunculaceae	*Clematis vitalba* L.	evergreen clematis
Rhamnaceae	*Ziziphus jujuba* (L.) Karst.	common jujube
Rosaceae	*Malus pumila* Mill.	apple
Rosaceae	*Prunus avium* (L.) L.	sweet cherry
Rosaceae	*Prunus domestica* L.	European plum
Rosaceae	*Prunus dulcis* (Mill.) D.A. Webb	sweet almond
Rosaceae	*Prunus persica* (L.) Batsch var. *nucipersica* (Suckow) C.K. Schneid.	nectarine
Rosaceae	*Prunus salicina* Lindl.	Japanese plum
Rosaceae	*Prunus spinosa* L.	blackthorn
Rosaceae	*Pyru communis* L.	common pear
Rosaceae	*Rubus caesius* L.	European dewberry
Rosaceae	*Rubus fruticosus* L.	shrubby blackberry
Rosaceae	*Rubus* L.	raspberry
Thymeleaceae	*Daphne gnidium* L.	flax-leaved daphne
Thymeleaceae	*Thymelaea hirsuta* (L.) Endl.	thymelaea
Vitaceae	*Parthenocissus quinquefolia* (L.) Planch.	Virginia creeper
Vitaceae	*Vitis vinifera* L.	wine grape

Preference for *V. vinifera* may have evolved only recently as damage to grape by *L. botrana* was not reported until the early part of the 20th century (Marchal 1912, Thiéry and Moreau 2005). Female *L. botrana* are attracted to volatiles of both *Vitis* and *Daphne* (Tasin 2005), but when given a choice, prefer to oviposit on *Daphne* rather than *Vitis* (Maher and Thiéry 2006). As a result, several authors (e.g., Bovey 1966, Maher and Thiéry 2006, Tasin 2005, Thiéry and Moreau 2005) have hypothesized that *Daphne gnidium* L. (Thymelaeaceae) is the native host plant for *L. botrana*. Polyphagy may persist because hosts other than *Vitis* provide better nutritional value. Thiéry and Moreau (2005) demonstrated that individuals feeding on alternate hosts had lower larval mortality, shorter development times, higher pupal weight, and increased female fecundity and mating success.

Larvae of *L. botrana* damage grape by feeding on flowers, buds, and fruit. First generation larvae feed on flower buds, resulting in reduced yields, whereas second and third generation larvae feed on ripening and mature grapes, respectively (Roehrich and Boller 1991). The most significant economic losses are due not to direct damage to berries, but to secondary infection of feeding sites by *B. cinerea* (e.g., Fermaud and Le Menn 1992, Roehrich and Boller 1991). Economic thresholds for *L. botrana* in grape vary with climatic conditions, type of grape (wine or table), and cultivar (Roehrich and Boller 1991). The species also is considered a minor pest of some ornamental and greenhouse plants, and has been reported infesting pear (*Pyrus communis* L.; Rosaceae) orchards in Israel (Ben-Yehuda et al. 1993).

Life cycle and biology

Lobesia botrana completes 2–3 generations annually in southern Europe, although the number of generations varies from one in northern Europe to as many as five in Central Asia (CPC 2007, Filip 1986). There are at least three and possibly four generations in Chile (González 2008). The lower temperature threshold for egg, larval, and pupal development is approximately 8°C (Gabel and Mocko 1984).

The following summary of the life cycle on grape is derived from Bovey (1966), Bradley et al. (1979), CPC (2007), Masante-Roca et al. (2007), and Meijerman and Ulenberg (2000). Females of the first generation deposit eggs singly or in groups of two or three on buds, pedicels, and flowers; those of the second and third generations deposit single eggs on individual grape berries. Eggs hatch in approximately 5–10 days or 75 degree-days above a

10°C threshold. Larvae complete five instars, with the first generation feeding on flowers and buds, the second generation feeding within a single unripened grape berry, and the third and subsequent generations feeding on ripened berries. Larval development is completed in approximately 20–28 days or 170 degree-days for larvae feeding on flowers and 225 degree-days for larvae feeding on berries. Non-diapausing individuals of the first and second generations pupate in rolled leaves or inflorescences tied with silk. Pupae complete development in approximately 12–14 days, or 130 degree-days, for non-diapausing individuals. Diapausing individuals of the third or subsequent generations pupate under bark, in the soil, or under leaf litter; adults emerge the following spring.

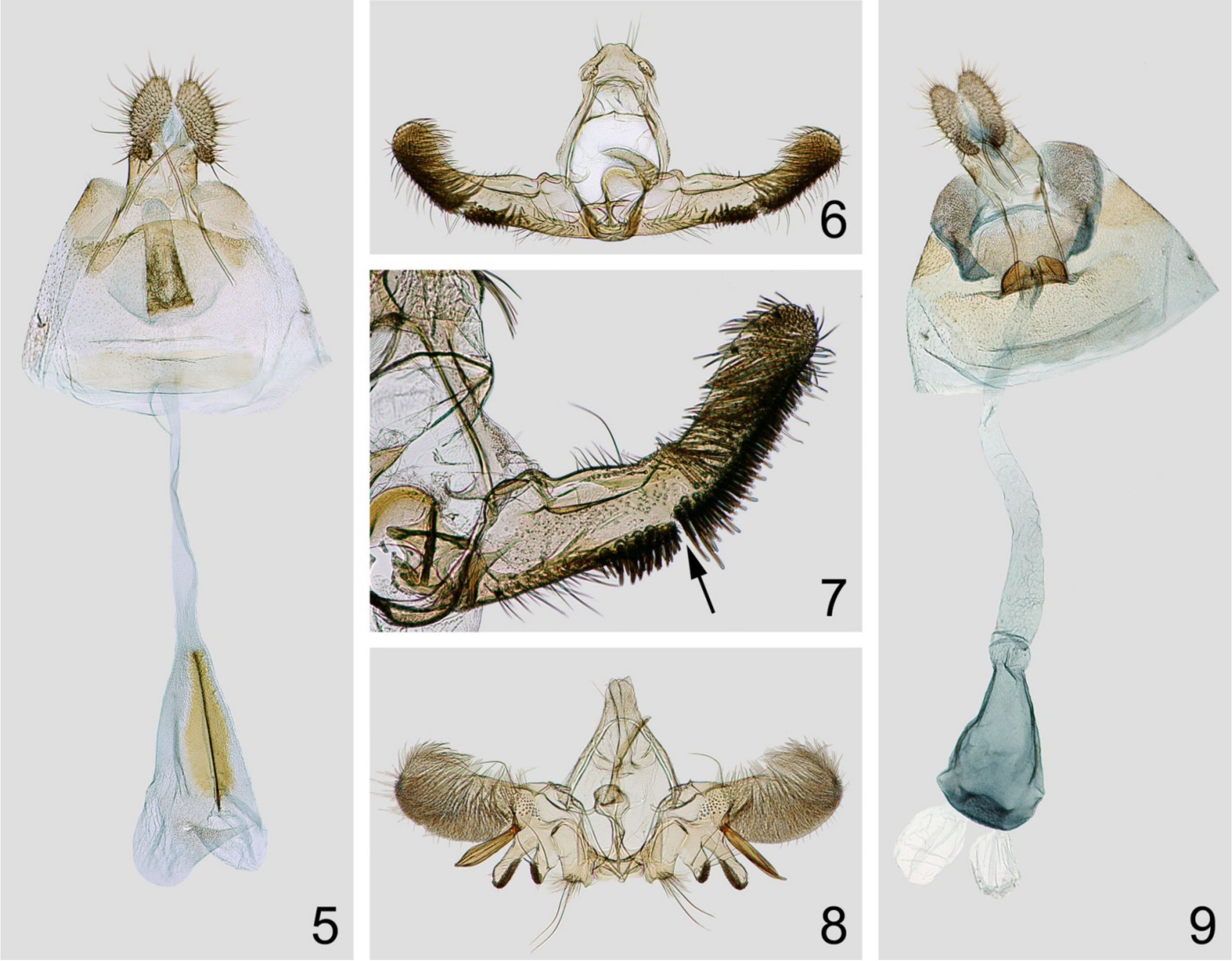


Figure 66. Male and female genitalia; 5: *L. botrana* female; 6: *L. botrana* male; 7: *L. botrana* male, arrow denotes gap in spines on valva; 8: *P. viteana* male; 9: *P. viteana* female.

Morphology

Egg.— The egg of *L. botrana* (Figs. 68.20–22), originally illustrated by Sylvestri (1912), is elliptical, flattened and slightly convex. Each egg is approximately 0.65–0.90 mm long by 0.45–0.75 mm wide (CPC 2007). The chorion is either smooth or there is a slight polygonal reticulation on the border and around the micropile (CPC 2007). When newly laid, eggs are translucent pale yellow, eventually turning “opalescent” gray (Meijerman and Ulenberg 2000), with the embryo becoming visible during incubation.

No morphological characters are available to separate eggs of *L. botrana* from related species in the Olethreutinae. However, many species of Tortricinae (i.e., Archipini and Sparganothini) oviposit in large overlapping clusters (Horak 1991). This oviposition pattern characterizes several common Californian tortricid pests such as *Platynota stultana* Walsingham*, Argyrotaenia franciscana* (Walsingham)*, and Epiphyas postvittana* (Walker). Eggs of *L. botrana* are laid singly or in small clumps, never in a mass.

Larva.— The first instar larva is yellowish green and approximately 1.0 mm in length. The head is black to dark brown and the paler prothoracic shield is concolorous with the rest of the body. The mature larva (Figs. 67.10–13) is 10–15 mm long and varies in color from light yellowish green to pale brown. The head is brown to light yellowish brown to honey colored, the antennae and thoracic legs are brown to black, and the prothoracic shield is variably shaded with dark brown to black on the posterior and lateral margins. All instars have a dark stemmatal area and genal dash. Prepupae are colored a "dull dark purple" (H. Nadel, pers. comm.). Irigaray et al. (2006) measured head capsule widths for each instar.

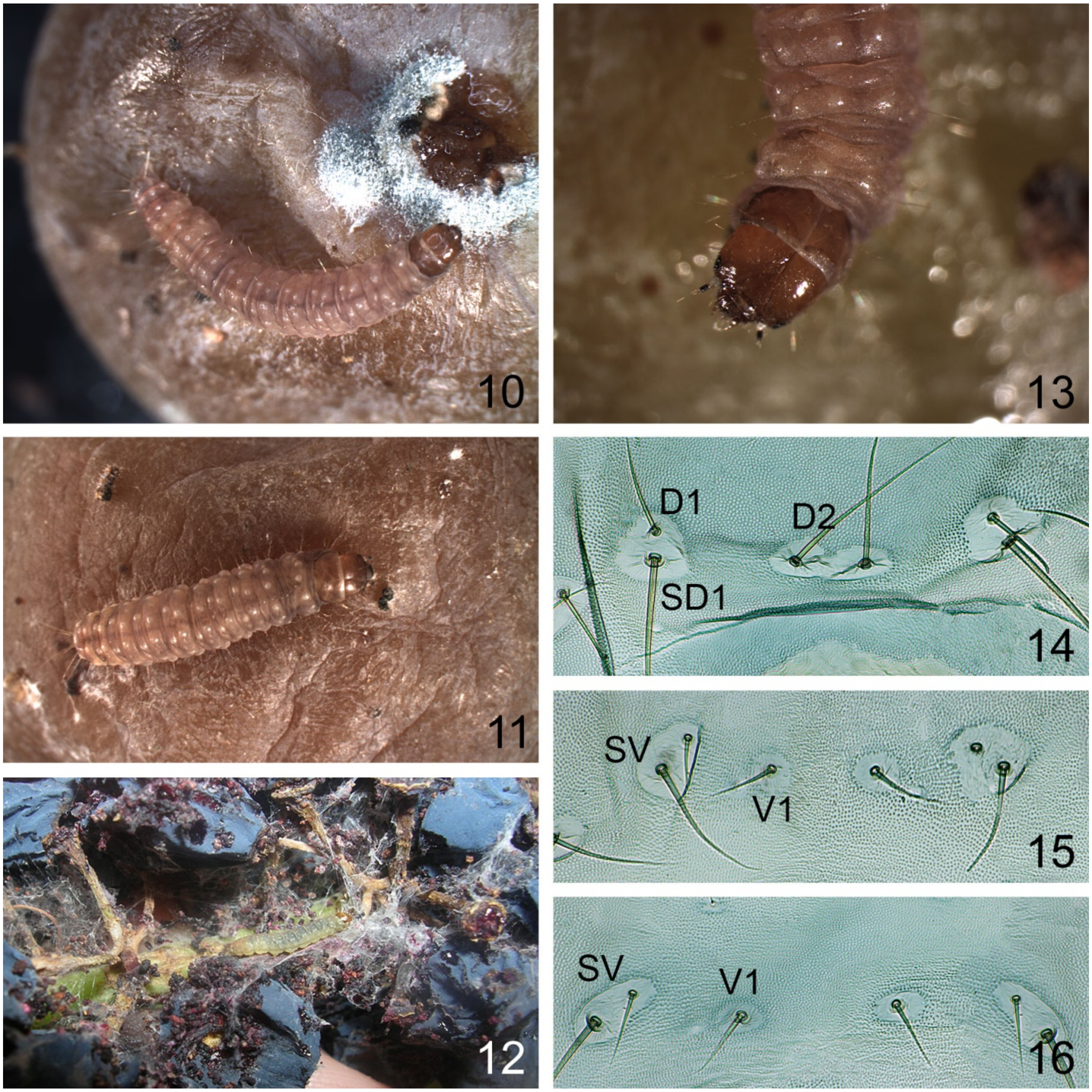


Figure 67. *Lobesia botrana* larvae; 10: larva feeding on *Botrytis cinerea*; 11: larva on grape; 12: typical damage to grapes (courtesy Roberto H. González, Universidad de Chile); 13: details of head and prothoracic shield; 14: dorsal aspect of segment A9 showing D2s on “saddle” pinaculum, and D1 and SD1 on same pinaculum; 15: ventral aspect of segment A8 showing relative spacing of V1 setae; 16: ventral aspect of segment A9 showing relative spacing of V1 setae.

Important structural features of the *L. botrana* larva (Figs. 69.23–25, 70.26–29) include: mandibles without inner teeth (sensu Passoa 1985) or a retinaculum; distance between P1 and AF2 on head equal to distance between P1 and P2; a horizontal line connecting the P2 setae on head passes through AF2; L pinaculum on T1 horizontal, not extending beneath spiracle; SV groups on A1, 2, 7, 8, 9 with 3:3:3:2:2 setae; SD2 on A1–8 absent; distance between V setae on A9 approximately 1.5–2x the distance between V setae on A8 (Figs. 67.15–16); distance between D1 setae on anal shield equal to the distance between D1 and SD1; anal comb with 5–6 teeth in California individuals (Fig. 70.29), other authors (e.g. Swatschek 1958) report 6–8 teeth; and body spicules relatively dense (at 200x or more).

Lobesia botrana belongs to the subfamily Olethreutinae, whose larvae can usually be recognized by a combination of three characters: L group on the prothorax trisetose, D2 setae of A9 closely spaced or joined on “saddle” pinaculum, and A9 with the D1 and SD1 setae on a single pinaculum (Fig. 67.14). Other common tortricid pests in California (*P. stultana*, *A. franciscana*, and *E. postvittana*) belong to the subfamily Tortricinae, whose larvae have D1 and SD2 of A9 on separate pinacula (see Gilligan and Epstein 2009).

The key in MacKay (1959), or the simplified version in Passoa (2008), can be used to separate *L. botrana* larvae from many other Olethreutinae in the United States. Because the A and L setae of the head are not in a straight line, all thoracic legs are concolorous, and the SV group on A7 is trisetose, *L. botrana* will group with Nearctic species of *Endothenia*, *Lobesia*, and *Paralobesia* using either of these keys. The larva of *L. botrana* also lacks a dark contrasting patch on the prothoracic shield (although a thin marginal line may be present) and extra setae on the abdominal segments.

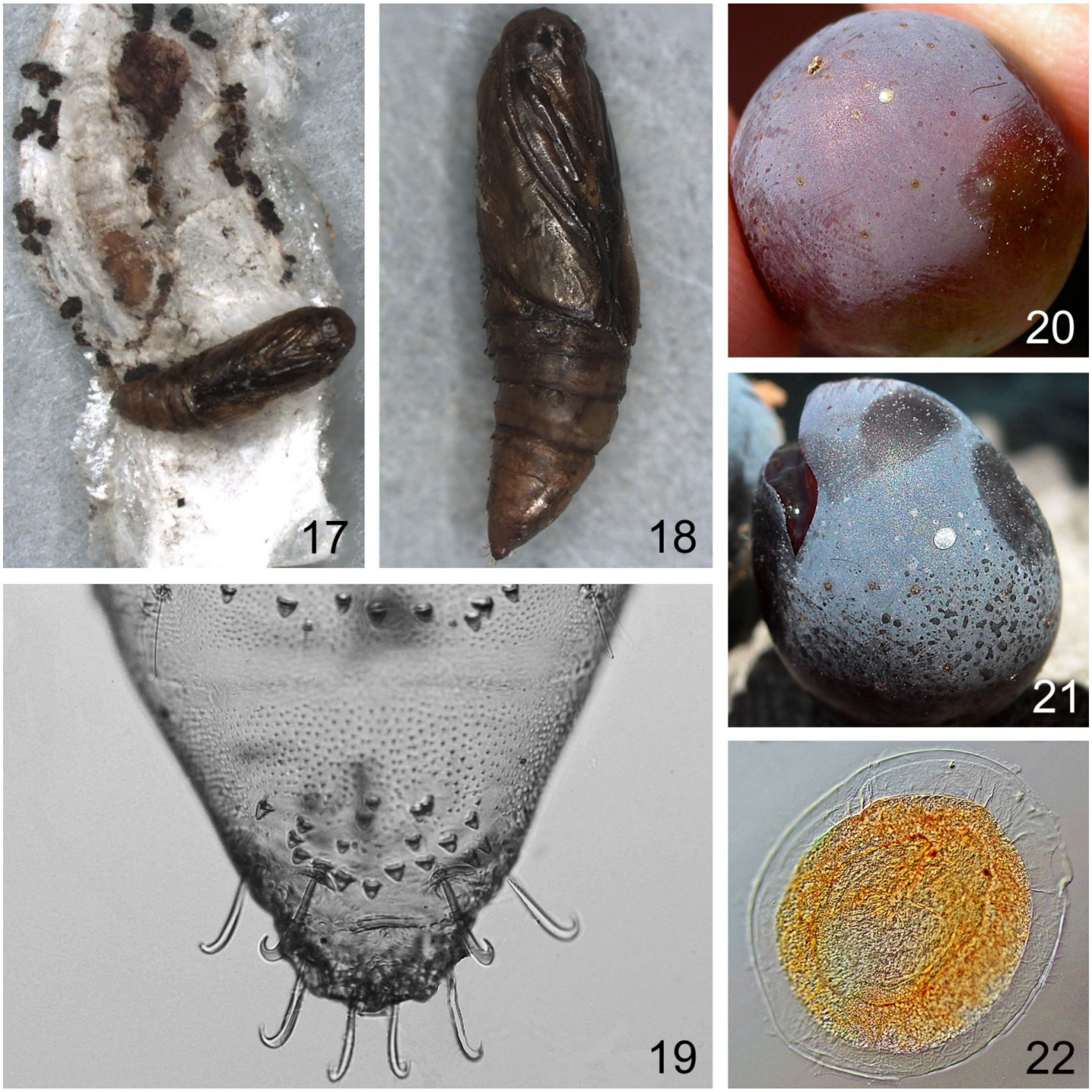


Figure 68. *Lobesia botrana* eggs and pupae; 17: pupa and cocoon; 18: pupa; 19: pupa, dorsal aspect of A10 with patch of spines; 20: egg on grape; 21: egg on grape; 22: embryo inside of translucent egg.

Paralobesia viteana (Clemens) is a native North American pest of grapes with an almost identical larval morphology to *L. botrana*. The two species presently have separate distributions: *P. viteana* occurs in the eastern U.S., ranging as far west as Colorado, while *L. botrana* is currently restricted to California. Although it appears that the brown to greenish yellow antenna of *P. viteana* (Ensminger 1958) differs from the black or dark brown antenna of *L. botrana*, these two species cannot be reliably separated based solely on larval morphology. Ensminger (1958) and MacKay (1959) illustrate the larvae of *P. viteana*.

Should *L. botrana* spread to areas outside of California, or become common on hosts other than grape, larval identification will be difficult. Characters in the published (MacKay 1959) and unpublished literature (USDA/APHIS/PPQ training aids) such as attenuated adfrontal areas, or the spiracle size compared to the SD1 pinaculum, seem too variable or difficult to interpret to be useful for identification of these closely related taxa. Minute differences in crochet patterns might be more useful. Immature stages should be reared to adults or molecular diagnostics implemented if accurate larval identifications are needed for quarantine decisions in areas outside of California and/or on hosts other than grape.

Pupa.—The most accurate pupal description of *L. botrana* was given by Patočka and Turčáni (2005). Like many tortricids, the pupa (Figs. 68.17–19) is initially greenish brown and later turns dark brown; however, cast pupal skins are somewhat unusual in retaining a greenish tint on the anterior abdominal segments. The average length of male and female pupae is 5.5 mm and 7.0 mm, respectively (CPC 2007). Important structural features of *L. botrana* include: head unmodified, without projections; clypeus with two pairs of setae; A4 and A5 with 22–24 spines between the D2 setae; dorsum of A10 with a patch of spines (Fig. 68.19) and no setae present on the anal rise (Patočka and Turčáni 2005). The cremaster is fan-shaped with a weakly emarginate caudal margin (Meijerman and Ulenberg 2000).

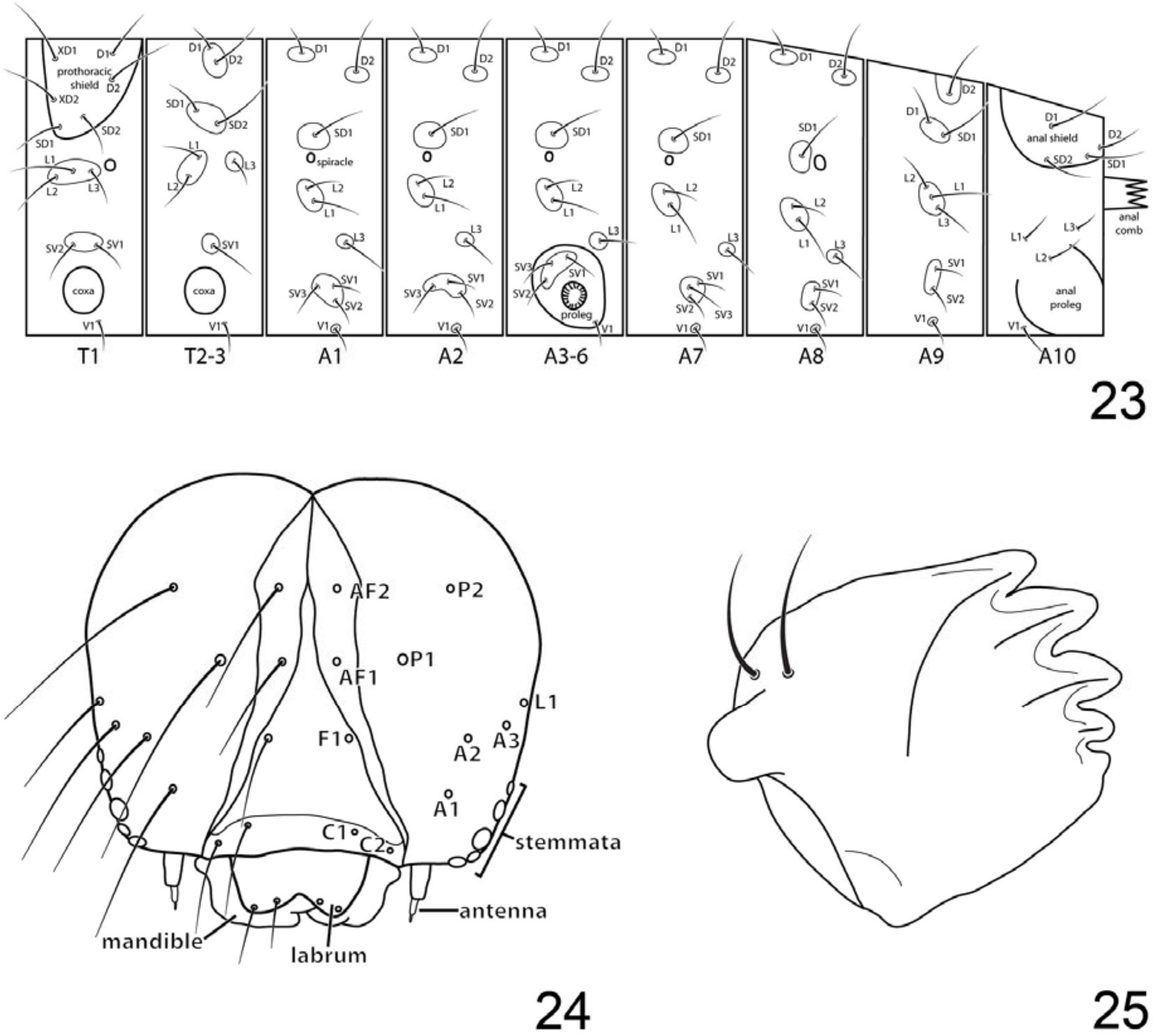


Figure 69. *L. botrana* larval details; 23: complete setal map; 24: head with setal map; 25: mandible.

On agriculturally important plants in North America, most pupae with two rows of abdominal spines are those of Tortricidae. Mosher's (1916) key to families is still the standard, although recent workers have modified this diagnosis slightly (Horak 1991, Passoa 2008), and her classification is outdated. Mosher (1916) examined the pupa of *L. botrana* and recognized it as distinct from *P. viteana*. The broad cremaster lacking thick curved hooks at the lateral margin, the presence of spines on A9, the lack of setae on the anal rise and presence of spine patch on A10 was used to define *Lobesia* (=*Polychrosis*).

As with the larvae, *L. botrana* pupae are morphologically similar to those of *P. viteana*. Both species have a patch (or weakly defined row) of spines on the dorsum of A10 (Fig. 68.19) and they can be separated by the number of spines on the anterior row of segments A4 and A5. There are 22–24 spines between the D2 setae in *L. botrana* (Patočka and Turčáni 2005) whereas *P. viteana* never has more than 15 spines in that region.

Larvae of *L. botrana* spin an opaque, white cocoon approximately 8-10 mm in length that is covered in frass (Fig. 68.17). The presence of a cocoon can be used to separate *L. botrana* pupae from other common California tortricid grape pests that do not spin a cocoon, such as *P. stultana* and *A. franciscana*.

Adult.—Forewing length ranges from 4.5–8.5 mm (Bradley et al. 1979). Forewing pattern (Figs. 65.1–2) exhibits little variation and no sexual dimorphism. Forewing pattern is as follows: ground color cream; interfascial areas overlaid with leaden gray; costal strigulae cream, well defined; fasciae brown to dark brown; subbasal fascia well defined, with black scaling medially; median fascia well defined, with triangular medial projection often suffused with black scaling; postmedian fascia broken, forming pretornal patch along dorsum with cluster of black scales; postmedian band forming large brown patch along termen; apex often with conspicuous black dot; termen outlined in cream; fringe brown. The males lack a forewing costal fold. The male hindwing is whitish with a brown periphery while the female hindwing is completely brown.

Male genitalia (Figs. 66.6–7) can be distinguished by a combination of the following characters: socii short, lateral, apex with numerous setae; uncus reduced to a small bilobed hump on tegumen; gnathos weakly sclerotized; valvae long and narrow with dense row of strong spines on ventral margin; cucullus densely setose, separated from sacculus by a distinct

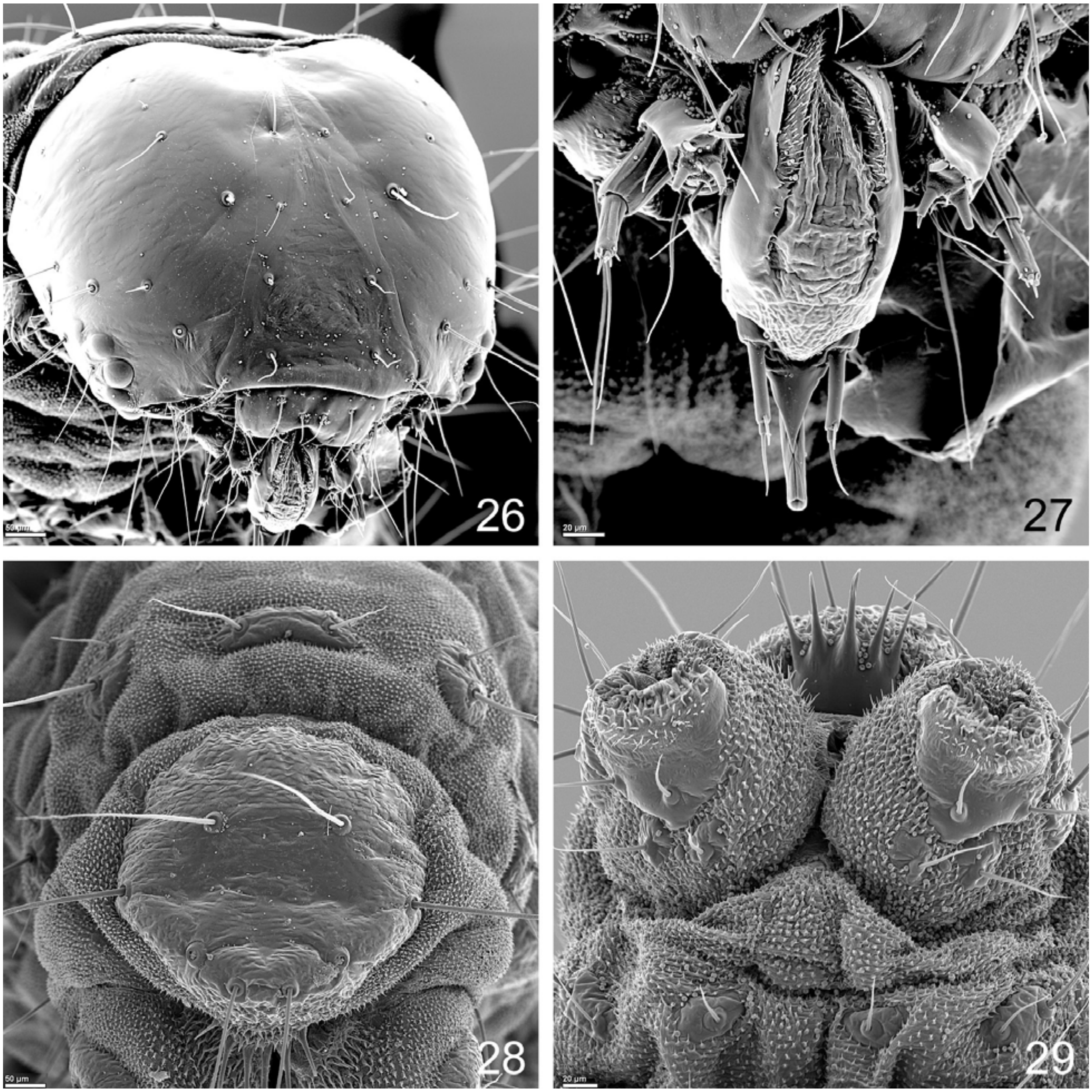


Figure 70. SEMs of *L. botrana* larva; 26: head; 27: spinneret; 28: dorsal aspect of segments A9-10 with anal shield; 29: ventral aspect of segments A9-10 showing spacing of V1 setae, 4 setae on the anal proleg, and anal comb with 5 teeth.

gap in the row of ventral spines (Fig. 66.7); sacculus weakly concave postmedially; phallus small; cornuti absent. Female genitalia (Fig. 66.5) are characterized by a long, slender ductus bursae that is undifferentiated from the corpus bursae, gradually expanded anteriorly, and an unusual, elongate, somewhat feather-shaped signum.

Lobesia botrana is similar in size and wing pattern to several species of *Paralobesia*, specifically *Paralobesia viteana* (Figs. 65.3–4), which is a native pest of grapes in eastern North America. The two species can be separated by genitalic structures: *P*. *viteana* has a sclerotized lobe projecting from the ventral base of the male cucullus that is absent in all other Nearctic olethreutines (Fig. 66.8), and the female corpus bursae lacks a signum and has two small lobelike anterior accessory bursae (Fig. 66.9) (Gilligan et al. 2008). Only three species of *Paralobesia* have been recorded from the West Coast. *Paralobesia palliolana* (McDunnough) was collected in the late 1990's near Olympia, Washington (E. LaGasa, pers. comm.). An apparently undescribed species of *Paralobesia* was reared from *Calocedrus* sp. (Cupressaceae) in California (J. Powell, pers. comm.). Another undescribed species of *Paralobesia* was discovered in Oregon as a non-target species in an *E*. *postvittana* pheromone trap on 6 August 2009 (R. Worth, pers. comm.); it is unknown if this species is native to or established in Oregon or present in California. All of the species of *Paralobesia* mentioned here can be separated from *L. botrana* by features of the male and female genitalia; it is unknown if any of them are attracted to *L. botrana* pheromone. Other species of *Paralobesia* in the eastern U.S. are treated in detail by Gilligan et al. (2008).

In California, common tortricid pests recorded feeding on grape include *P*. *stultana*, *A*. *franciscana*, and *E*. *postvittana*. Gilligan and Epstein (2009) provided diagnostic characters for these three species, the adults of which are easily distinguished from *L. botrana* by wing pattern and genitalic structure (e.g., see Brown et al. 2010, Gilligan et al. 2010).

Two other species of *Lobesia*, *Lobesia carduana* (Busck) and *Lobesia bicinctana spiraeae* (McDunnough), are present in North America. The former is a leaf-roller on thistle (Asteraceae) in the eastern United States. The latter was described as *Polychrosis spiraeae* by McDunnough (1938) from individuals feeding in flower heads of *Spiraea* (Rosaceae) in Nova Scotia. It was relegated to a subspecies of *Lobesia bicinctana* (Duponchel) by Obraztsov

(1953). Both *Lobesia* species can be distinguished from *L. botrana* by genitalic characters. The Palearctic *L. bicinctana* is illustrated by Razowski (2003), and *L. carduana* is illustrated by Gilligan et al. (2008).

Discovery of *Lobesia botrana* in California

The introduction of *L. botrana* into the United States was predicted as early as 1904 by W. D. Kearfott who wrote: “It is not at all unlikely that the European *botrana* will be at some time introduced in this country with imported grape vines…” Kearfott’s prediction came true on September 15, 2009 when several Lepidoptera larvae were collected from a vineyard 18 km northwest of Napa, Napa County, California. Napa County officials at the site noted that every grape cluster in the vineyard was damaged and that many clusters were infected with *Botrytis*. Larvae were forwarded to M. Epstein at the California Department of Agriculture’s (CDFA) Plant Pest Diagnostics Branch for identification. DNA was extracted from several larvae, amplified and sequenced. On September 30, barcode sequences (650 base-pair region of the mitochondrial gene cytochrome oxidase I) from the unknown larvae were compared with sequence data of a known *L. botrana* specimen obtained from Spain in 2009 by T. Gilligan and sequenced by USDA-CPHST Mission Laboratory. The two sequences were identical. One adult and several larvae were received by J. Brown at the USDA Systematic Entomology Laboratory on October 8 where the adult was confirmed as *L. botrana* and the six larvae identified as *Lobesia* sp. CDFA immediately initiated pheromone trapping in grape-growing regions of Napa and Sonoma Counties. The discovery of *L. botrana* in California was informally reported in several newspaper articles and press releases on October 12 and 13. Adult *L. botrana* were collected in pheromone traps in Napa and Sonoma Counties through late October, 2009.

Larvae of *L. botrana* were unknowingly collected in California the previous year. On September 11, 2008, several larvae were found in grape clusters in Napa Valley. A second series of larvae were collected on September 17. The larvae, originally thought to be those of orange tortrix, *Argyrotaenia franciscana* (Walsingham), were sent to M. Epstein for identification. The larvae were confirmed as Tortricidae but could not be identified to genus or species. Attempts to rear the larvae to adults were unsuccessful as they died as pupae in cocoons during the winter, sealed in a growth chamber. Immediate molecular diagnosis of the larvae was not possible because public barcode reference sequences of *L. botrana* were not available in 2008 and attempts to obtain reference larvae for *P. viteana* from the eastern United States and *L. botrana* from Europe were unsuccessful. In October, 2009, DNA sequence data from larvae captured in 2008 were found to match (e.g., 100%) sequence data of larvae captured from the original collecting site in 2009.

A statewide survey of California for *L. botrana* was initiated in early 2010. As of October 2010, individuals of *L. botrana* have been collected from the following California counties (dates represent the first collection date): Fresno, April 27, 2010; Mendocino, April 26, 2010; Merced, May 10, 2010; Monterey, May 10, 2010; Napa, September 17, 2008; San Joaquin, August 2, 2010; Santa Clara, September 15, 2010; Santa Cruz, September 1, 2010; Solano, April 19, 2010; and Sonoma, March 29, 2010. A total of 100,945 individual *L. botrana* have been captured in California through October 2, 2010 (K. Hoffman, pers. comm.).

Conclusions

Increased globalization has led to a concomitant increase in the potential transport, introduction, and establishment of plant and animal species around the world. Invasive species cost the U.S. billions of dollars per year in agricultural and forest ecosystem losses (Pimentel et al. 2000). The introduction of pests such as *L. botrana* into North America represents not only potential economic impacts to agroecosystems but also may result in costly quarantine actions with trading partners. The European grape vine moth may negatively affect grape production in

California and elsewhere if it proves able to spread from the Napa Valley to other grape growing regions. The association with stone and pome fruits, although rare, is equally disturbing. *Eupoecilia ambiguella* sometimes coexists with *L. botrana* in Europe although the distribution and abundance of each species is greatly influenced by climate (Roehrich and Boller 1991). Regulatory officials should consider *E. ambiguella* to be another potential pest of grapes that could follow *L. botrana* into the vineyards, especially in cooler areas.

Control and management of invasive species begin with their early detection and accurate identification. The need to support local moth surveys in analyzing exotic pest establishments has already been demonstrated (Brown et al. 2010). Hence, entomologists at the local, state, and federal levels are encouraged to cooperate in the continued monitoring and management of this newly established invasive pest.

References

Bae, Y.-S. and F. Komai. 1991. A revision of the Japanese species of the genus *Lobesia* Guenée (Lepidoptera, Tortricidae), with description of a new subgenus. Tyo to Ga. 42: 115–141.

Ben-Yehuda, S., Y. Izhar, M. Wysoki and Q. Argaman. 1993. The grape berry moth, *Lobesia botrana* (Denis & Schiffermüller) (Lepidoptera: Tortricidae), in pear orchards in Israel. International Journal of Pest Management. 39: 149–151.

Bovey, P. 1966. Super-famille des Tortricoidea, pp. 456–893. *In* A. S. Balachowsky, ed. Entomologie appliquée a l'agriculture. Volume 2 (1), Lepidoptères. Mason & Cie, Paris. 1057 pp.

Bradley, J. D., W. G. Tremewan and A. Smith. 1979. British Tortricoid Moths, Tortricidae: Olethreutinae. The Ray Society, London. 336 pp.

Brown, J. W. 2005. Tortricidae (Lepidoptera). *In* World Catalogue of Insects 5: 1–741.

Brown, J. W. and J. A. Powell. 1991. Systematics of the *Chrysoxena* Group of Genera (Lepidoptera: Tortricidae: Euliini). University of California Publications in Entomology 111. 87 pp. + figs.

Brown, J. W., G. Robinson and J. A. Powell. 2008. Food plant database of the leafrollers of the world (Lepidoptera: Tortricidae) (Version 1.0.0). http://www.tortricidae.com/foodplants.asp [accessed 24 January 2011].

Brown, J. W., M. E. Epstein, T. M. Gilligan, S. C. Passoa and J. A. Powell. 2010. Biology, discovery, and history of the light brown apple moth, *Epiphyas postvittana* (Walker) (Lepidoptera: Tortricidae: Archipini) in California: an example of the importance of local faunal surveys to document the establishment of exotic insects. American Entomologist. 56 (1): 34–43.

CPC (Crop Protection Compendium). 2007 Edition. CAB International, Wallingford, UK. http://www.cabicompendium.org/cpc/home.asp [accessed 29 October 2009].

Dufrane, A. 1960. Microlépidoptères de la faune belge. Bulletin de l'Institut Royal des Sciences Naturelles de Belgique. 36 (29): 1–16.

Ensminger, L. G. 1958. Identification of stored product insects by the micromorphology of the exoskeleton. VIII. Moth larvae. Journal of the Association of Official Agricultural Chemists. 41: 704–738.

Fermaud, M. and R. Le Menn. 1992. Transmission of *Botrytis cinerea* to grapes by grape berry moth larvae. Phytopathology. 82: 1393–1398.

Filip, I. 1986. Breeding zones of the grape moth (*Lobesia botrana* Den. & Schiff.) in Romania. Probleme de Protectia Plantelor. 14: 25–30.

Fowler, G. and K. Lakin. 2002. Risk assessment: Vine moth, *Lobesia botrana* (Denis and Schiffermüller), (Lepidoptera: Tortricidae). USDA-APHIS-CPHST Internal Report. Raleigh, North Carolina. 15 pp.

Gabel, B. and V. Mocko. 1984. Forecasting the cyclical timing of the grape vine moth, *Lobesia botrana* (Lepidoptera, Tortricidae). Acta Entomologica Bohemoslovaca. 81: 1–14.

Gilligan, T. M., D. J. Wright and L. D. Gibson. 2008. Olethreutine Moths of the Midwestern United States, an Identification Guide. Ohio Biological Survey, Columbus, Ohio. 334 pp.

Gilligan, T. M. and M. E. Epstein. 2009. LBAM ID: Tools for Diagnosing Light Brown Apple Moth and Related Western U.S. Leafrollers (Tortricidae: Archipini) [CD-ROM]. USDA-APHIS.

Gilligan, T. M., S. C. Passoa and M. E. Epstein. 2010. European grapevine moth *Lobesia botrana* (Denis & Schiff.) screening and identification aid, pp. 6–11. *In* 2010 taxonomic guidelines, screening & identification aid for the European grapevine moth survey. *Lobesia botrana* (Denis & Schiff.) Tortricidae. USDA-APHIS-PPQ. 11 pp.

González, R. H. 2008. Biología, desarrollo, caracterización, de daños y manejo fitosanitario de la polilla Europea de la vid, Lobesia botrana (D and S) (Lep., Tortricidae). Universidad de Chile, Direccion de Extensión. Santiago, Chile. 25 pp.

Horak, M. 1991. Morphology, pp. 1–22. *In* L. P. S. van der Geest and H. H. Evenhuis, eds. Tortricid Pests: Their Biology, Natural Enemies and Control. Elsevier, Amsterdam, The Netherlands.

International Commission on Zoological Nomenclature. 1999. International Code of Zoological Nomenclature, Fourth Edition. 306 pp.

Irigaray, F. J., F. Moreno, I. Pérez-Moreno and V. Marco. 2006. Larval growth and the effect of temperature on head capsule size in *Lobesia botrana* (Lepidoptera: Tortricidae). Environmental Entomology. 35: 189–193.

Kearfott, W. D. 1904. North American Tortricidae. Transactions of the American Entomological Society. 30: 287–299.

MacKay, M. R. 1959. Larvae of the North American Olethreutidae (Lepidoptera). The Canadian Entomologist Supplement 10. 338 pp.

Maher, N. and D. Thiéry. 2006. *Daphne gnidium*, a possible native host plant of the European grapevine moth *Lobesia botrana*, stimulates its oviposition. Is a host shift relevant? Chemoecology. 16: 135–144.

Marchal, P. 1912. Rapport sur les travaux accomplis par la mission d’études de la Cochylis et de l’Eudémis. Librairie Polytechnique, Paris et Liège. 326 pp.

Masante-Roca, I., S. Anton, L. Delbac, M-C. Dufour and C. Gadenne. 2007. Attraction of the grapevine moth to host and non-host plant parts in the wind tunnel: effects of plant phenology, sex, and mating status. Entomologia Experimentalis et Applicata. 122: 239–245.

McDunnough, J. H. 1938. Some apparently new Eucosmidae (Lepid.). Canadian Entomologist. 70: 90–100.

Meijerman, L. and S. A. Ulenberg. 2000. Arthropods of Economic Importance: Eurasian Tortricidae. CDROM. Zoological Museum, University of Amsterdam.

Mosher, E. 1916. A classification of the Lepidoptera based on characters of the pupa. Bulletin of the Illinois State Laboratory of Natural History. 12: 17–159.

Obraztsov, N. S. 1953. Classification of Holarctic species of the genus *Lobesia* Guenée, with description of *Paralobesia* gen. nov. (Lepidoptera, Tortricidae). Tijdschrift voor Entomologie. 96: 85–94.

Passoa, S. 1985. Taxonomy of the Larvae and Pupae of Economically Important Pyralidae in Honduras. M.S. thesis, University of Florida, Gainesville. 486 pp.

Passoa, S. 2008. Part III: Immature stages, pp. 295–314. *In* Gilligan, T. M., D. J. Wright, and L. D. Gibson. Olethreutine Moths of the Midwestern United States, an Identification Guide. Ohio Biological Survey, Columbus, Ohio.

Patočka, J. and M. Turčáni. 2005. Lepidoptera pupae. Central European species. Apollo Books. Stenstrup, Denmark. 863 pp.

Pierce, W. D. 1918. A manual of dangerous insects likely to be introduced in the United States through importations. United States Department of Agriculture Bureau of Entomology. Government Printing Office, Washington, D. C. 256 pp.

Pimentel, D., L. Lach, R. Zuniga, and D. Morrison. 2000. Environmental and economic costs of nonindigenous species in the United States. Bioscience. 50: 53–65.

Ragonot, E. L. 1894. Notes synonymiques sur les Microlepidoptè et descriptions d'éspèces peu connues or inédites. Annales de la Société Entomologique de France. 209: 161–226.

Razowski, J. 2003. Tortricidae of Europe, Vol. 2, Olethreutinae. Frantisek Slamka, Slovakia. 301 pp.

Roehrich, R. and E. Boller. 1991. Tortricids in vineyards, pp. 507–514. *In* L. P. S. van der Geest and H. H. Evenhuis, eds. Tortricid Pests: Their Biology, Natural Enemies and Control. Elsevier, Amsterdam, The Netherlands.

SAG (Servicio Agrícola y Ganadero). 2010. Gobierno de Chile, Ministerio de Agricultura. http://www.sag.gob.cl/OpenNews/asp/pagDefault.asp?argInstanciaId=1&argNoticiaId=464&argEdicionId=9 [accessed 17 March 2010].

SENASA (Servicio Nacional de Sanidad y Calidad Agroalimentaria). 2010. El Senasa declaró la emergencia fitosanitaria por la detección de la plaga *Lobesia botrana*. http://www.senasa.gov.ar/ contenido.php?to=n&in=1453&ino=0&io=12316 [accessed 15 March 2010].

Silvestri, F., 1912. Contribuzioni alla conoscenza degli insetti dannosi e dei loro simbionti III. La tignoletta dell'uva (*Polychrosis botrana* Schiff.) con un cenno sulla Tignola dell'uva (*Conchylis ambiquella* Hb.). Bollettino del Laboratorio di Zoologia generale e Agraria della R. Scuola Superiore in Portici. 6: 246–307.

Swatschek B. 1958. Die larvalsystematik der Wickler (Tortricidae und Carposinidae). Abhandlungen Zur Larvalsystematik der Insekten nr. 3. Berlin, Germany: Akademie- Verlag, 269 pp.

Tasin, M. 2005. Sex, Wine and Chemical Communication in Grapevine Moth *Lobesia botrana*. Doctoral thesis. Swedish University of Agricultural Sciences, Department of Crop Science, Alnarp, Sweden. 30 pp.

Thiéry, D. and J. Moreau. 2005. Relative performance of European grapevine moth (*Lobesia botrana*) on grapes and other hosts. Oecologia. 143: 548–557.

Venette, R. C., E. E. Davis, M. DaCosta, H. Heisler and M. Larson. 2003. Mini risk assessment. Grape berry moth, *Lobesia botrana* (Denis and Schiffermüller) [Lepidoptera: Tortricidae]. USDA/APHIS/PPQ Pest Risk Assessment. 29 pp.

Voigt, E. 1972. Biologie und Bedeutung der Traubenwickler im ungarischen Weinbau. Weinberg und Keller. 19: 615–632.

Whittle, K. 1985. Pests not known to occur in the United States or of limited distribution. No 60. European grape vine moth. USDA/APHIS/PPQ 81–46. 10 pp.

Chapter 5

Discovery of false codling moth, *Thaumatotibia leucotreta* (Meyrick), in California (Lepidoptera: Tortricidae)*

Introduction

Thaumatotibia leucotreta (Meyrick), the false codling moth (FCM), is a native of sub-Saharan Africa. It is a highly polyphagous species, recorded on more than 50 species of plants in over 30 families (van der Geest et al. 1991, Brown et al. 2008), feeding primarily on the fruit. Many of its hosts are important cash crops, and larvae are known to cause serious damage to avocado, citrus, cotton, macadamia, mangoes, and many others (Reed 1974, van der Geest et al. 1991, Erichsen and Schoeman 1994, CPC 2007). Live and dead larvae are frequently intercepted at U.S. ports-of-entry, primarily on bell peppers (*Capsicum* sp.), eggplant (*Solanum melongena*), and clementines (*Citrus* sp.). During 2005, two live FCM larvae were found in California inside previously cold treated clementines from South Africa (USDA 2005). This event prompted a reevaluation and strengthening of the treatment protocol, and no further such detections have been reported.

On July 24, 2008, a male false codling moth was identified from a pheromone trap located in Ventura County, California. This is the first record of a live adult *T. leucotreta* in North

---

*This chapter is published as:

Gilligan, T. M., M. E. Epstein & K. M. Hoffman. 2011. Discovery of false codling moth, *Thaumatotibia leucotreta* (Meyrick), in California (Lepidoptera: Tortricidae). Proceedings of the Entomological Society of Washington. 113(4).

America. The purpose of this chapter is to document the discovery of *T. leucotreta* in California and to provide descriptions, illustrations, and other information to help in the identification of this species.

Materials and methods

The single specimen of *T. leucotreta* from California was dissected and compared to illustrations in Komai (1999) and authoritatively identified specimens from South Africa in the collections of the California Department of Food and Agriculture and the National Museum of Natural History. Recently collected specimens from Kenya were used for wing pattern and morphological illustrations. Dissection methods essentially follow those presented by Brown and Powell (1991) except some preparations were not transferred to xylene and were mounted in Euparal (Bioquip Products, Rancho Dominguez, CA) rather than Canada balsam. Adults were photographed with a Canon EOS 40D digital SLR camera (Canon U.S.A., Lake Success, NY) mounted on a Visionary Digital BK Lab System (Visionary Digital, Palmyra, VA). Photographs of genitalia were taken using a Nikon DXM 1200 digital camera mounted on a Nikon Labophot2 compound microscope (Nikon Instruments, Melville, NY). Some photographs are a combination of several layers produced with Helicon Focus 4.80 software (Helicon Soft Ltd., Kharkov, Ukraine). All photographs were edited using Adobe Photoshop CS3 Extended (Adobe Systems Inc., San Jose, CA). Morphological terminology and wing pattern descriptions follow Komai (1999) and Gilligan et al. (2008). Natural history and host information are compiled from Brown et al. (2008), CPC (2007), Erichsen and Schoeman (1994), Komai (1999), Reed (1974), and van der Geest et al. (1991).

Results and discussion

Nomenclature and distribution

Argyroploce leucotreta Meyrick, 1913 was described from a female collected in South Africa. Although the species was long placed in the genus *Cryptophlebia* Walsingham, 1899 (e.g., Bradley et al. 1979, Daiber 1980, van der Geest et al. 1991), Komai (1999) recognized that a group of species traditionally assigned to *Cryptophlebia* belonged in *Thaumatotibia* Zacher, 1915, and he transferred *leucotreta* and its relatives to that genus (see Brown 2006 for a detailed explanation). Despite this action, the incorrect combination *Cryptophlebia leucotreta* (Meyrick) is still used in much of the contemporary economic literature.

The false codling moth has been reported as present in approximately 40 countries on the African continent and several islands, including Madagascar, Mauritius, Réunion, and Saint Helena (CPC 2007, EPPO 2007, van der Geest et al. 1991); it is not considered established outside of Africa (EPPO 2007). The first reports of *T. leucotreta* in Europe were from Finland in 1965 (Karvonen 1983, Lopez-Vaamonde et al. 2010), and it subsequently has been found in Israel, where it was declared "present, but of limited distribution, limited host range and subject to official control" by the Israeli Plant Protection and Inspection Services in 2003 (EPPO 2003, Hamburger et al. 2000).

False codling moth is one of the most commonly intercepted tortricids on pepper (Solanaceae: *Capsicum annuum* L.) and eggplant (Solanaceae: *Solanum melongena* L.) at U.S. ports-of-entry (Brown 2006). Venette et al. (2003) report over 1,500 interceptions of "*C. leucotreta*" or "*Cryptophlebia* sp." since 1984 at a total of 34 U.S. ports. Over one-third of the interceptions were from John F. Kennedy International Airport in New York (Venette et al. 2003). Individuals have also been intercepted during quarantine inspections in Denmark, the Netherlands, New Zealand, and the United Kingdom (Dugdale et al. 2005, USDA 2010).

Larval hosts and biology

Thaumatotibia leucotreta larvae are highly polyphagous and have been recorded feeding on more than 50 species of plants in over 30 families; a list of hosts compiled from Brown et al. (2008), CPC (2007), Reed (1974), and van der Geest et al. (1991) is presented in Table 111.

Table 111: Documented host plants for *T. leucotreta*

Family	**Genus/species**	**Common name**
Anacardiaceae	*Mangifera indica* L.	mango
Anacardiaceae	*Sclerocarya birrea* (A. Rich.) Hochst.	marula
Annonaceae	*Annona muricata* L.	soursop
Annonaceae	*Annona reticulata* L.	custard apple
Asclepiadaceae	*Calotropis procera* (Aiton) W. T. Aiton	roostertree
Bombacaceae	*Ceiba pentandra* (L.) Gaertn.	kapoktree
Bromeliaceae	*Ananas comosus* (L.) Merr.	pineapple
Capparaceae	*Capparis* L.	caper
Celastraceae	*Catha edulis* (Vahl) Forssk. ex Endl.	khat
Clusiaceae	*Garcinia mangostana* L.	mangosteen
Combretaceae	*Combretum apiculatum* Sond.	red bushwillow
Combretaceae	*Combretum zeyheri* Sond.	large-fruited bushwillow
Crassulaceae	*Crassula* L.	pygmyweed
Ebenaceae	*Diospyros* L.	diospyros
Ebenaceae	*Diospyros virginiana* L.	common persimmon
Euphorbiaceae	*Ricinus communis* L.	castorbean
Fabaceae	*Acacia karroo* Hayne	sweet thorn
Fagaceae	*Quercus* L.	oak
Lauraceae	*Persea americana* Mill.	avocado
Malvaceae	*Abelmoschus esculentus* (L.) Moench	okra
Malvaceae	*Abutilon* Mill.	mallow
Malvaceae	*Gossypium* L.	cotton
Malvaceae	*Hibiscus* L.	rosemallow
Myrtaceae	*Eugenia* L.	stopper
Myrtaceae	*Psidium guajava* L.	guava
Olacaeae	*Ximenia caffra* Sond.	sourplum
Oleaceae	*Olea europaea* L.	olive
Oxalidaceae	*Averrhoa carambola* L.	carambola
Poaceae	*Saccharum officinarum* L.	sugarcane
Poaceae	*Sorghum* Moench	sorghum
Poaceae	*Zea mays* L.	corn

Table 111: Documented host plants for *T. leucotreta*

Family	**Genus/species**	**Common name**
Proteaceae	*Macadamia integrifolia* Maiden & Betche	macadamia nut
Punicaceae	*Punica granatum* L.	pomegranate
Rosaceae	*Prunus persica* (L.) Batsch	peach
Rosaceae	*Prunus* L.	plum
Rubiaceae	*Coffea arabica* L.	Arabian coffee
Rubiaceae	*Coffea* L.	coffee
Rubiaceae	*Vangueria infausta* Burch.	medlar
Rutaceae	*Citrus sinensis* (L.) Osbeck	navel orange
Rutaceae	*Citrus* L.	citrus
Sapindaceae	*Litchi chinensis* Sonn.	lychee
Sapotaceae	*Englerophytum magaliesmontana* (Sond.) T. D. Penn.	stem fruit
Solanaceae	*Capsicum annuum* L.	cayenne pepper
Solanaceae	*Solanum melongena* L.	eggplant
Stericulaceae	*Cola nitida* (Vent.) A. Chev.	ghanja kola
Theaceae	*Camellia sinensis* (L.) Kuntze	tea

In Africa, false codling moth is a serious pest of citrus (*Citrus* spp. L.), cotton (*Gossypium* spp. L.), and avocado (*Persea americana* Mill.) (Erichsen and Schoeman 1994, Reed 1974, van der Geest et al. 1991). It has also been reported causing serious damage to corn (*Zea mays* L.), guava (*Psidium guajava* L.), macadamia (*Macadamia integrifolia* Maiden & Betche), mango (*Mangifera indica* L.), peach (*Prunus persica* (L.) Batsch), and other horticultural crops (CPC 2007, Erichsen and Schoeman 1994).

The biology of *T. leucotreta* has been thoroughly reviewed by Reed (1974; on cotton), van der Geest et al. (1991; on citrus), Erichsen and Schoeman (1994; on avocado), and Daiber (1979a, b, c, 1980; in controlled laboratory conditions); the following is a summary from those accounts. Development times, egg production, and adult lifespan are all highly temperature dependent. Daiber (1979a, b, c, 1980) and Venette et al. (2003) provide a thorough summary of *T. leucotreta* phenology and developmental thresholds.

Females deposit eggs singly or in small groups on the surface of smooth fruit. A single female may produce between 87–456 eggs in her lifetime (with a maximum of 799) (Daiber

1980). On fruit, larvae tunnel into the pith or feed beneath the surface. On cotton, larvae mine the wall of the boll and later move into the center of the boll to feed on the seeds. Larvae complete five instars. Last instar larvae exit the fruit or boll, drop from the host plant, and pupate in a silken cocoon in the soil, under leaf litter, or in bark crevices. The false codling moth is not known to diapause, and development is continuous; as many as 10 generations are possible per year in South Africa (Begemann and Schoeman 1999). The absence of a diapause may lead to host shifts and varied developmental rates in times of drought or when preferred host plants are unavailable.

Larvae cause significant damage by feeding directly on fruit or bolls. Feeding in citrus fruit can result in premature ripening and fruit drop as well as secondary infection by fungi. Larval feeding in cotton results in secondary infection by fungus and bacteria, causing rotting of the bolls. Feeding in avocado fruit results in lesions on the fruit and secondary infection by bacteria and fungi. Chemical control of this species is difficult due to the highly polyphagous, internal feeding larvae, and crop losses can be as high as 10–20% during serious citrus infestations (van der Geest et al. 1991). Control of false codling moth in South Africa is achieved through a combination of chemical control, mating disruption, attract and kill, natural enemies, and sterile insect technique (SIT) (Hofmeyr et al. 2005).

Morphology

The morphology of *T. leucotreta* has been described in detail by many authors: Daiber (1979a) describes the egg; Williams (1953), Daiber (1979b), Komai (1999), Dugdale et al. (2005) and Timm et al. (2007) describe the larvae; Daiber (1979c), Komai (1999), and Timm et al. (2007) describe the pupa; and Meyrick (1913), Bradley et al. (1979), Komai (1999), and Dugdale et al. (2005) detail the adult morphology. Unless otherwise noted, the following abridged description is summarized from these sources or personal observations.

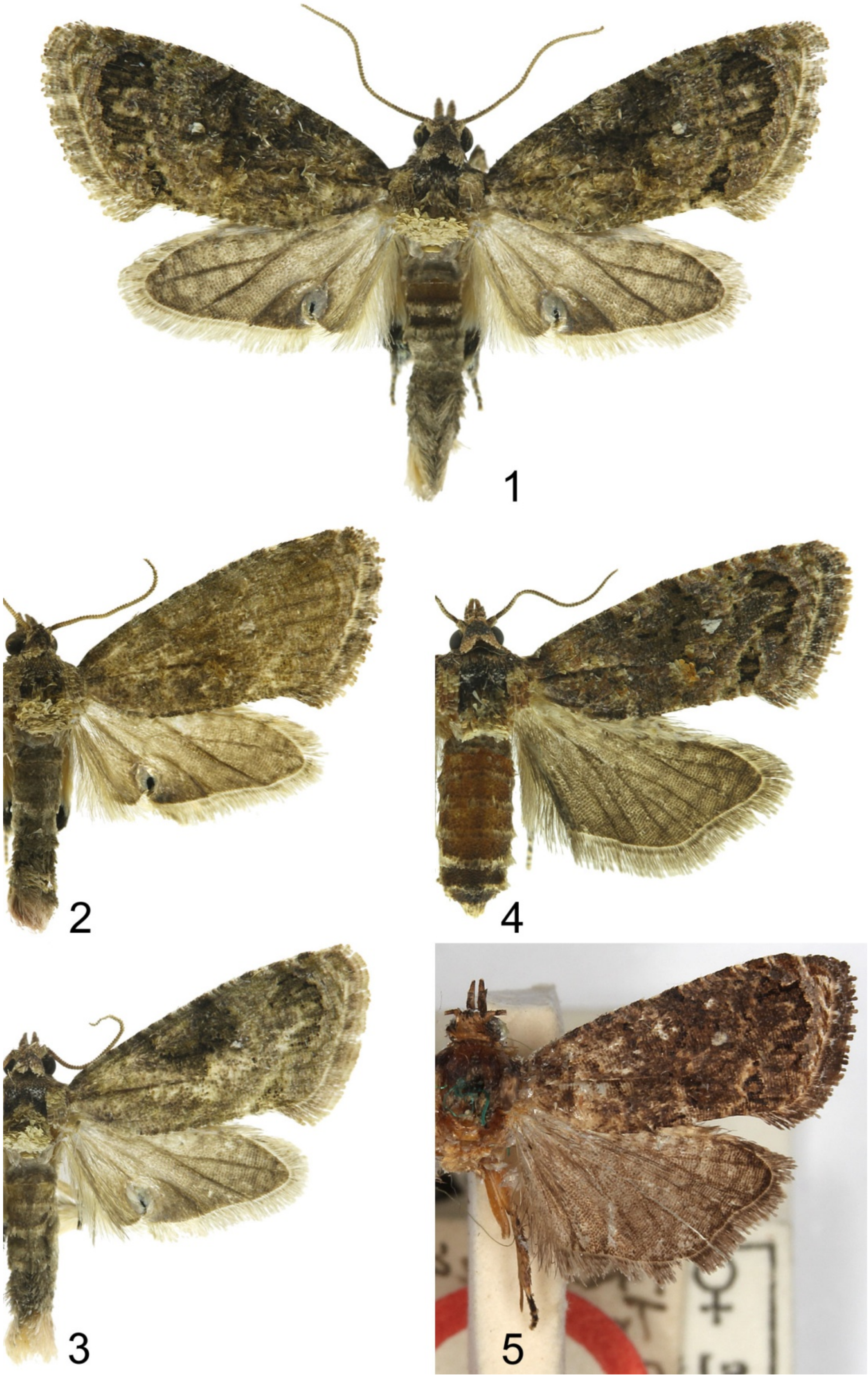


Figure 71. Adult *T. leucotreta* wing patterns; 1–3: males; 4: female; 5: female holotype.

Egg.—The egg of *T. leucotreta* is flat and elliptical with a granulated chorion. Each egg is approximately 0.77 mm long by 0.60 mm wide. When newly laid, eggs are cream colored, eventually turning a reddish color with the embryo becoming visible prior to eclosion.

Larva.—First instar larvae are ca. 1 mm in length and are pale with dark pinacula. Mature larvae are ca. 12–18 mm long with a yellowish brown to dark brown head and prothoracic shield. The abdomen is orange to pink with large pinacula that are darker than the body color. Daiber (1979b) gives head capsule measurements for each instar.

Thaumatotibia leucotreta larvae can be distinguished from many tortricids in California by the following combination of characters: L pinaculum on T1 enlarged, extending beneath and beyond (posterad of) spiracle; D1 and SD1 on A9 on same pinaculum, separate from D2; L group on A9 trisetose; anal comb present with 2–10 teeth. Other larval characters of *T. leucotreta* include: SD2 on A1–8 highly reduced or appearing absent; SV groups on A1, 2, 7, 8, 9 with 3:3:2:2:1 setae; spiracle on A8 displaced posterad of SD pinaculum; D2 setae on A9 on shared mid-dorsal “saddle” pinaculum; V setae on A9 slightly further apart than those on A8.

The larval characters listed here are not completely diagnostic, and *T. leucotreta* larvae are difficult to separate from Cochylini larvae, which can share the same set of character states (e.g., enlarged L-pinaculum on A9, spiracle on A8 displaced posterad, anal comb present). Two possible distinguishing features are SV counts of 3:3:2:2:2 and a bisetose L-group on A9 in many Cochylini (J. Brown pers. comm.). Should *T. leucotreta* become established in California, a DNA-based system may be necessary to assist with larval identification.

Pupa.—The pupa is pale yellowish brown, ca. 8–10 mm long, and is contained in a silken cocoon constructed with organic debris and soil particles. Important morphological features of the pupa include: segments A2–A7 with two rows of dorsal spines; segments A8–A10 with one row of dorsal spines except in males, where A8 has two rows of spines; A10 with two pairs of hooked setae and one pair of strong spines along anal rise.

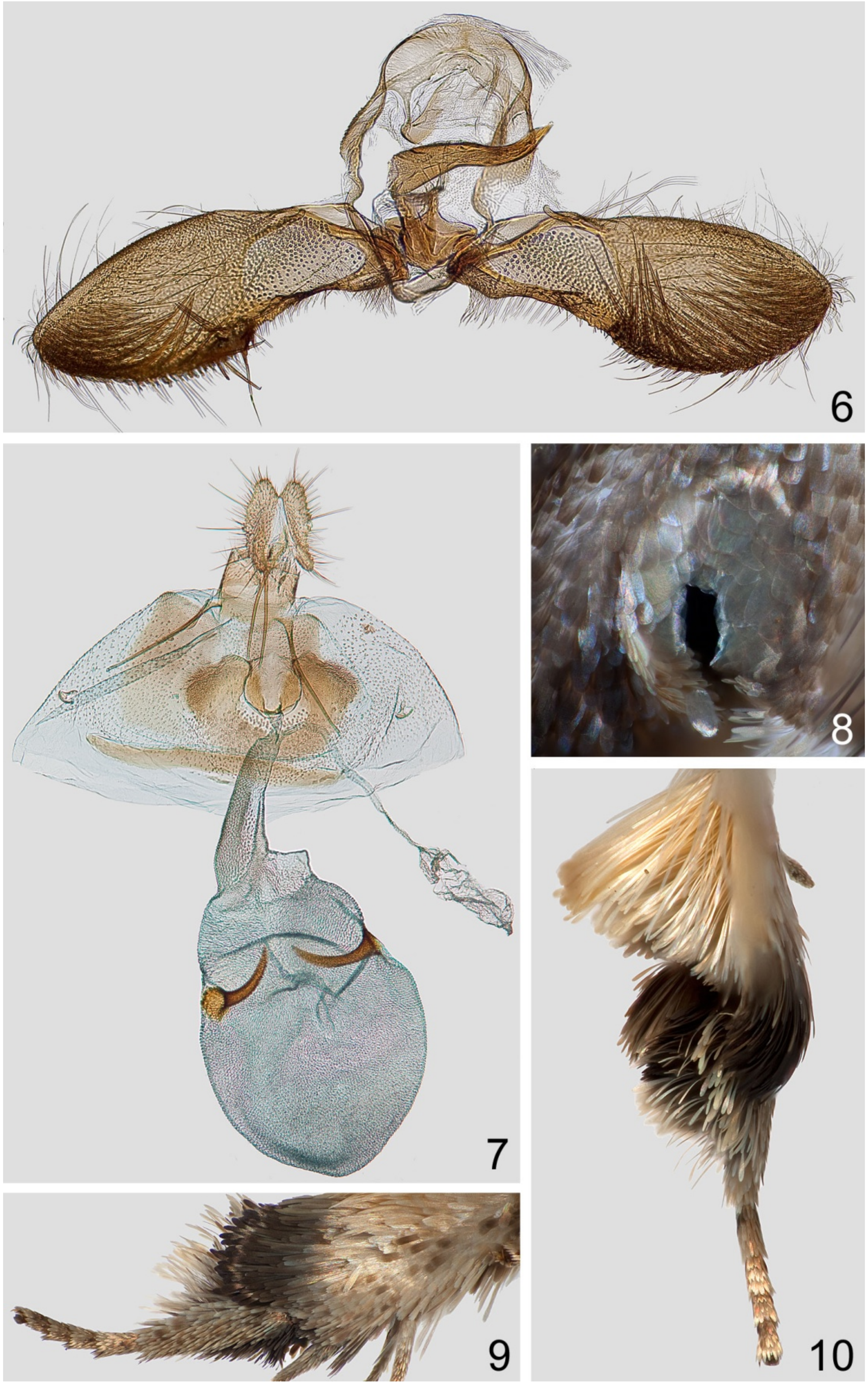


Figure 72. Morphological details of *T. leucotreta* adults; 6: male genitalia; 7: female genitalia; 8: pocket of opalescent scales on hind wing; 9–10: hind tibia.

Adult.—Adults (Figs. 71.1–5) are grayish brown to dark brown with forewing length of 7–8 mm for males and 9–10 mm for females. Males and females are sexually dimorphic, and the two differ in overall size, wing shape, and male secondary sexual characters. The male forewing is triangular, with an acute apex, while the female forewing is more elongate with a rounded apex. Male secondary characters include tufts of modified scales on the inner side of the hind tibia (Figs. 72.9–10), enlargement of the inner apical spur on the hind tibia with a batch of scales, and semicircular pocket of opalescent scales on the distal end of vein CuA2 on the hind wing (Fig. 72.8); the last character can be used to separate *T. leucotreta* males from all other North American tortricids. Forewing pattern can vary among individuals, especially in males where forewing color and pattern expression is not as consistent as in females. Most individuals exhibit a combination of four forewing pattern elements: a small white dot near the end of the discal cell; a patch of raised, usually rust colored scales near the middle of the wing; a distinct "question-mark-shaped" band of dark scales along the termen; and a semicircular band of dark scales in the middle of the costa. Male genitalia (Fig. 72.6) are characterized by a rounded tegumen lacking an uncus or socii, large rounded valvae, and a tapered aedeagus that is upcurved distally. Female genitalia (Fig. 72.7) are characterized by a semicircular sterigma, narrow ductus bursae, and large rounded corpus bursae with a pair of thorn-shaped signa.

Discovery of *Thaumatotibia leucotreta* in California

The first general survey for *T. leucotreta* in California was conducted by the California Department of Food and Agriculture (CDFA) in 2005 as part of the USDA's Cooperative Agricultural Pest Survey (CAPS) program. Pherocon® IIC traps (Trécé, Inc.) baited with *T. leucotreta* pheromone obtained from USDA were placed one per square mile in both rural agricultural land and urban residential properties, and traps were rotated to new locations every six weeks from May through October. The survey focused mainly on areas where primary hosts were most abundant and in areas identified as having a high risk for introductions. Similar surveys were conducted in 2006 and 2007, with 1000 traps deployed statewide. No individuals of *T. leucotreta* were detected in California during the 2005–2007 CAPS surveys.

In 2008, 911 traps were deployed in 26 counties from May through October as part of the CAPS false codling moth survey. A single male *T. leucotreta* was found in one of these traps on July 24, 2008 in a residential area near the Port Hueneme Harbor, Port Hueneme, Ventura County. In response, the CDFA and the USDA, in cooperation with the Ventura County Agricultural Commissioner, deployed an additional 1181 traps in a five mile delimitation radius, at a density of 36 traps per square mile. In addition, 188 properties within a 400-meter radius of the detection site were searched during August and September, and host fruit was sampled from 56 properties. No additional *T. leucotreta* were detected in 2008. Delimitation trapping continued through June 2009, for three projected life cycles, and statewide detection trapping has continued during 2009 and 2010 through the CAPS program with no additional *T. leucotreta* captures.

Conclusions

Increased globalization and the expanded flow of international commerce have led to a concomitant increase in the transport, introduction, and establishment of plant and animal species around the world. The continuing introduction of nonindigenous (non-native) species into the United States is a significant and growing problem, costing taxpayers via lost agricultural productivity, expensive prevention and eradication efforts, environmental degradation, and increased health problems. The most recent study to attempt a nationwide estimate of the economic costs to the U.S. of nonindigenous species concluded that annual costs exceed $120 billion (Pimentel et al. 2005), which equates to costs of $1,100 per U.S. household per year (Lodge et al. 2006). The false codling moth is one of many tortricids considered to have high potential for invasion into the U.S. owing to its highly polyphagous habits, which include many important agricultural crops grown in the U.S., and its ability to thrive in climates such as those found in Florida, Texas, and California. This species is commonly intercepted at ports-of-entry in North America and Europe. Vigilance at these ports and early detection methods are critical to minimize the probability of this species' introduction and establishment in the U.S.

References

Begemann, G. J. and A. S. Schoeman. 1999. The phenology of *Helicoverpa armigera* (Hübner) (Lepidoptera: Noctuidae), *Tortrix capensana* (Walker) and *Cryptophlebia leucotreta* (Meyrick) (Lepidoptera: Tortricidae) on citrus at Zebediela, South Africa. African Entomology. 7(1): 131–148.

Bradley, J. D., W. G. Tremewan, and A. Smith. 1979. British Tortricoid Moths, Tortricidae: Olethreutinae. The Ray Society, London. 336 pp.

Brown, J. W. 2006. Scientific names of pest species in Tortricidae (Lepidoptera) frequently cited erroneously in the entomological literature. American Entomologist. 52(3): 182–189.

Brown, J. W. and J. A. Powell. 1991. Systematics of the *Chrysoxena* group of genera (Lepidoptera: Tortricidae: Euliini). University of California Publications in Entomology. 111: 1–87 pp.

Brown, J. W., G. Robinson and J. A. Powell. 2008. Food plant database of the leafrollers of the world (Lepidoptera: Tortricidae) (Version 1.0.0). http://www.tortricidae.com/foodplants.asp [accessed 24 January 2011].

CPC (Crop Protection Compendium). 2007. CAB International, Wallingford, UK. http://www.cabicompendium.org/cpc/home.asp [accessed 24 January 2011].

Daiber, C. C. 1979a. A study of the biology of the false codling moth [*Cryptophlebia leucotreta* (Meyr.)]: the egg. Phytophylactica. 11: 129–132.

Daiber, C. C. 1979b. A study of the biology of the false codling moth [*Cryptophlebia leucotreta* (Meyr.)]: the larva. Phytophylactica. 11: 141–144.

Daiber, C. C. 1979c. A study of the biology of the false codling moth [*Cryptophlebia leucotreta* (Meyr.)]: the cocoon. Phytophylactica. 11: 151–157.

Daiber, C. C. 1980. A study of the biology of the false codling moth *Cryptophlebia leucotreta* (Meyr.): the adult and generations during the year. Phytophylactica. 12: 187–193.

Dugdale, J. S., D. Gleeson, L. H. Clunie and P. W. Holder. 2005. A diagnostic guide to Tortricidae encountered in field surveys and quarantine inspections in New Zealand: Morphological and molecular characters. National Plant Pest Reference Laboratory, MAF Information Bureau, Wellington. 161 pp.

EPPO (European and Mediterranean Plant Protection Organization). 2003. EPPO Reporting Service 2003, No. 02. 2003/015.

EPPO (European and Mediterranean Plant Protection Organization). 2007. PQR, EPPO plant quarantine information retrieval system, Version 4.6 (2007–07).

Erichsen, C. and A. S. Schoeman. 1994. Moth pests of avocados. South African Avocado Growers' Association Yearbook. 17: 109–112.

Gilligan, T. M., D. J. Wright and L. D. Gibson. 2008. Olethreutine Moths of the Midwestern United States, an Identification Guide. Ohio Biological Survey, Columbus, Ohio. 334 pp.

Hamburger, M., L. Zarabi, M. Weiss, Q. Argaman, W. Kuslitzky, and Z. Kein. 2000. False codling moth (*Cryptophlebia leucotreta*) in Israel. Phytoparasitica. 29: 84.

Hofmeyr, J. H., J. E. Carpenter and S. Bloem. 2005. Developing the sterile insect technique for *Cryptophlebia lecotreta* (Lepidoptera: Tortricidae): influence of radiation doses and release ratio on fruit damage and population growth in field cages. Journal of Economic Entomology. 98: 1924–1929.

Karvonen, J. 1983. *Cryptophlebia leucotreta* imported into Finland (Lepidoptera, Tortricidae). Notulae Entomologicae. 63: 94.

Komai, F. 1999. A taxonomic review of the genus *Grapholita* and allied genera (Lepidoptera: Tortricidae) in the Palaearctic region. Entomologica Scandinavica Supplementum. 55: 1–226.

Lodge, D. M., S. Williams, H. J. MacIssac, K. R. Hays, B. Leung, S. Reichard, R. N. Mack, P. B. Moyle, M. Smith, D. A. Andow, J. T. Carlton and A. McMichael. 2006. Biological invasions: recommendations for U.S. policy and management. Ecological Applications. 16: 2035–2054.

Lopez-Vaamonde, C., D. Agassiz, S. Augustin, J. De Prins, W. De Prins, S. Gomboc, P. Ivinskis, O. Karsholt, A. Koutroumpas, F. Koutroumpa, Z. Laštůvka, E. Marabuto, E. Olivella, L. Przybylowicz, A. Roques, N. Ryrholm, H. Šefrová, P. Šima, I. Sims, S. Sinev, B. Skulev, R. Tomov, A. Zilli and D. Lees. 2010. Lepidoptera, Chapter II. BioRisk. 4(2): 603–668.

Meyrick, E. 1913. Descriptions of South African Micro-Lepidoptera. IV. Annals of the Transvaal Museum. 3(4): 267–336.

Pimentel, D., R. Zuniga and D. Morrison. 2005. Update on the environmental and economic costs associated with alien-invasive species in the United States. Ecological Economics. 52: 273–288.

Reed, W. 1974. The false codling moth, *Crytophlebia leucotreta* Meyr. (Lepidoptera: Olethreutidae) as a pest of cotton in Uganda. Cotton Growing Review. 51: 213–225.

Timm, A. E., L. Warnich, and H. Geertsema. 2007. Morphological and molecular identification of economically important Tortricidae (Lepidoptera) on tropical and subtropical fruit in South Africa. African Entomology. 15(2): 269–286.

USDA (United States Department of Agriculture). 2005. Regulatory response to detections of false codling moth (FCM) by California inspectors in South African citrus. http://www.pestalert.org/oprDetail_print.cfm?oprid=145 [accessed 12 July 2011].

USDA (United States Department of Agriculture). 2010. New pest response guidelines: false codling moth *Thaumatotibia leucotreta*. U.S. Department of Agriculture, Animal Plant Health Inspection Service, Plant Protection and Quarantine, Emergency and Domestic Programs, Riverdale, Maryland. http://www.aphis.usda.gov/import_export/plants/manuals/online_manuals.shtml [accessed 24 January 2011].

van der Geest, L. P. S., C. H. Wearing and J. S. Dugdale. 1991. Tortricids in miscellaneous crops, pp. 563-577. *In* L. P. S. van der Geest and H. H. Evenhius, eds. Tortricid Pests: Their Biology, Natural Enemies, and Control. World Crop Pests, Vol. 5. Elsevier, Amsterdam.

Venette, R. C., E. E. Davis, M. DaCosta, H. Heisler and M. Larson. 2003. Mini risk assessment, false codling moth, *Thaumatotibia* (=*Cryptophlebia*) *leucotreta* (Meyrick) [Lepidoptera: Tortricidae]. USDA/APHIS/PPQ Pest Risk Assessment. University of Minnesota, St. Paul, Minnesota. 30 pp.

Walsingham, Lord [de Grey, T.] 1899. Descriptions of two new species of Tineina from Bengal. Indian Museum Notes. 4: 105–107.

Williams, J. R. 1953. The larvae and pupae of some important Lepidoptera. Bulletin of Entomological Research. 43: 691–701.

Zacher, F. 1915. Beiträge zur Kenntnis der Westafrikanischen Planzenschädlinge. Tropenpflanzer. 18: 504–534.

Chapter 6

A new avocado pest in Central America (Lepidoptera: Tortricidae) with a key to Lepidoptera larvae threatening avocados in California

Introduction

Cryptaspasma Walsingham is almost exclusively pan-tropical, comprising 34 described species (Brown 2005). It has been recorded from Central America, South America, southern North America, Africa, Madagascar, Australia, New Zealand, New Caledonia, the Orient, and the eastern Palearctic (Horak 2006). The genus has traditionally been considered the sole representative of the tribe Microcorsini (Diakonoff 1959, Horak and Brown 1991, Horak 1999); however, the Australian genus *Collogenes* Meyrick was recently transferred to the Microcorsini by Horak (2006). Microcorsini are hypothesized to be the most basal group in Olethreutinae (Razowski 1976, Horak 2006), and this is supported by recent phylogenetic analyses of the Tortricidae using molecular data (Regier et al. 2012). The genus is currently divided into seven subgenera based on geographic distribution and structures of the male genitalia; Aarvik (2005) provides a key to the subgenera.

Species concepts and subgeneric classification in *Cryptaspasma* are poorly resolved, with Diakonoff (1959) providing the only complete revision of the genus to date. In addition to describing new species and illustrating type specimens, he defined five subgenera based primarily on male genitalic structures and geographic distribution. Kuznetsov (1970) proposed

---

*This chapter is published as:

Gilligan, T. M., J. W. Brown & M. S. Hoddle. 2011. A new avocado pest in Central America (Lepidoptera: Tortricidae) with a key to Lepidoptera larvae threatening avocados in California. Zootaxa. 3137: 31-45.

the tribe Microcorsini and elevated the five *Cryptaspasma* subgenera to genus rank. Subsequent authors have followed Diakonoff (1959) rather than Kuznetsov (1970) and treated the subgenera as such, although the taxonomic rank of these groups is largely subjective (Aarvik 2005) and questionable (Tuck, pers. comm.). Several new species were described in the later part of the 20th century from islands in the western Pacific and Indian Oceans (Clarke 1976, Bradley 1982, Diakonoff 1983). Brown and Brown (2004) described a new species of *Cryptaspasma* from the southeastern U.S. and provided the first complete world catalogue for the genus. Aarvik (2005) revised the African species of *Cryptaspasma* and proposed two new subgenera, increasing the total to seven. Horak (2006) revised the Australian *Cryptaspasma* and transferred *Collogenes* to Microcorsini. Most recently, Razowski (2011) reviewed the Neotropical Microcorsini and described a new species of *Cryptaspasma* from Costa Rica.

An undescribed species of *Cryptaspasma* was brought to the attention of the USDA in 2002, when larvae discovered in Hass avocados in Michoacán, México were thought to be that of *Stenoma catenifer* Walsingham (Elachistidae). The larvae were identified as those of an undescribed *Cryptaspasma* species (mentioned in Brown and Brown 2004). In 2006–2007, visits to Guatemala confirmed the discovery of an undescribed species of *Cryptaspasma* infesting avocado fruit (detailed in Hoddle and Hoddle 2008, Hoddle 2011). Examination of the Mexican and Guatemalan specimens verified that they are conspecific, supported by both morphology and mtDNA analysis. The presence of a tortricid avocado pest in Central America and Mexico (Hoddle and Brown 2010) raises quarantine issues given the recent lifting of a U.S. import ban on Mexican Hass avocados.

This new species of *Cryptaspasma* from Guatemala and Mexico is described here. Morphological details of adults and immature stages are presented, along with details of its biology and life history, and characters for separating it from other tortricids and avocado-feeding Lepidoptera threatening avocado in California. In addition, a complete list of tortricids documented as feeding on avocado is presented.

Materials and Methods

Specimens were examined and dissected using a Wild M5A stereomicroscope (Wild Heerbrugg AG, Switzerland). Genitalia dissection methodology follows that summarized in Brown and Powell (1991) except that some preparations were mounted using Euparal mounting medium (Bioquip Products, Rancho Dominguez, CA). Larvae were prepared using the following method (O. Sage, pers. comm.): a small incision was made on the ventral mid-line near the head and the intact larva heated in 10% KOH for approximately 15 minutes; the larva was placed in water and the incision extended the full length of the body; the head capsule was carefully removed using forceps; the internal contents of the larva were removed with a brush and the cleaned larval skin stained lightly with chlorazol black; the skin was placed under glass pieces in a petri dish with 100% ethanol and left to dehydrate for one hour; the skin was then mounted in Euparal on a microscope slide; the head was stored in 80% ethanol. Adult, larval, and pupal photographs were taken using a Canon EOS 40D digital SLR (Canon U.S.A., Lake Success, NY) mounted on a Visionary Digital BK Lab System (Visionary Digital, Palmyra, VA). Microscope slide photos were taken using a Nikon DXM1200 digital camera mounted on a Nikon Labophot2 compound microscope (Nikon Instruments, Melville, NY). Some photographs are a combination of several layers produced with Helicon Focus 4.80 (Helicon Soft Ltd., Kharkov, Ukraine). All photographs were edited using Adobe Photoshop CS3 Extended and drawings were produced using Adobe Illustrator CS3 (Adobe Systems Inc., San Jose, CA).

Morphological terms and wing pattern descriptions follow that in Diakonoff (1959), Aarvik (2005), Horak (2006), and Gilligan et al. (2008). The weakly sclerotized posteriorly projecting flap on the median portion of the male valva is referred to as a pulvinus (*sensu* Horak 2006) – “a pad-shaped densely bristled basal lobe.” The shape of the scales in the coremata on the eighth abdominal tergite is referred to as rhopaloid (*sensu* Diakonoff 1959) – “distally enlarged or inflated.”

Abbreviations and symbols are as follows: AR = forewing aspect ratio, calculated by dividing forewing length by medial forewing width; ca. = circa (approximately); FW = forewing; FWL = forewing length, measured from base to apex including fringe; HW = hindwing; n = number of specimens examined. Abbreviations for depositories are as follows: CSCA, California State Collection of Arthropods, California Department of Food and Agriculture, Sacramento, Calif.; UCR, University of California, Riverside, Calif.; USNM, National Museum of Natural History, Smithsonian Institution, Washington, D.C.

Description

Cryptaspasma **(*Cryptaspasma*)** ***perseana*** **Gilligan and Brown, new species**

(Figs. 73–76)

Systematics. *Cryptaspasma perseana* is assigned to the subgenus *Cryptaspasma* based on the following combination of characters (Aarvik 2005): uncus with hair-pencil; valva not swollen; pulvinus without spike(s); sacculus of valva without triangular, pointed prominence; and outer edge of valva not emarginate, thus sacculus and cucullus undifferentiated.

Diagnosis. *Cryptaspasma perseana* is separated from other species in the subgenus *Cryptaspasma* by the following characters: *C. bipenicilla* Brown and Brown has two lengths of coremata on the male abdomen versus one length in *C. perseana*; *C. lugubris* (Felder & Rogenhofer) and *C. acrolophoides* Meyrick males have an uncus that is parallel-sided versus widened above the base and tapering distally in *C. perseana*; *C. athymopis* Diakonoff and *C. microloga* Diakonoff females have signa that taper to a dull point versus signa that are distally rounded in *C. perseana*. Other undescribed males in the subgenus *Cryptaspasma* from Central and South America differ from *C. perseana* in having either two lengths of coremata, setae absent from the base of the valva and/or sex scaling on the male hindwing; females of undescribed species generally lack the ventrolateral triangular processes on the sterigma and/or have differently shaped signa.

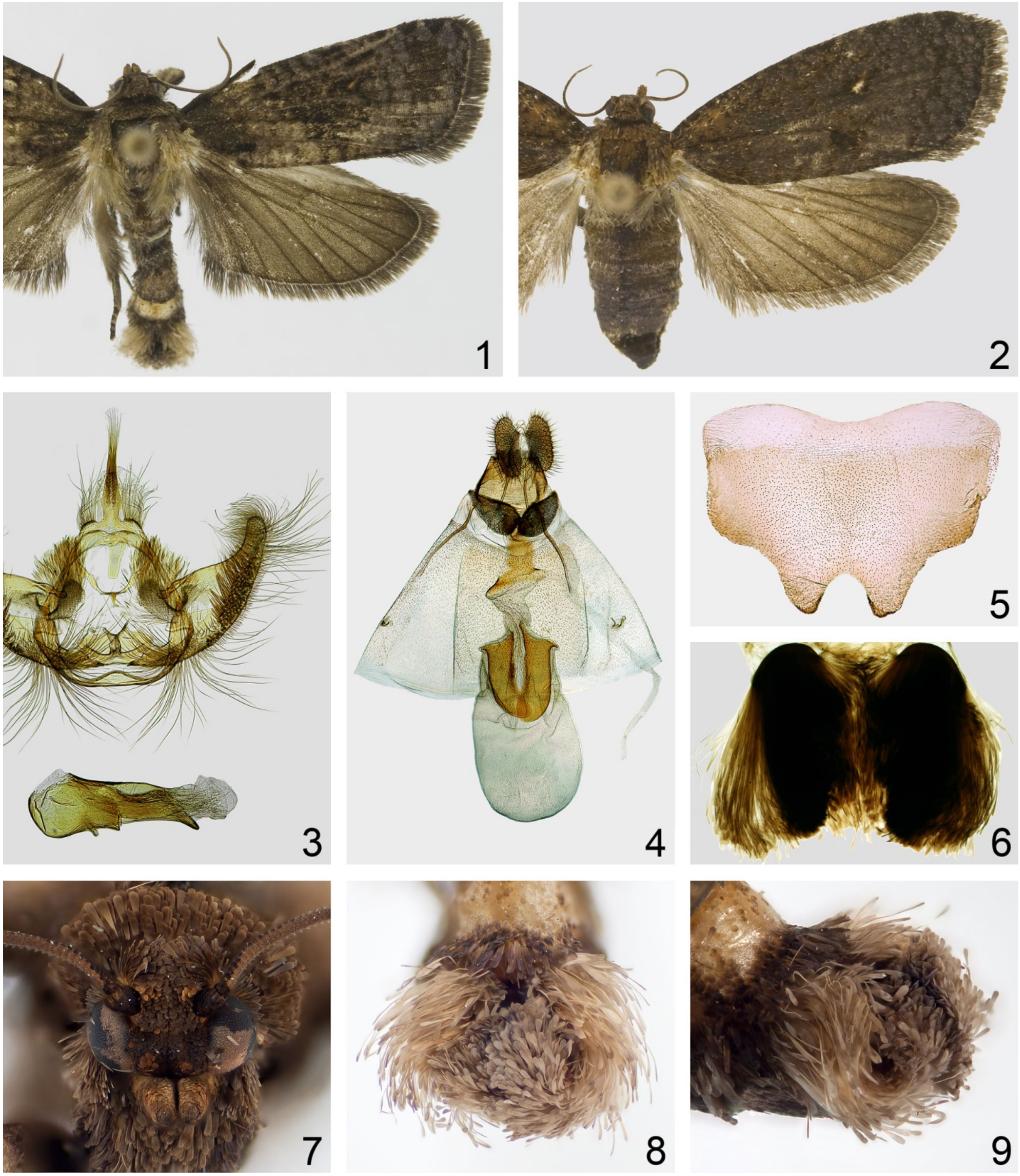


Figure 73. *C. perseana* adult characters; 1: male holotype; 2: female; 3: male genitalia; 4: female genitalia; 5: male eighth abdominal sternite; 6: male coremata, slide mounted; 7: head; 8: male coremata, posterior aspect; 9: male coremata, lateral aspect.

Description. Adult. Male. *Head* (Fig. 73.7): Vertex rough scaled, dark brown intermixed with golden-brown; upper portion of frons rough scaled, concolorous with vertex, lower portion of frons smooth scaled, dark brown to black intermixed with orange-brown; labial palpus porrect, all segments combined ca. 1.3 times diameter of compound eye, first and second segments golden brown, second segment enlarged, third segment rounded, ca. 0.3 as long as second segment, dark brown distally; antenna ca. 0.4 as long as forewing, dark brown basally, brown apically, scape dark brown to black, sensory cilia ca. 0.2 times width of flagellomere; ocellus conspicuous. *Thorax* (Fig. 73.1): Dorsal and ventral surfaces brown; tegula covered with long pale-brown scales; fore leg densely covered with dark brown scales; mid- and hind femur with long broad brown scales on ventral surface; mid- and hind tibia with long hairlike brown scales on dorsal and ventral surfaces; metathorax with posterior pair of scale tufts composed of long thin pale brown scales. Forewing length 9.8–10.9 mm (mean = 10.3, n = 10); costal fold absent; costal margin weakly convex; ground color pale brown intermixed with black and grayish purple; costal strigulae present as paired or single pale brown marks, strigulae 3–9 paired or unpaired on costa, strigula 10 unpaired between R_4 and R_5 on apex; conspicuous orange spot between bases of M_1 and M_2; fasciae dark brown to black, poorly defined; basal fascia expressed as black scaling at base along dorsum; subbasal and median fasciae coalesced to form broad band of black scaling extending from costa to dorsum across discal cell; postmedian and preterminal fasciae undefined, apical half of wing variably mottled with dark brown and grayish-purple; fringe brown. Hindwing: Uniformly brown; fringe brown with lighter apices. *Abdomen* (Figs. 73.3, 73.5–6, 73.8–9, 74.10): Brown to pale brown; eighth abdominal tergite with posterior pair of coremata (Figs. 73.6, 73.8–9, 74.10) consisting of a single tuft of scales composed as follows: outer scales elongate and pale brown, middle scales rhopaloid and dark brown, inner scales larger, rhopaloid, and covered with secretions; eighth abdominal sternite (Fig. 73.5) tapered distally with large mesal excavation forming two distal lobes. Genitalia (Fig. 73.3) with uncus variably elongate, weakly widened above base and tapered towards apex with tuft of setae extending from distal half, setal tuft approximately same length as uncus; socii a pair of setose arched lobes laterad to uncus; gnathos a narrow band with triangular subscaphium;

Figure 74. SEM of male coremata scales.

tegumen narrow; pedunculus a triangular lobe; valva triangular, covered in long thin setae concentrated at base and rounded apex; a row of small pointed spinelike setae running entire length of saccular margin projecting ventrally; sacculus with several rows of long blunt spinelike setae ca. 0.50–0.85 distance from base to apex of valva; median portion of valva with weakly sclerotized pulvinus densely covered in long rhopaloid scales; phallus ca. 0.50 length of valva, broad, tapering weakly to blunt apex; cornuti absent.

Female. *Head*: As in male. *Thorax* (Fig. 73.2): As in male except: Forewing length 10.5–12.1 mm (mean = 11.3, n = 9); costal margin weakly convex; ground color grayish brown; costal strigulae subdued, remnants of strigulae 5–9 visible in some individuals; conspicuous white and orangish-brown spot between bases of M_1 and M_2; fasciae undefined; basal half of wing infused with orangish-brown, apical half of wing variably mottled with dark brown and grayish-purple;

black patch surrounded by orangish-brown in area of median fascia below discal cell; fringe brown. Hindwing: Uniformly brown; fringe brown. *Abdomen* (Fig. 73.4): Brown to pale brown, without modified scales. Genitalia with papillae anales broad and densely setose; apophyses anteriores and posteriores slender, anteriores ca. 1.6 times as long as posteriores; sterigma composed of a pair of ventrolateral, slightly convex, triangular processes separated posteriorly by a ventromedial gap, each process strongly and finely reticulated with cellular spaces bordered by walls and covered with fine spicules, posteromedial corners of processes with several long, thin setae; ductus bursae widened asymmetrically at middle and sclerotized on ventral side from antrum to middle of ductus, forming a ring encircling ductus at middle posterior to inception of ductus seminalis; bulla seminalis absent; corpus bursae large, pear-shaped, with reticulated wall; a pair of large, hollow, paddle-shaped signa, nearly equal in size, with base of right signum arising more posterad than that of left.

Egg (Figs. 76.18–19). Flat, oval, translucent, white when first laid, turning grey as larva develops. Eggs are laid in masses (described below).

Larva (Figs. 75.11–17, 76.21–22). Mandibles (Fig. 75.13) with inner teeth (sensu Passoa 1985) modified into long ridge with tooth at the distal end, retinaculum present as a triangular projection; distance between P1 and AF2 on head (Fig. 75.12) ca. 0.7 times distance between P1 and P2; a horizontal line connecting the AF2 setae on head (Fig. 75.12) passes closer to P2 than P1; all pinacula weakly sclerotized; L pinaculum on T1 (Fig. 75.11) anteriad to spiracle; SV groups on A1, 2, 7, 8, 9 with 3:3:3:2:2 setae; SD2 on A1–8 (Fig. 75.14) reduced, located on same pinaculum as SD1; SD pinaculum on A8 (Fig. 75.11) anteriad to spiracle; D2 setae on A8 (Fig. 75.15) on same “saddle” pinaculum and closer together than D1 setae; D1, D2, and SD1 setae on A9 (Fig. 75.17) all on same large “saddle” pinaculum; D1 and SD1 setae on A9 (Fig. 75.17) closer together than D2 and D1 setae; L pinaculum on A9 (Fig. 75.11) bisetose; distance between D1 setae on anal shield (Fig. 75.16) ca. 1.7 times distance between D1 and SD1 setae; anal comb absent.

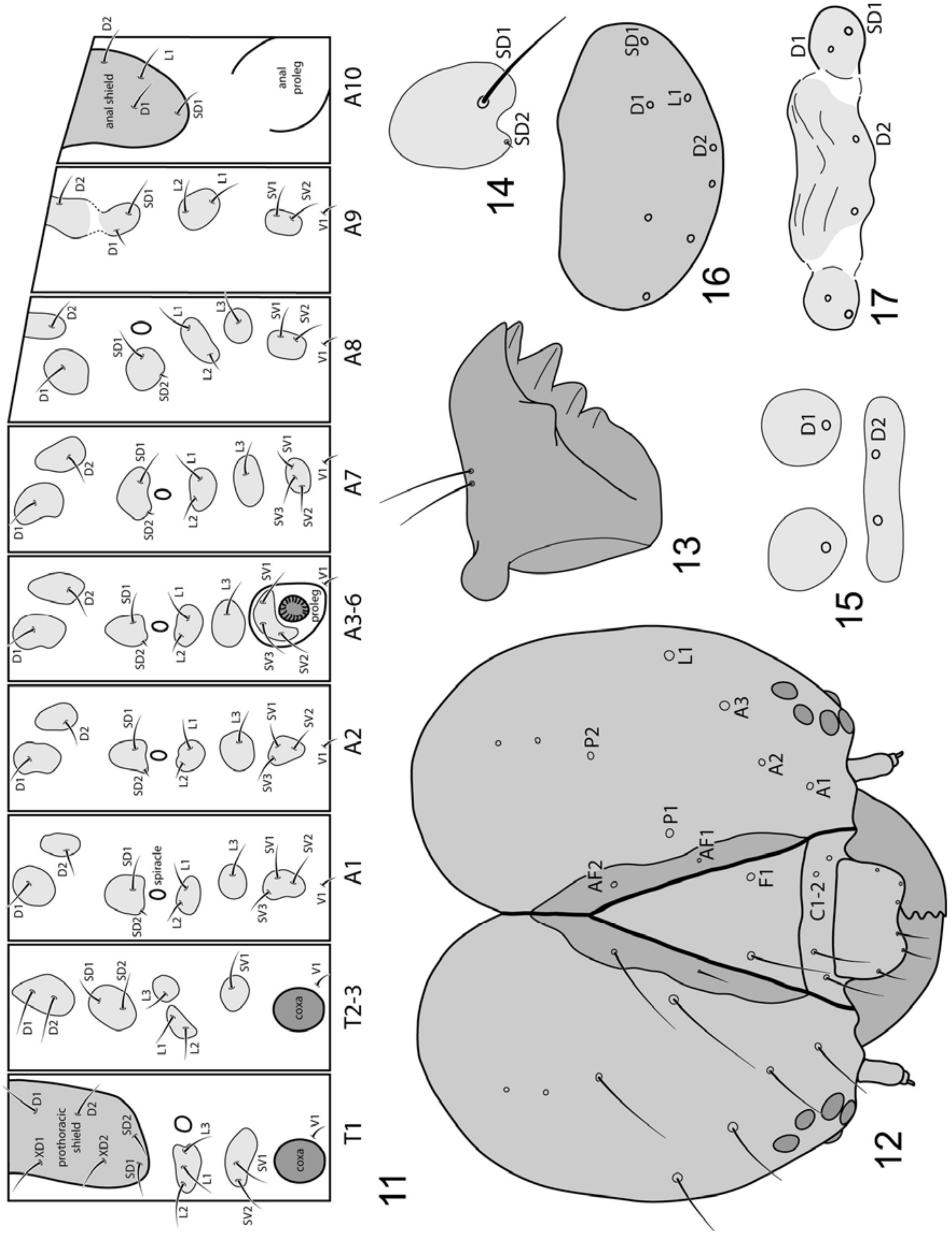


Figure 75. *Cryptaspasma perseana* larval morphology; 11: complete setal map; 12: setal map of head; 13: mandible; 14: SD pinaculum on A1–8; 15: D1 and D2 setae on A8; 16: anal shield; 17: D1, D2, and SD1 setae on A9.

Pupa (Figs. 76.23–26). Length 9.0–11.6 mm (mean = 10.6, n = 13); abdominal segments 3-8 with two dorsal rows of spines: anterior row larger with ca. 18-32 spines; posterior row smaller with ca. 25-42 spines.

HOLOTYPE: ♂: GUATEMALA, Sacatepéquez, Finca San Miguel Urias, San Miguel Dueñas, 1495m, 14°31’27” N, 90°46’34” W, ex. Hass avocado fruit, XII 2006-III 2007, M. & C. Hoddle, USNM.

PARATYPES: 40 ♂, 60 ♀: GUATEMALA, same data as for holotype (4 ♂ CSCA; 1 ♂ TMG; 5 ♂ UCR; 8 ♂ USNM, genitalia slides USNM 124,660, TMG 488, TMG 489; 6 ♀ CSCA; 1 ♀ TMG; 5 ♀ UCR; 8 ♀ USNM, genitalia slides USNM 124,661, TMG 491); Sacatepéquez, Palin, Finca El Recuerdo, 1390m, 14°26’07” N, 90°40’52” W, ex. Hass avocado fruit, 8 XI 2006-16 XII 2006, M. Hoddle (5 ♂ UCR; 1 ♂ USNM, genitalia slide USNM 124,475; 6 ♀ UCR); Sacatepéquez, San Pedro de las Huertas, 14°31’55” N, 90°44’4” W, ex. Non-Hass avocado fruit, lab-reared, II-III 2007, M. Hoddle (15 ♂ UCR; 30 ♀ UCR); MÉXICO, Michoacán, near Morelia, Nuevo Parangaricutiro, Huerta Ladera 3, ex-larva, 7 XI 2002, Luis Cervantes (1 ♂ USNM, genitalia slide TMG 492; 4 ♀ USNM, genitalia slide TMG 493).

Etymology. The specific name is derived from the larval host, *Persea americana*.

Distribution. *Cryptaspasma perseana* is recorded from Michoacán, México and central Guatemala.

Biology. Hoddle and Hoddle (2008) described the biology of *C. perseana* (as “*Cryptaspasma* sp. nr. *lugubris*”) reared under laboratory conditions; their findings are summarized here. Females laid eggs in masses (Figs. 76.18–19) containing an average of approximately 21 individual eggs (range of 2 to 73 eggs), primarily on smooth plastic surfaces, although some eggs were laid directly on intact avocado fruit and exposed avocado seeds. Eggs hatched in approximately 11 days (at 22°C) and larvae bored into avocado seeds where they completed development. A single avocado seed supported between 5–8 individual larvae. Larvae completed development in approximately three weeks (at 22°C) and left the seeds in

Figure 76. *Cryptaspasma perseana* immature stages; 18–19: egg masses; 20: larval damage to avocado fruit; 21–22: larvae; 23–25: pupae; 26: pupa exuvium extruding from avocado; 27: adult resting on avocado.

search of pupation sites, although 5% of larvae pupated within feeding tunnels in the seed (Fig. 76.20). Larval damage was typically characterized by feeding tunnels in seeds, damaged pulp, and accumulation of frass at the opening of feeding tunnels. In the laboratory, larvae pupated between layers of paper towels; we hypothesize that pupation occurs primarily under bark or in leaf litter in natural settings. Larvae remained in a prepupal stage for approximately 2–4 days and in the pupal stage for 8–12 days (at 22°C). The pupal exuvium extrudes from the pupal chamber upon adult eclosion (Fig. 76.26). Adult sex ratio was found to 46:54 male:female under laboratory conditions.

Adult females use pheromones to attract males. Pheromone gland extracts have been made, potential pheromone compounds isolated and identified, but field testing has not been conducted (Millar and Hoddle unpublished). Female calling was observed in the laboratory, characterized by rapid wing vibrations and a slight upturn of the abdomen with large pheromone glands protruding from the posterior. Calling was observed to commence almost immediately following sunset and lasted for approximately one hour, after which all obvious locomotor activity associated with mating by males and females ceased. At sunrise, adult moths rapidly moved to dark concealed hiding places at ground level to rest. When provided with access to a mix of 10% honey and water, adults lived for up to seven days (at 22°C).

It is unclear if females prefer to oviposit on fresh fruit still on the tree or on intact fruit that has recently fallen to the ground. Brown and Brown (2004) hypothesized that *Cryptaspasma* species are specialists on hard seeds of fallen fruit, suggesting that females do not oviposit on fruit that remains on the tree. Hoddle and Hoddle (2008) reviewed this issue and concluded that, based on field observations of dropped fruit, the likelihood of dropped avocado fruit being consumed by animals in a few days was very high. Thus any eggs laid by *Cryptaspasma* females on freshly dropped fruit would be eaten by animals as would any larvae that had not yet tunneled into the seed. Because Hoddle and Hoddle (2008) reared *C. perseana* from intact green fruit that was stated to be freshly picked from trees by a vendor (this was corroborated by circumstantial evidence; the fruit, at time of purchase were in a net sack on the vendor’s back and the vendor was carrying a fruit picking pole), they suggest that oviposition and subsequent larval boring to the seed could have occurred before intact fruit dropped to the ground. Fruit may drop because of larval feeding, or because of strong winds causing fruit drop independently of larval infestation.

One species of endoparasitoid, a *Pseudophanerotoma* sp. (Hymenoptera: Braconidae: Cheloninae), was reared from ca. 30% of field collected *C. perseana* larvae. This is the first host record for a *Pseudophanerotoma* species; members of the Cheloninae are generally egg and larva parasitoids of tortricoids and pyraloids (J. Luhman pers. comm.).

Additional color photographs of all life stages of *C. perseana* and *Pseudophanerotoma* sp. are available (Hoddle 2011).

Discussion

Avocado (*Persea americana* Miller) is a member of the Lauraceae native to México and Central America. This species has a long history of domestication, and over 1000 varieties of avocado are grown worldwide (UCR 2010). One of the most popular varieties is the Hass avocado, which is the primary commercial cultivar for major avocado producers in California, México, and Central America (Hoddle and Hoddle 2008). Worldwide, México has been the top avocado producer and the United States the top avocado importer for the past several years. It is estimated that yearly worldwide avocado production will reach 4.7 million tons worth over $7.5 billion retail dollars by 2012 (Market 2008).

The import of Hass avocados into the United States from México has been a controversial topic in recent years. Avocado fruit imports from México were banned under U.S. Federal law from 1914 to 2005 due to the risk of importing specialist avocado fruit pests into the United States (Lambert 2004). On January 31, 2005, Federal regulations were lifted, allowing import of avocadoes from Michoacán, México into all states except California, Florida, and

Hawaii for a period of two years; after January 31, 2007, avocados could be imported and distributed in all 50 States (Lambert 2004). The import of Mexican avocados into California that commenced in February 2007 was especially controversial, as Mexican imports competed with the California crop, which was valued at $327 million (U.S.) during the 2007–2008 growing season (CAC 2008, Hoddle and Brown 2010). In addition to economic impacts, import of avocados from other countries into the United States increases the risk of introducing avocado-feeding pests, especially internal-feeding species of Lepidoptera, Coleoptera, and Diptera. Although the number of leaf-feeding avocado pests has increased in California over the past two decades, the ban on avocado imports from México may have been significant in preventing the establishment of internal fruit-feeding pests (Hoddle and Hoddle 2008).

Worldwide, nearly 100 species of Lepidoptera in 20 different families have been recorded from avocado fruit and/or foliage (HOSTS 2009, Hoddle and Brown 2010). Among these are 37 species of tortricids (Table 1), most of which appear to be generalists. The following is a summary of select Lepidoptera that either are known pests or represent potential pests of avocados in California.

Table 112: Tortricidae reported to feed on avocado

Genus/species	**Subfamily**	**Reference**	**Region/country**
"*Cnephasia*" *jactatana* (Walker)	Tortricinae	Stevens et al. 1995	New Zealand
Amorbia cuneana (Walsingham)	Tortricinae	LACM Index; Busck 1929; Faber et al. 2010; Waite & Barrera 2002; Wysoki et al. 2002	California
Amorbia emigratella Busck	Tortricinae	MacKay 1962; Zimmerman 1978; Coria et al. 2007	Hawaiian Islands, Mexico
Amorbia santamaria Phillips and Powell	Tortricinae	Hoddle & Brown 2010	Guatemala
Archips capsigerana (Kennel)	Tortricinae	Liu 1983; Liu & Li 2002	China
Archips machlopis (Meyrick)	Tortricinae	Yunus & Ho 1980	Malaysia
Archips micaceana (Walker)	Tortricinae	Lee & Winney 1981	Hong Kong
Archips occidentalis (Walsingham)	Tortricinae	Erichsen & Schoeman 1994	South Africa
Argyrotaenia amatana (Dyar)	Tortricinae	LACM Index; Freeman 1958	USA
Argyrotaenia franciscana (Walsingham) [1]	Tortricinae	LACM Index; Powell 1964b	USA
Argyrotaenia montezumae (Walsingham)	Tortricinae	USNM collection	Mexico
Argyrotaenia urbana (Busck)	Tortricinae	Hoddle & Brown 2010	Guatemala

Table 112: Tortricidae reported to feed on avocado

Genus/species	**Subfamily**	**Reference**	**Region/country**
Cacoecimorpha pronubana (Hubner)	Tortricinae	Swirski et al. 1995; Waite & Barrera 2002	Israel
Clarkeulia dimorpha (Clarke)	Tortricinae	d'Araujo Silva et al. 1968	Brazil
Cryptoptila immersana (Walker)	Tortricinae	Waite & Barrera 2002	Australia
Ctenopseustis herana (Felder and Rogenofer)	Tortricinae	Stevens et al. 1995; Waite & Barrera 2002	New Zealand
Ctenopseustis obliquana (Walker)	Tortricinae	Stevens et al. 1995; Waite & Barrera 2002	New Zealand
Epiphyas postvittana (Walker)	Tortricinae	Stevens et al. 1995; Brown et al. 2010	Australia, New Zealand
Homona spargotis Meyrick	Tortricinae	Pinese & Brown 1986	Australia
Isotenes miserana (Walker)	Tortricinae	Waite & Barrera 2002	Australia
Lozotaenia capensana (Walker)	Tortricinae	Erichsen & Schoeman 1994	South Africa
Netechma pyrodelta (Meyrick)	Tortricinae	Hoddle & Brown, 2010; USNM collection	Guatemala
Orthocomotis herbacea Clarke	Tortricinae	Clarke 1956; Brown 2003	Costa Rica
Planotortrix excessana (Walker)	Tortricinae	Stevens et al. 1995	New Zealand
Planotrotrix octo Dugdale	Tortricinae	Stevens et al. 1995	New Zealand
Platynota stultana Walsingham	Tortricinae	Powell 1983	North America
Cryptaspasma bipenicilla Brown & Brown	Olethreutinae	Brown & Brown 2004	Puerto Rico
Cryptaspasma perseana Gilligan & Brown	Olethreutinae	Brown & Brown 2004; Hoddle & Hoddle 2008; Hoddle & Brown 2010	Guatemala, Mexico
Lobesia stericta Meyrick	Olethreutinae	Erichsen & Schoeman 1994	South Africa
Sorolopha elaeodes temenopis (Meyrick) [2]	Olethreutinae	Meyrick 1936	Taiwan
Sorolopha phyllochlora (Meyrick) [3]	Olethreutinae	Meyrick 1936	Asia
Sorolopha semiculta (Meyrick) [4]	Olethreutinae	Meyrick 1936	Asia, Taiwan
Thaumatotibia leucotreta (Meyrick)	Olethreutinae	Bradley et al. 1979; Erichsen & Schoeman 1994	Europe
Thaumatotibia zophophanes (Turner)	Olethreutinae	Waite & Barrera 2002; Horak 2006	Australia
Histura perseavora Brown	Chlidanotinae	Brown & Hoddle 2010	Guatemala
Histuroides costaricana Razowski	Chlidanotinae	Brown & Hoddle 2010	Costa Rica
Polyortha Dognin [new species]	Chlidanotinae	Hoddle & Brown 2010	Guatemala

[1] as *Argyrotaenia citrana*; [2] as *Argyroploce temenopis*; [3] as *Olethreutes phyllochlora*; [4] as *Argyroploce heteraspis* in Taiwan

***Amorbia* spp. (Tortricidae: Tortricinae)**

Amorbia cuneana (Walsingham), the western avocado leafroller, is considered a primary but sporadic pest of avocado in California (Faber et al. 2010). Its putative synonym, *A. essigana* Busck, was described as an avocado pest in southern California nearly 80 years ago, and avocado feeding populations may actually represent this “pheromone race” (e.g., Hoffman et al. 1983; Bailey et al. 1986; Bailey et al. 1988). Adults are present from late April to July and again in September to November in central California and year round in southern California (Powell and Opler 2009). Larvae cause damage by feeding on leaves, skeletonizing them in early instars and consuming the entire leaf in later instars. Economic losses occur when larvae web leaves to fruit or feed on the skin of fruit in a cluster, causing cosmetic damage (Faber et al. 2010).

In Hawaii, the Mexican leafroller, *Amorbia emigratella* Busck, is considered a pest on a variety of plants, including avocado, although this species is native to México and Central America (Zimmerman 1978). Larval damage appears similar to that of *A. cuneana* and also *E. postvittana* (Zimmerman 1978).

An additional species, *Amorbia santamaria* Phillips & Powell, was reared from avocado in Guatemala by Hoddle and Hoddle (2008).

***Argyrotaenia franciscana* (Walsingham) (Tortricidae: Tortricinae)**

The orange tortrix, *Argyrotaenia franciscana*, is a polyphagous species that is an occasional pest of avocado in California. This species is found primarily in cooler coastal areas and river valleys, where adults may be present year round; only two generations are present in warmer inland areas (Powell 1964). Larvae feed in silken shelters on outer shoots and can cause economic damage by chewing holes in fruit and even causing fruit to drop by feeding on the stem (Faber et al. 2010).

***Cacoecimorpha pronubana* (Hübner) (Tortricidae: Tortricinae)**

The carnation tortrix, *Cacoecimorpha pronubana*, was discovered feeding on winter daphne (*Daphne odora* Thunb.) in California in early 2011 (M. Epstein, pers. comm.). A native of northern Africa, this species was first documented from North America in Oregon in 1964 (Powell 1969), and it is also present in Washington (E. LaGasa, pers. comm.). Larvae are highly polyphagous, having been recorded from more than 160 species of plants in 42 families

(Razowski 2002). Wyoski and Izhar (1976) reported *C. pronubana* as a pest of avocado in Israel in the early to mid-1970s, causing surface damage to fruit in orchards.

Epiphyas postvittana **(Walker) (Tortricidae: Tortricinae)**

The light brown apple moth, *Epiphyas postvittana*, native to Australia, was discovered in California in 2006 (Brown et al. 2010). Larvae are highly polyphagous, having been recorded on over 500 species of plants, including avocado (Brown et al. 2010). Although not yet recorded on avocado in California (M. Epstein, pers. comm.), larvae can cause economic damage by webbing leaves to fruit or feeding on the surface of fruit (Brown et al. 2010).

Caloptilia**,** ***Marmara*****, and** ***Phyllocnistis*** **spp. (Gracillariidae: Gracillariinae, Phyllocnistinae)**

Marmara gulosa Guillén and Davis, the citrus peelminer, has been reported as a pest of avocado in California (Waite and Barrera 2002; as *M. salictella* Clemens). Larvae cause damage by mining the surface of fruit, causing significant scarring, and they may also mine shoots and leaves (Guillén et al. 2001, Waite and Barrera 2002). *Caloptilia perseae* (Busck) is considered a pest of avocado in México, where larvae mine leaves but rarely cause significant damage (Wysoki et al. 2002). Davis and Wagner (2011) described several new species of *Phyllocnistis* from Central America and the southeastern U.S. that mine leaves of avocado; none of these species are recorded from California.

Platynota stultana **Walsingham (Tortricidae: Tortricinae)**

Platynota stultana, the omnivorous leafroller, was introduced into southern California in the late 1800’s (Powell and Opler 2009). It has subsequently expanded its range into northern California, while at the same time expanding its host range onto a variety of non-native plants (Powell 1983). This species is considered a pest in greenhouses and vineyards, but it also attacks row crops, citrus, and occasionally avocado. Adults are present year round in southern and central California. Larvae feed primarily in shelters constructed of rolled or folded leaves. Economic damage occurs when larvae web leaves to fruit or feed directly on fruit, causing superficial damage and secondary infection by bacteria and fungi.

Sabulodes aegrotata **(Guenée) (Geometridae: Ennominae)**

Sabulodes aegrotata, the omnivorous looper, is a highly variable and polyphagous species that sometimes causes damage to avocado, citrus, and walnuts (Powell and Opler 2009). In California, adults are present year round, with populations increasing in the warmer months. Larvae cause economic damage by feeding on both leaves and fruit, leading to disfigurement and scarring of fruit (Faber et al. 2010).

Stenoma catenifer **Walsingham (Elachistidae: Stenomatinae)**

Stenoma catenifer is the most important lepidopteran pest of avocado in the Neotropics (Wysoki et al. 2002). Although not yet found in California, this species has been recorded damaging avocados in Argentina, Brazil, Colombia, El Salvador, Guatemala, Guyana, Honduras, México, Panama, Perú, and Venezuela, with crop losses as high as 60–80% (Hoddle and Hoddle 2008). This pest has demonstrated the ability to invade new areas because of the accidental movement of infested avocado fruit. This situation was realized in 2000 in the Galápagos Islands (Landry and Roque-Albelo 2003). *Stenoma catenifer* larvae cause damage similar to those of *C. perseana*; larvae of the latter discovered in México in 2002 were initially thought to be those of *S. catenifer*. Larvae of the two species are separated by the following characters: D2 pinacula on A9 fused dorsally (creating a mid-dorsal saddle) in *C. perseana*, whereas D2 pinaculum and D1 pinaculum are fused subdorsally in *S. catenifer* (creating a shared subdorsal pinaculum); L pinaculum on A9 trisetose in *S. catenifer*, bisetose in *C. perseana*; and SV pinaculum on A1 and A7 bisetose in *S. catenifer*, trisetose in *C. perseana*. Cervantes (1999) provides a complete setal map and larval description for *S. catenifer*.

Thaumatotibia leucotreta **(Meyrick) (Tortricidae: Olethreutinae)**

The false codling moth, *Thaumatotibia leucotreta*, is a native of sub-Saharan Africa. This polyphagous species is a serious pest of citrus and cotton and has been recorded from more than 50 plant species in over 30 families (Brown et al. 2008, van der Geest et al. 1991). In South Africa, *T. leucotreta* is considered the most significant Lepidoptera pest of avocado (Erichsen and Schoeman 1994). Larvae cause direct damage by tunneling in the fruit just beneath the skin, and larval feeding often results in secondary damage caused by bacteria and fungi (Erichsen and Schoeman 1994).

Thaumatotibia leucotreta is not established in California; however, a single male was collected in a pheromone trap in Ventura County in July, 2008 (Gilligan et al. 2011). This species is one of the most commonly intercepted tortricids on pepper (Solanaceae: *Capsicum annuum* L.) and eggplant (Solanaceae: *Solanum melongena* L.) at U.S. ports-of-entry (Brown 2006, Gilligan et al. 2011).

Other Lepidoptera pests

Other species of Lepidoptera have been occasionally recorded on avocado in California (M. Epstein, pers. comm.); although there are no reports of other species reaching pest status in the state. Noctuidae, Arctiinae, Papilionidae, Hesperiidae, and Psychidae have been reported on avocado in México (Waite and Barrera 2002, USDA 2004); however, these species, where known, are not present in California. It is conceivable that any highly polyphagous species of Lepidoptera would feed on avocado if given an opportunity as many of the known pests are generalist feeders.

Key to Lepidoptera larvae threatening avocado in California

The following key, with characters modified from Gilligan and Epstein (2009), includes avocado-feeding species currently found in California and avocado-feeding species in other parts of the world that would threaten California avocado production should they become introduced. This is not a complete list of avocado-feeding Lepidoptera and the key is not intended to definitively key out all Lepidoptera larvae found on avocado. The purpose of the key is to assist identifiers and growers in recognizing the most commonly encountered avocado-feeding species in California and to help separate native from potentially harmful introduced species.

This key assumes that: 1) the larva was found feeding on avocado; 2) the larva is mid- to late instar; and 3) the user is familiar with larval morphology and terminology. Because many Lepidoptera larvae are difficult to identify to species, any identifications made with this key should be confirmed by an expert, and larvae should be reared to adulthood if possible to confirm larval identifications.

1. Larval damage apparent as mines in surface of fruit, leaves, or shoots .... Gracillariidae
1’. Larval damage not as above or unknown 2

2. Prothoracic prespiracular pinaculum bisetose .... 3 (macrolepidoptera and Pyraloidea)
2.’ Prothoracic prespiracular pinaculum trisetose .... 4 (microlepidoptera)

3. Prolegs present on abdominal segment 6 (A6) and A10 only... .... Geometridae (including *Sabulodes aegrotata*)
3.’ Prolegs present on A3-6 and A10 or A5-6 and A10... .... Other macrolepidoptera (including Noctuidae)

4. L1 and L2 setae on segments A2-7 close together or on same pinaculum .... 5 (Gelechioidea/Tortricidae)

4.’ L1 and L2 setae on segments A2-7 widely separated .... Other microlepidoptera

5. D2 setae on A9 on shared mid-dorsal (saddle) pinaculum; D2 and D1 pinacula not fused subdorsally 6

5.’ D2 setae on A9 on separate pinacula (no mid-dorsal saddle); D2 and D1 pinacula fused subdorsally .... *Stenoma catenifer*

6. Anal comb present; D1 and SD1 setae separate from D2 on A9 7

6.’ Anal comb absent; D2, D1, and SD1 setae all on same large pinaculum on A9 .... *Cryptaspasma perseana*

7. Prothoracic prespiracular pinaculum extended below spiracle .... *Thaumatotibia leucotreta*

7.’ Prothoracic prespiracular pinaculum not extended below spiracle 8

8. Prothoracic shield with dark lateral line .... *Amorbia* spp.

8.’ Prothoracic shield with irregular dark marks on posterolateral corners.............................. .... *Cacoecimorpha pronubana*

8.’’ Prothoracic shield without dark lateral line or other markings .... Other Tortricidae* (including *E. postvittana*, *P. stultana*, *A. franciscana*)

* It is difficult or impossible to separate larvae of the three tortricid species listed here without using advanced characters. See Gilligan and Epstein (2009) for additional characters that can be used to separate tortricid larvae in California.

References

Aarvik, L. (2005) Revision of African *Cryptaspasma* Walsingham, 1900 (Lepidoptera: Tortricidae). Norwegian Journal of Entomology. 51: 193–201.

Bailey, J. B., Hoffmann, M. P., McDonough L. M. and Olsen, K. N. 1988. Field-testing the sex pheromone for *Amorbia cuneana* in avocados. California Agriculture. 1988 May/June: 17–18.

Bailey, J. B., McDonough, L. M. and Hoffmann, M. P. 1986. Western avocado leafroller, *Amorbia cuneana* (Walsingham), (Lepidoptera; Tortricidae). Discovery of populations utilizing different ratios of sex pheromone components. Journal of Chemical Ecology. 12: 1239–1245.

Bailey, J. B., Olsen, K. N., McDonough, L. M. and Hoffmann, M. P. 1988. Possible new race of *Amorbia cuneana* discovered in avocado. California Agriculture. 1988 Sept./Oct.: 11–12.

Bradley, J. D. 1982. New species of Microlepidoptera from Norfolk Island. Journal of Natural History. 16: 367–379.

Brown, J. W. 2003. An illustrated guide to the *Orthocomotis* Dognin (Tortricidae) of Costa Rica, with summaries of their spatial and temporal distributional. Journal of the Lepidopterists' Society. 57: 253–269.

Brown, J. W. 2005. Tortricidae (Lepidoptera). In World Catalogue of Insects, Volume 5: 1–741.

Brown, J. W. 2006. Scientific names of pest species in Tortricidae (Lepidoptera) frequently cited erroneously in the entomological literature. American Entomologist. 52(3): 182–189.

Brown, J. W. and Powell, J. A. 1991. Systematics of the *Chrysoxena* group of genera (Lepidoptera: Tortricidae: Euliini). University of California Publications in Entomology 111. 87 pp.

Brown, J. W. and Brown, R. L. 2004. A new species of *Cryptaspasma* Walsingham (Lepidoptera: Tortricidae: Olethreutinae) from Central America, the Caribbean, and southeastern United States, with a catalogue of the world fauna of Microcorsini. Proceedings of the Entomological Society of Washington. 106: 288–297.

Brown, J. W., Robinson, G. and Powell, J. A. 2008. Food plant database of the leafrollers of the world (Lepidoptera: Tortricidae) (Version 1.0.0). http://www.tortricidae.com/foodplants.asp [Accessed 12 August 2010].

Brown, J. W., Epstein, M. E., Gilligan, T. M., Passoa, S. and Powell, J. A. 2010. Biology, identification, and history of the light brown apple moth, *Epiphyas postvittana* (Walker) (Lepidoptera: Tortricidae: Archipini) in California: an example of the importance of local faunal surveys to document the establishment of exotic insects. American Entomologist. 56(1): 34–43.

Brown, J. W. and Hoddle, M. S. 2010. A new species of *Histura* Razowski (Lepidoptera: Tortricidae: Polyorthini) from Guatemala attacking avocados (*Persea americana*) (Lauraceae). Proceedings of the Entomological Society of Washington. 112(1): 10–21.

Busck, A. 1929. A new avocado moth. Monthly Bulletin of the Department of Agriculture, State of California. 18: 276–277.

CAC (California Avocado Commission). 2008. Energizing the CA in CAlifornia avoCAdos. Annual Report 2007-2008, Irvine California. 21 pp.

Clarke, J. F. G. 1956. Neotropical moths in the genus *Orthocomotis* (Lepidoptera: Tortricidae). Transactions of the Royal Entomological Society of London. 107 (1955): 139–168.

Clarke, J. F. G. 1976. Microlepidoptera: Tortricoidea. Insects of Micronesia. 9: 1–144.

Coria, V. M., Aguilera, M., Vidales, A. and Muñoz, J. 2007. Contribution of the knowledge on Mexican leafrollers *Amorbia emigratella* Busck (Lepidoptera: Tortricidae) in avocado orchards in Michoacán, México. Proceedings of the World Avocado Congress VI, Viña Del Mar, Chile. [Abstract].

d'Araújo Silva, A. G., Gonçalves, C. R., Galvão, D. M., Gonçalves, A. J. L., Gomes, J., do Nascimento Silva, M. and de Simoni, L. 1968. Quarto catálogo do Insetos que vivem nas plantas do Brasil, seus parasitos e predadores. Ministério da Agricultura, Rio de Janeiro 2(1). 622 pp.

Davis, D. R. and Wagner, D. L. 2011. Biology and systematics of the New World *Phyllocnistis* Zeller leafminers of the avocado genus *Persea* (Lepidoptera, Gracillariidae). ZooKeys. 97: 39–73.

Diakonoff, A. 1959. Revision of *Cryptaspasma* Walsingham, 1900 (Lepidoptera, Tortricidae). Zoologische Verhandelingen (Leiden). 43: 1–60.

Diakonoff, A. 1983. Tortricidae from the Comoro Islands (Lepidoptera). Annales de la Société Entomologique de France (N. S.). 19: 55–68.

Erichsen, C. and Schoeman, A. S. 1994. South African Avocado Growers' Association Yearbook. 17: 109–112.

Faber, B. A., Morse, J. G. and Hoddle, M. S. 2010. UC IPM Pest Management Guidelines – Avocado. University of California IPM Online. http://www.ipm.ucdavis.edu/PMG/selectnewpest.avocado.html [Accessed 10 January 2011].

Freeman, T. N. 1958. The Archipinae fauna of North America (Lepidoptera: Tortricidae). The Canadian Entomologist, 90 (supplement 7). 87 pp.

Gilligan, T. M., Wright, D. J. and Gibson, L. D. 2008. Olethreutine moths of the midwestern United States, an identification guide. Ohio Biological Survey, Columbus, Ohio. 334 pp.

Gilligan, T. M. and Epstein, M. E. 2009. LBAM ID: Tools for diagnosing light brown apple moth and related western U.S. leafrollers (Tortricidae: Archipini). Center for Plant Health Science and Technology, USDA, APHIS PPQ, Raleigh, NC. [CD-ROM.]

Gilligan, T. M., Epstein, M. E. and Hoffman, K. M. 2011. Discovery of false codling moth, *Thaumatotibia leucotreta* (Meyrick), in California (Lepidoptera: Tortricidae). Proceedings of the Entomological Society of Washington. 113(4): 426–435.

Guillén, M., Davis, D. R. and Heraty, J. M. 2001. Systematics and biology of a new, polyphagous species of *Marmara* (Lepidoptera: Gracillariidae) infesting grapefruit in the southwestern United States. Proceedings of the Entomological Society of Washington. 103: 636–654.

Hoddle, M. S. 2011. *Cryptaspasma* sp. (Lepidoptera: Tortricidae: Olethreutinae) http://www.biocontrol.ucr.edu/Cryptaspasma/Cryptaspasma.html [Accessed 9 January 2011].

Hoddle, M. S. and Brown, J. W. 2010. Lepidoptera associated with avocado fruit in Guatemala. Florida Entomologist. 93: 649–650.

Hoddle, M. S. and Hoddle, C. D. 2008. Lepidoptera and associated parasitoids attacking Hass and non-Hass avocadoes in Guatemala. Journal of Economic Entomology. 101: 1310–1316.

Hoffman, M. P., McDonough, L. M. and Bailey, J. B. 1983. Field test of the sex pheromone of Amorbia cuneana (Walsingham) (Lepidoptera: Tortricidae). Environmental Entomology. 12: 1387–1390.

Horak, M. 1999. Tortricoidea, pp. 199–215. *In*: Kristensen, N. (ed.), Lepidoptera, moths and butterflies. Volume 1: Evolution, systematics, and biogeography. Handbook of Zoology 4 (35), Arthropoda: Insecta. Walter de Gruyter, Berlin & New York.

Horak, M. 2006. Olethreutinae moths of Australia (Lepidoptera: Tortricidae). Monographs on Australian Lepidoptera 10. 522 pp.

Horak, M. and Brown, R. L. 1991. 1.2 Taxonomy and phylogeny, pp. 23–48. *In*: van der Geest, L. P. S. and H. H. Evenhuis (eds.), Tortricid pests, their biology, natural enemies and control. Elsevier Science Publishers B. V., Amsterdam.

HOSTS 2009. HOSTS – A database of the world’s lepidopteran host plants. http://www.nhm.ac.uk/researchcuration/research/projects/hostplants/ [Accessed 12 August 2010].

Kuznetzov, V. I. 1970. New peculiar leaf-rollers (Lepidoptera, Tortricidae) from the Far-East of USSR. Entomologicheskoe Obozrenie. 49: 434–451. [In Russian].

LACM Index. Records from the card file at the Los Angeles County Museum of Natural History, Los Angeles, California; transcribed by Gaeden Robinson (BMNH). [Unpublished notes].

Lambert, C. D. 2004. Part IV, Department of Agriculture Animal and Plant Health Inspection Service 7 CFR Part 319, Mexican Avocado Import Program; Final Rule. Federal Register Rules and Regulations, 69 (229): Tuesday, November 30.

Landry, B. and Roque-Albelo, L. 2003. Presence of *Stenoma catenifer* Walsingham (Lepidoptera: Elachistidae: Stenomatinae), the avocado seed moth, in the Galápagos. Noticias de Galápagos. 62: 15–16.

Lee, L. H. Y. and Winney, R. 1981. Check list of agricultural insects of Hong Kong 1981. Hong Kong Agriculture and Fisheries Department Bulletin. 2: 1–164.

Liu, Y.-Q. 1983. Cochylidae and Tortricidae, pp. 26–56. *In*: Liu et al. (eds.), Iconographia Heterocerorum Sinicorum 1. Animal Research Institue of Chinese Academy of Sciences, Beijing. 439 pp.

Liu, Y.-Q. and Li, G. 2002. Tortricidae. Fauna Sinica (Insecta) 27. Science Press, Beijing. 463 pp.

MacKay, M. R. 1962. Larvae of the North American Tortricinae (Lepidoptera: Tortricinae). The Canadian Entomologist, Supplement 28: 1–181.

Market Research Analyst (Market). 2008. Avocado market forecast 2008-2012: Production, exports, imports. http://www.marketresearchanalyst.com/2008/01/14/avocado-market-forecast-2008-2012-production-exports-imports/ [Accessed 9 January 2011].

Meyrick, E. 1936. Exotic Microlepidoptera. 5: 24. 160 pp.

Pinese, B. and Brown, J. D. 1986. The avocado leafroller, a pest of increasing significance. Queensland Agricultural Journal (November-December). 1986: 289–292.

Powell, J. A. 1964. Biological and taxonomic studies on tortricine moths, with reference to the species in California. University of California Publications in Entomology. Vol. 32. 317 pp.

Powell, J. A., 1969. Occurrence of *Cacoecimorpha pronubana* (Hbn.) in Oregon. The Pan-Pacific Entomologist. 45(1): 70.

Powell, J. A. 1983. Expanding geographical and ecological range of *Platynota stultana* in California (Lepidoptera: Tortricidae). The Pan-Pacific Entomologist. 59: 233–239.

Razowski, J. 1976. Phylogeny and system of Tortricidae (Lepidoptera). Acta Zoologica Cracoviensia. 21: 73–120.

Regier, J. C., J. W. Brown, C. Mitter, J. Baixeras, S. Cho, M. P. Cummings and A. Zwick. 2012. A molecular phylogeny for the leaf-roller moths (Lepidoptera: Tortricidae) and its implications for classification and life history evolution. PLoS ONE. 7(4): e35574. doi:10.1371/journal.pone.0035574.

Stevens, P. S., McKenna, C. E. and Steven, D. 1995. Management for avocados in New Zealand. Proceedings of the World Avocado Congress III, 1995. 429–432.

Swirski, E., Wysoki, M. and Izhar, Y. 1995. Avocado pests in Israel. Proceedings of the World Avocado Congress III, 1995. 419–428.

University of California Riverside (UCR). 2010. Avocado Variety Database. http://www.ucavo.ucr.edu [Accessed 26 July 2010].

USDA. 2004. Importation of avocado fruit (*Persea americana* Mill. var. 'Hass') from Mexico. A Risk Assessment. U.S. Department of Agriculture, Animal and Plant Health Inspection Service, Plant Protection and Quarantine, Center for Plant Health Science and Technology. [Unpublished report].

Waite, G. K. and Barrera, M. 2002. Insect and mite pests, pp. 339–361. *In*: Whiley, A. W., Schaffer, B. and Wolstenholme, B. N. (eds.), The avocado: botany, production and uses. CAB International.

Wyoski, M. and Izhar, Y. 1976. The carnation leaf-roller *Cacoecimorpha* (*Cacoecia*) *pronubana* Huebner (Lepidoptera, Tortricidae) on avocado trees in Israel. California Avocado Society Yearbook. 60: 92–95.

Wysoki, M., van den Berg, M. A., Ish-Am, G., Gazit, S., Peña, J. E. and Waite, G. K. 2002. Pests and pollinators of avocado, pp. 223–294. *In*: Peña, J. E., Sharp, L. J. and Mysoki, M. (eds.), Tropical fruit pests and pollinators, biology, economic importance, natural enemies and control. CAB International.

Yunus A. and Ho, T. H. 1980. List of economic pests, host plants, parasites and predators in West Malaysia (1920-1978). Bulletin of the Malaysian Department of Agriculture. 153: 1–538.

Zimmerman, E. C. 1978. Insects of Hawaii, Volume 9, Microlepidoptera, Part 1. University of Hawaii Press, Honolulu, Hawaii. 881 pp.

Chapter 7

A sequence search tool for the identification of Lepidoptera through DNA barcoding*

Introduction

DNA barcoding, or the use of a standardized segment of DNA for species identification and discrimination, is a widely accepted procedure that has been applied in different formats to many fields in the biological sciences. Hebert et al. (2003) were some of the first authors to advocate using a standardized gene region to identify species across all of life: the "Folmer region" at the 5-prime end of the cytochrome c oxidase 1 mitochondrial region (abbreviated COI or cox1). They suggest that DNA barcoding could be applied to solve major taxonomic problems, including the identification of known species and the discovery of new species, thus solving the biodiversity crisis (Hebert et al. 2003). Proponents of DNA barcoding have proposed other ambitious goals, such as creating a barcode library for all eukaryotic life in the next 20 years (Ratnasingham and Hebert 2007). Such aspirations have resulted in the creation of the Consortium for the Barcode of Life (CBOL, www.barcoding.si.edu) with 170 member

---

*Portions of this chapter are published in:

Gilligan, T. M. & M. E. Epstein. 2009. LBAM ID: Tools for diagnosing light brown apple moth and related western U.S. leafrollers (Tortricidae: Archipini). CD-ROM. Center for Plant Health Science and Technology, USDA/APHIS/PPQ, Raleigh, NC.

Gilligan, T. M. & M. E. Epstein. 2012. TortAI, Tortricids of Agricultural Importance to the United States (Lepidoptera: Tortricidae). CD-ROM. Identification Technology Program (ITP), USDA/APHIS/PPQ/CPHST, Fort Collins, CO.

organizations in more than 50 countries, more than $80 million in funding for DNA barcoding in Canada alone (Kwong et al. 2012), and more than $150 million in requested funding to support initiatives such as the International Barcode of Life (iBOL, www.ibolproject.org).

Numerous publications have been authored demonstrating the usefulness (e.g., Hebert et al. 2004a, Hebert et al. 2004b, Janzen et al. 2005, Ward et al. 2005, Hajibabaei et al. 2006) or outlining the downfalls (e.g., Moritz and Cicero 2004, Will and Rubinoff 2004, Will et al. 2005, Brower 2006, Rubinoff 2006, Rubinoff et al. 2006a, Rubinoff et al. 2006b, Sundberg et al. 2010, Kwong et al. 2012) of DNA barcoding. Many critics of DNA barcoding in its current form (i.e., as defined by Hebert et al. 2003 and implemented through www.boldsystems.org) point to problems with novel species delimitation based on percent threshold values (Will and Rubinoff 2004, Rubinoff et al. 2006a, Virgilio et al. 2010), or to various other problems related to using mitochondrial DNA to define species, including: introgression, recombination, maternal inheritance, heteroplasmy, infection by endosymbiotic bacteria, and nuclear mitochondrial pseudogenes (Rubinoff et al. 2006a, Song et al. 2008, Virgilio et al. 2010).

Despite these shortcomings, even opponents of barcoding conclude that “if DNA barcoding limited itself to [the identification of species previously defined by other criteria] it would be relatively uncontroversial” (Rubinoff et al. 2006a). It is this specific application of DNA barcoding that can be used to assist in making regulatory decisions by Federal or State agencies related to the protection of agriculture from pests and diseases. In the United States, the U. S. Department of Agriculture Animal and Plant Health Inspection Service regulates “organisms and products that are known or suspected to be plant pests or to pose a plant pest risk” (USDA-APHIS, www.aphis.usda.gov). Regulation refers to the detection, quarantine, management, and control of these pests and/or agricultural products infested by these pests. Often regulatory decisions involving infested agricultural products result in a substantial cost to the grower, producer, importer, or exporter in the form of treatment, loss of product by rejection or halt of production, or fines. As a result, careful and accurate identification of pest species in every life stage is needed to support such decisions. DNA barcoding has the ability to provide or corroborate such identifications if implemented in a proper manner.

The application of DNA barcoding to the identification of invasive or quarantine pests has been demonstrated by several authors (e.g., Barcenas et al. 2005, Ball and Armstrong 2006, Nagoshi et al. 2011, Scheffer et al. 2006). Scheffer et al. (2006) had success in differentiating different species of invasive leafmining Diptera in the Philippines using hierarchical clustering of DNA barcodes. Overall intraspecific genetic p-distances varied from 0% to 1.9% while interspecific p-distances were greater than 8%; however the study failed to separate a “genetically and behaviorally distinct species” that varied less than 1% from its closest relative. Barcenas et al. (2005) constructed a diagnostic PCR test using a fragment of COI (112–116 bp) to separate four species of apple-feeding olethreutine moths (Tortricidae: Olethreutinae) that are pests on pome fruit (Rosaceae) in North America. The four pests could be successfully separated if several assumptions regarding the host, origin, and tribal placement of the specimens were satisfied. Ball and Armstong (2006) used DNA barcoding to successfully differentiate between 20 species of tussock moths (Noctuidae: Lymantriinae), including the highly destructive Gypsy moth, *Lymantria dispar* (Linnaeus). Nagoshi et al. (2011) used DNA barcoding to supplement morphological characters in separating two of the most destructive species of armyworm (Noctuidae): *Spodoptera littoralis* (Boisduval) and *Spodoptera litura* (Fabricius). Prior to using DNA data, identification of both targets and non-targets required time-consuming genitalic dissections for every specimen. Armstrong and Ball (2005) outlined several key attributes for developing a system to identify invasive species, including the ability to detect novel or unpredicted species, the desire to use consistent genes and protocols that can be replicated across laboratories, the necessity of incorporating the complete range of intraspecific variation via increased sampling into the database, and the need for taxonomists to accurately identify reference specimens and to describe newly discovered species.

Public barcode databases

Two large public databases currently house the majority of DNA barcodes: NCBI-GenBank and Barcode of Life Data Systems (BOLD). GenBank is a general sequence storage database, while BOLD is dedicated to storing only DNA barcodes. Both allow users to query their database using a sequence from an unknown individual in order to obtain an identification, although the type of query and amount of data available to a random user is vastly different in each system. The advantages and disadvantages of using either system to produce identifications used for making regulatory decisions are discussed in the following section.

NCBI-GenBank

GenBank, hosted by the National Center for Biotechnology Information, is the largest database of publically available DNA sequences on the internet. Nucleotide sequences are available for more than 300,000 named organisms (Benson et al. 2010), although COI sequences, including many that would not be of sufficient length to be considered full DNA barcodes by BOLD, are only available for approximately 60,000 species (Kwong et al. 2012).

The primary advantages of GenBank are its large size and the public availablility of all information in the database. All records, especially those formally designated as “DNA barcodes,” are heavily annotated and associated with a publication. The data set is very large, is comprised of many types of sequence records, and the entire data set can be searched via a variety of fast search tools that are accessible on the web. GenBank has been established for over 20 years and is funded by the National Institutes of Health (Benson et al. 2010).

Wholesale public data availablility is also one of GenBank’s primary disadvantages. The publically available database can be updated by anyone who correctly follows the submittal procedure. Errors in GenBank have been outlined by numerous authors (e.g., Korning et al. 1996, Pennisi 1999, Wesche 2004). Public submission of data also allows for the database to include sequences from incorrectly identified taxa and these problems are nearly impossible to detect and fix on a large scale. GenBank’s primary search tools, although fast and efficient, rely on the BLAST algorithm (Altschul et al. 1990, Johnson et al. 2008), which is not ideal for DNA barcode analysis because its similarity scores are optimized based on local alignments (but see Little and Stevenson 2007). Although it is expected that economically important species are “best represented in the reference databases” (Virgilio et al. 2010), this is actually not the case, as GenBank contains very few sequences of some quarantine pests, and even fewer DNA barcodes of those pests. For example, at the time of initial detection of the grape pest *Lobesia botrana* in California (September, 2009), there were no sequences of COI available for this species on GenBank (or BOLD) (Gilligan et al. 2011). Developing diagnostics with a public database is complicated, as the dynamic, constantly growing database makes repeatability difficult over time. The unregulated addition of new sequences creates problems for any protocols that rely on specific database content.

BOLD

Barcode of Life Data Systems, or BOLD, is the largest publically available database of DNA barcodes. The associated website (www.boldsystems.org) is described as “an informatics workbench aiding the acquisition, storage, analysis and publication of DNA barcode records” (Ratnasingham and Hebert 2007).

The primary advantage of BOLD for performing identification with barcode data is the number of DNA barcodes in the database. As of September, 2012, BOLD contained more than 1.7 million barcode sequences representing approximately 117,000 animals and 40,000 plants. Sequences are separated into “verified” and “unvalidated” categories, with “verified” sequences used for the default search library. Several search options are available, giving the user the ability to search all barcode records (including those that are unvalidated), all barcode records with a species-level identification, all public barcode records (those that have been published; includes GenBank records), and full length records, both public and private. The search tools implemented on BOLD are web-based, fast, and based on a Hidden Markov Model (HMM) used to construct a pairwise alignment that is appropriate for DNA barcode analysis (see Eddy 1996 for an overview of HMM algorithms). In contrast to GenBank, tighter control is maintained over the data placed in the database, with sequence submission requiring a prior arrangement between the researcher and BOLD.

Despite being a purpose-built DNA barcode library, there are several disadvantages when using BOLD for species identification. The most important issue is that the majority of the barcode records in the database are not publically available. As of September, 2012, the animal (COI) database contained a total of 1,395,901 sequences, with only 274,881, or less than 20%, of these listed in the public record database (numbers obtained from http://www.boldsystems.org/index.php/IDS_OpenIdEngine). Thus, more than 80% of the records in BOLD cannot be viewed by a random user performing a query, and it is impossible for a diagnostician to verify sequences or to check for errors if the system returns an identification using non-public data. Problems also arise when private sequence data is transferred to GenBank before being published. Because of an early data release agreement, BOLD is required to submit trace files, sequence records, and specimen data to GenBank within one week of sequence analysis (P. Hebert, pers. comm.). In order to ensure that the data remains private, a majority of these records are submitted to GenBank identified only to order with minimal information; Kwong et al. (2012) found that 74% of the DNA barcodes in GenBank were not identified to species. Further complicating matters, “several hundred thousand” of these barcode sequences were suppressed by GenBank because they did not satisfy the early release agreement (Kwong et al. 2012).

Problems are also evident within the design of BOLD, particularly in the fixed percentage cutoffs used to delimit species. In order to be “verified,” species in the search library must have a representative sample of at least three specimens with a maximum sequence divergence of 2% (Ratnasingham and Hebert 2007). This percentage is fixed across the entire database and

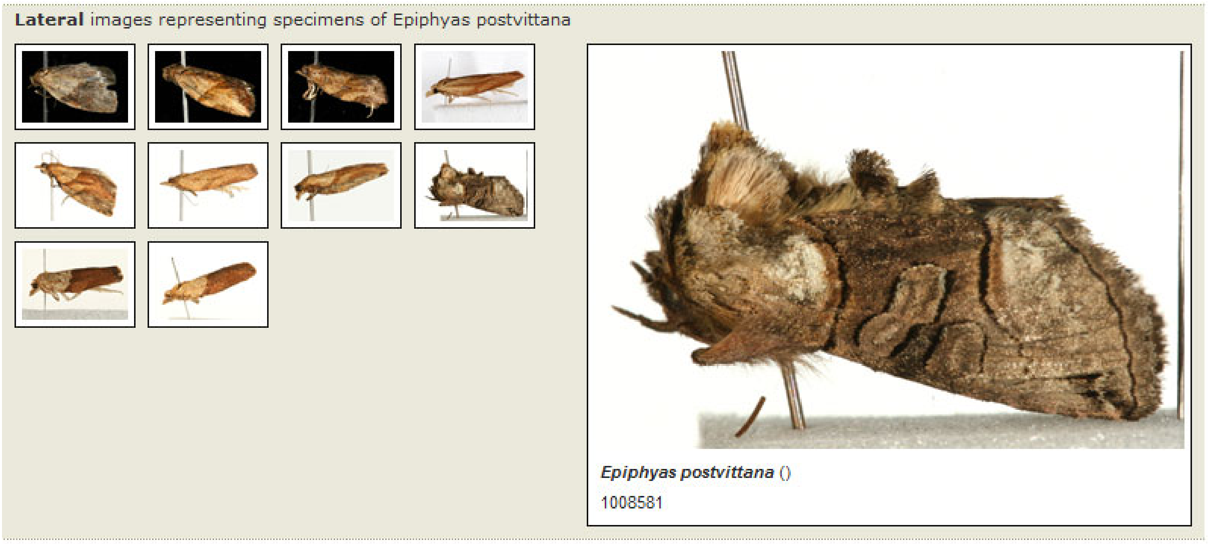


Figure 77: An obvious error in the BOLD database. The specimen illustrated is not a tortricid, but rather a noctuid. This mistake (screenshot on August 1, 2009) has since been corrected.

is not a true representation of the genetic diversity found across all species in the database. In addition, fixed percent cutoffs are used for identifications: less than 1% from a reference sequence for a species-level identification, and less than 3% from a reference sequence for a genus-level identification. In either case, it is impossible to evaluate a less than 100% match without knowing the genetic variation in the DNA barcode region for the species in question. Even if the genetic variation in a particular species is evaluated, and suitable percentage cutoffs established in an identification protocol using BOLD, the dynamic, growing database makes repeatability difficult over time. Although the addition of new sequences is regulated (in contrast to GenBank), a continually updated database creates problems for any protocols that rely on specific content.

The BOLD database contains a number of taxonomic errors that are likely a result of misidentifications or mistakes in sequence or data handling (Fig. 77). These issues are possible to fix over time given the retention of voucher specimen photographs and information; however, as the database grows at an accelerated rate, constant checking by taxonomic experts is required to correct errors. What may be more problematic is the large number of specimens in the database that are “reverse identified.” Reverse identification occurs when one specimen is identified using traditional characters, and other specimens with similar DNA barcodes, evaluated either through a neighbor-joining tree (NJT) analysis or that match within a certain percentage, are identified as that species and submitted to the database. This method of identification is most commonly implemented by “pre-sorting” specimens into species groups based on a NJT and asking an expert to identify a single specimen from each group (T. Gilligan, pers. obs.). The problem with this methodology is that not all specimens that cluster together on a NJT are necessarily the same species, and misidentification errors are easily perpetuated in the database if barcodes are used as a substitute for expert identification. If the species in question are closely related and are similar morphologically, these errors may be nearly impossible to detect.

Species-specific tools

An alternative to using large public databases for specimen identification is to develop species-specific tools that treat a limited range of taxa with more detail and output results that are tailored for persons making regulatory decisions. Several such tools have been developed for Tortricidae, including a diagnostic procedure for four quarantine olethreutines based on a 112-116 bp segment of COI (Barcenas et al. 2005), and numerous diagnostics, mostly involving banding patterns, by Dugdale et al. (2005). Such methods are generally very species-specific, and interpretation of results may rely heavily on assumptions regarding host plants, geographic distribution, or even preliminary assignment of specimens to tribes or genera. However, if the appropriate assumptions are met, species-specific diagnostics may result in a more definitive identification than can be obtained by matching sequences to those in a constantly changing public database.

DNA barcoding for regulatory decisions

To successfully implement a DNA barcoding system for quarantine and regulatory functions, the following criteria must be met:

1) The system should have a low probability of false positives. A false positive occurs when a match is returned for a search taxon that is not represented in the reference database (Type II error; Virgilio et al. 2010).
2) The system should have a low probability of false negatives. A false negative occurs when a match is not returned for a search taxon that is represented in the reference database (Type I error; Virgilio et al. 2010).
3) The system should be able to account for a level of user error.
4) The database should reflect the current taxonomic understanding of the pest species.
5) The database should be consistent and not constantly changing from the addition or removal of sequences or taxa.
6) The system should be able to identify common non-target specimens.
7) The system should be easy to use and protocols easy to replicate.

One of the major problems with any barcode database is the large proportion of taxa that have not yet been barcoded. According to Virgilio et al. (2010), 98% of insect species are not represented in any current barcode database, although this number is not consistent across all orders (BOLD sampling is heavily skewed towards Lepidoptera). Missing taxa in the reference database subject the identification system to Type II errors, or misidentification of queries which are not represented in the reference database (Virgilio et al. 2010). These would be considered false positives if matches are returned when the search taxon is not represented in the database. Type I errors occur when a query is misidentified that is represented in the reference database (Virgilio et al. 2010). These would be considered false negatives if matches are not returned when the search taxon is represented in the database. Given the large amount of species not yet barcoded, Type II errors are much more common than Type I errors; however, this problem can be avoided if the system is used to verify or reject the identification of a specimen against a well-referenced database of pest species – termed “negative identification” by Virgilio et al. 2010.

A majority of studies that attempt to identify unknown species with DNA barcodes use a hierarchical clustering method (primarily neighbor-joining) to associate and differentiate between species groups (e.g., Hebert et al. 2003, Hebert et al. 2004a, Hebert et al. 2004b, Janzen et al. 2005, Ward et al. 2005, Hajibabaei et al. 2006). Other methods have been employed, including BLAST, which attempts to find regions of local similarity between sequences (Altschul et al. 1990), and combinations of clustering, similarity, and alignment-free tree-based algorithms (Little and Stevenson 2007). All of these methods are subject to Type I errors, although the error rate may be low (< 5%; Virgilio et al. 2010).

One novel method of identifying species using DNA barcodes that has not been previously employed is use of the entire COI barcode region in a similarity search. Such a method would greatly reduce false positives (a subset of Type II errors), as a confirmed identification would only result from a 100% match with the query sequence. False positives could only occur if two species shared identical COI barcode sequences, in which case all other matching algorithms would fail as well, and such circumstances are rare in Lepidoptera (but see Kaila and Ståhls 2006). Other Type II errors caused by missing taxa in the database could be reduced by using the system to confirm or deny the identities of a group of heavily-sampled pest species (“negative identification” *sensu* Virgilio et al. 2010). False negatives (a subset of Type I errors) would also be greatly reduced with an exact-matching algorithm, except in cases of haplotype variation unaccounted for in the database (technically a Type II error).

Tortricid DNA sequence search tool

Here I provide details of a DNA barcode identification tool developed using the above methods to assist with the identification or verification of tortricid pests (Lepidoptera: Tortricidae). The tool compares a reference sequence against a local database of COI DNA barcodes using an exact-matching algorithm and returns any matching haplotypes to the user. The tool has been tested and implemented in two USDA-funded projects: *LBAM ID: Tools for diagnosing light brown apple moth and related western U.S. leafrollers (Tortricidae: Archipini)* (Gilligan and Epstein 2009), and *TortAI, Tortricids of Agricultural Importance to the United States (Lepidoptera: Tortricidae)* (Gilligan and Epstein 2012). The first, *LBAM ID*, is used to identify adults and larvae of the light brown apple moth, *Epiphyas postvittana* (Walker), found in California during regulatory inspections. The second, *TortAI*, is used to identify larvae of tortricid pest species intercepted at U.S. ports of entry. As the sequence search tool implemented in *TortAI* is simply an expanded version of the *LBAM ID* tool, the remainder of this chapter focuses on the *TortAI* implementation.

Materials and Methods

DNA extraction was performed using a Qiagen DNeasy Blood and Tissue kit (Qiagen, Valencia, CA). Two or three legs of an adult or a portion of a larva were crushed, incubated overnight at 56°C, and eluted in 100 µl of AE buffer after following the manufacturer's recommended protocol. The DNA barcode region of the cytochrome c oxidase I (COI) was amplified using conventional PCR on an Eppendorf Mastercycler gradient 5331 thermal cycler (Eppendorf AG, Hamburg, Germany). Reactions were performed with TaKaRa Ex Taq HS polymerase (Takara Bio, Shiga, Japan) in total volumes of 50 µl using the manufacturer's recommended volumes of 10X Ex Taq buffer and dNTP mixture. Three sets of primers were used to amplify approximately 658 bp of DNA: LCO-1490—HCO-2198 (Folmer et al. 1994), LepF1—LepR1 (Hajibabaei et al. 2006), or TY-J-1460—C1-N-2191 (Simon et al. 1994); complete primer sequences are listed in Table 113. PCR conditions included an initial denaturation step of 94°C (3min), 32 cycles of 94°C (20 sec)/ 50°C (20 sec)/ 72°C (30 sec), and an extension step of 72°C (5 min). Amplicons were purified using a Qiaquick PCR Purification Kit (Qiagen, Valencia, CA) and eluted into 35 µl of EB buffer. The purified PCR product was sequenced by the University of Chicago Cancer Research Center DNA Sequencing Facility using an Applied Biosystems 3730XL DNA sequencer (Applied Biosystems, Foster City, California). Individual contigs were assembled using Geneious Pro 5.3.4 (Biomatters Ltd., Auckland, New Zealand).

Table 113: COI primers used in this study

Forward Primer	**Sequence**	**Reverse Primer**	**Sequence**
LepF1	ATTCAACCAATCATAAAGATATTGG	LepR1	TAAACTTCTGGATGTCCAAAAAATCA
LCO-1490	GGTCAACAAATCATAAAGATATTGG	HCO-2198	TAAACTTCAGGGTGACCAAAAAATCA
TY-J-1460	TACAATTTATCGCCTAAACTTCAGCC	C1-N-2191	CCCGGTAAAATTAAAATATAAACTTC

Additional target and non-target COI barcode sequences were downloaded from GenBank. All sequences were aligned in Geneious using MAFFT v6.814b (Katoh et al. 2002) and trimmed between the primer regions to obtain sequences of the same length (657 bp). In some cases, a string of N's was added to the beginning or end of a sequence to increase its length consistent with other sequences in the alignment. PAUP* (Swofford 2002) was used to construct a neighbor-joining tree from the final alignment, which was visually inspected for obvious misidentifications or errors. Sequences were extracted into individual files from the alignment using Geneious. Consensus sequences were generated using Geneious alignments for several species, genera, and tribes; an overall "tortricid" consensus sequence generated from the initial alignment was too ambiguous to be useful and was not included in the final tool. Individual and consensus sequences were exported from Geneious in .fasta format; all of the sequences (and associated specimen data) included in the database are provided at the following URL: http://idtools.org/id/leps/tortai/TortAI_DNA_sequence_search_tool_log.pdf.

Abbreviations for depositories, institutions, and organizations are as follows: ANIC = Australian National Insect Collection, CSIRO, Canberra; BOLD = Barcode of Life Data Systems; CSCA = California State Collection of Arthropods, CDFA, Sacramento; CSU = Colorado State University; GenBank = NCBI GenBank; Mission Lab = USDA/APHIS/PPQ Moore Air Base, Mission, TX; TMG = Todd M. Gilligan collection, Loveland, CO; UCB = University of California, Berkeley, Essig Museum of Entomology; UCR = University of California, Riverside; USNM = National Museum of Natural History, Smithsonian, Washington, D.C.

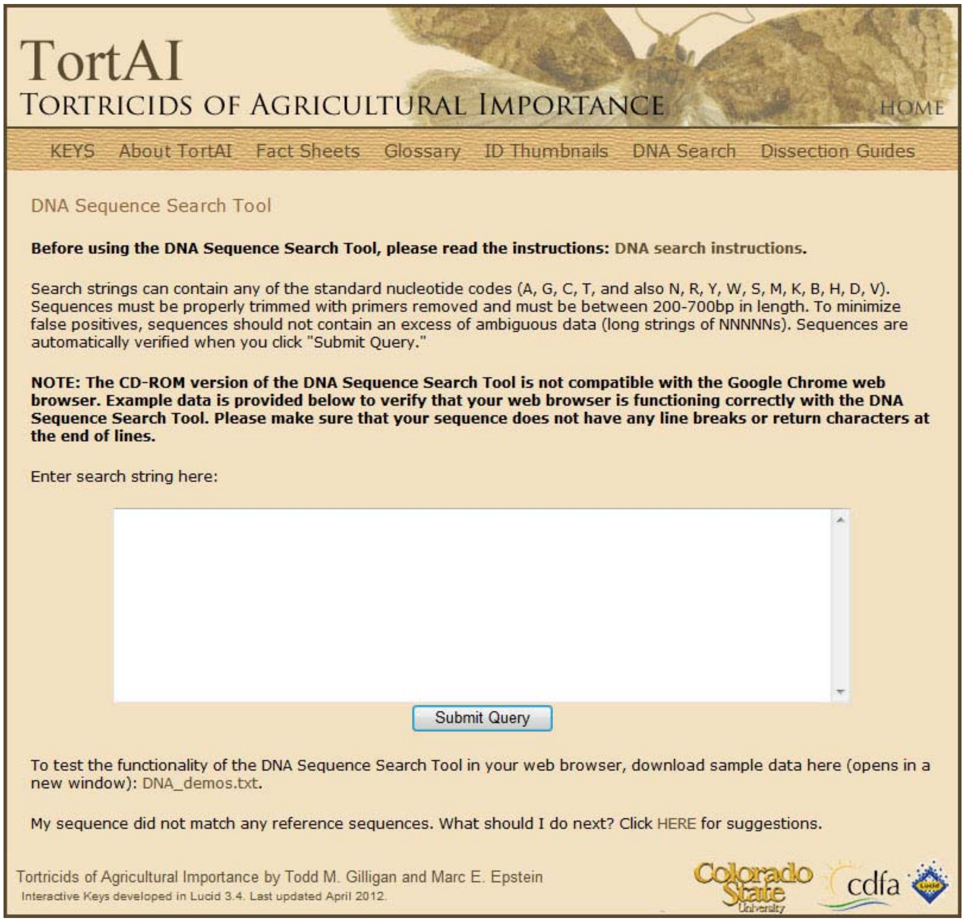
Results

A total of 734 sequences were included in the final database file. Four hundred and seventy-nine sequences were obtained from the following sources: USDA Mission Laboratory, Edinburg, Texas (190; many included in Barr et al. 2011); GenBank (228; most initially submitted through BOLD); light brown apple moth population studies (40; Tooman et al. 2011); and consensus sequences generated in Geneious (21 total). The remaining 255 sequences were obtained from specimens collected by or sent to T. Gilligan from Europe, Africa, Australia, and the Americas. The database represents approximately 300 target (pest) and non-target tortricid species.

Sequence data was incorporated into an XML file; an example of file formatting is presented in Fig. 78. Each sequence is entered individually, with records being separated by <item> </item> tags, which contain species information and the DNA sequence. To account for user error in properly trimming sequences, which was a common problem in testing, a string of 25 N’s was added to the start and end of each DNA sequence in the XML file. Ambiguity codes

```
<dna_sequence>
<item><species>Genus_species_(identifier)</species>
<dna>TTAGGAGCCCCTGATATAGCTTTCCCACGAATAAATAATATAAGATTTTGATTATTGCCACCCTCTATTATACTTTTA
ATTTCAAGAAGAATCGTAGAAAATGGAGCAGGAACAGGATGAACGGTTTATCCCCCCCTTTCATATAGAT</dna></item>
<item><species>Genus_species_(identifier)</species>
<dna>TAGCTATTTTCTCCTTACATTTAGCTGGTATTTCATCAATTTTAGGAGCAGTAAATTTTATTACTACAATTATTAATA
TACGACCAAATAATATATCATTAGATCAAATACCATTATTTGTATGAGCTGTAGGAATTACAGCTTTATT</dna></item>
<item><species>Genus_species_(identifier)</species>
<dna>CCAAATAATATATCATTAGATCAAATACCATTATTTGTATGAGCTGTAGGAATTACAGCTTTATTATTACTTTTATCT
TTACCTGTATTAGCAGGTGCTATTACTATATTATTAACAGATCGAAATTTAAATACCTCTTTTTTTGACC</dna></item>
</dna_sequence>
```

Figure 78: Example of the XML file format used to house DNA data. A complete record, including species and DNA information, is contained between the <item></item> tags.



DNA Sequence Search Tool

Before using the DNA Sequence Search Tool, please read the instructions: DNA search instructions.

Search strings can contain any of the standard nucleotide codes (A, G, C, T, and also N, R, Y, W, S, M, K, B, H, D, V). Sequences must be properly trimmed with primers removed and must be between 200-700bp in length. To minimize false positives, sequences should not contain an excess of ambiguous data (long strings of NNNNNs). Sequences are automatically verified when you click "Submit Query."

NOTE: The CD-ROM version of the DNA Sequence Search Tool is not compatible with the Google Chrome web browser. Example data is provided below to verify that your web browser is functioning correctly with the DNA Sequence Search Tool. Please make sure that your sequence does not have any line breaks or return characters at the end of lines.

Enter search string here:

To test the functionality of the DNA Sequence Search Tool in your web browser, download sample data here (opens in a new window): DNA_demos.txt.

My sequence did not match any reference sequences. What should I do next? Click HERE for suggestions.

Tortricids of Agricultural Importance by Todd M. Gilligan and Marc E. Epstein

Interactive Keys developed in Lucid 3.4. Last updated April 2012.

Figure 79: The HTML front-end for the DNA sequence search tool incorporated into *TortAI*.

```
window.onload = loadIndex;

String.prototype.wordWrap = function(m, b, c){
          var i, j, l, s, r;
          if(m < 1)
                    return this;
          for(i = -1, l = (r = this.split("\n")).length; ++i < l; r[i] += s)
                    for(s = r[i], r[i] = ""; s.length > m; r[i] += s.slice(0, j) + ((s = s.slice(j)).length ? b : ""))
                              j = c == 2 || (j = s.slice(0, m + 1).match(/\S*(\s)?$/))[1] ? m : j.input.length - j[0].length
                              || c == 1 && m || j.input.length + (j = s.slice(m).match(/^\S*/)).input.length;
          return r.join("\n");
};


function loadIndex() { // load indexfile
          // most current browsers support document.implementation
          if (document.implementation && document.implementation.createDocument) {
                    //alert("not ie");
                    //xmlDoc = document.implementation.createDocument("", "", null);
                    //xmlDoc.load("USDA-TORTAI_version1.xml");
                    var xmlhttp = new window.XMLHttpRequest();
                    xmlhttp.open("GET","USDA-TORTAI_version1.xml",false);
                    xmlhttp.send(null);
                    xmlDoc = xmlhttp.responseXML.documentElement;
          }
          // MSIE uses ActiveX
          else if (window.ActiveXObject) {
                    //alert("ie");
                    xmlDoc = new ActiveXObject("Microsoft.XMLDOM");
                    xmlDoc.async = "false";
                    xmlDoc.load("USDA-TORTAI_version1.xml");
          }

          //alert("xmldoc=" + xmldoc);
}


function searchIndex() { // search the index
          //alert("searchIndex");
          if (!xmlDoc) {
                    //alert("inside !xmldoc");
                    loadIndex();
          }
          // get the search term from a form field with id 'searchme'
          var searchterm = document.getElementById("searchme").value;
          var allspecies = xmlDoc.getElementsByTagName("species");
          var alldna = xmlDoc.getElementsByTagName("dna");
          resultsSpecies = new Array;
          resultsDNA = new Array;

//alert("searchterm=" + searchterm);
//alert("alldna=" + alldna);
          if (searchterm.length < 1) {
                    alert("Enter at least one character");
          }
          else {
                    // replace N with ALL wildcard "."
                    var searchtermWildcard1 = searchterm.replace(/N/gi, ".");
                    // replace Y with C or T wildcard "[CT]"
                    // loop through each letter/character, if it is not N or Y replace with "[<letter/character>N]"
                    // so that it matches the letter/character or N in the DNA sequence list
                    var searchtermWildcard2 = "";
                    for (var i=0;i<searchtermWildcard1.length;i++) {
                              var charSearchTerm = searchtermWildcard1.charAt(i);
                              //alert("charSearchTerm=" + charSearchTerm);
                              if ((charSearchTerm != ".") && (charSearchTerm != "R") && (charSearchTerm != "r")
                                        && (charSearchTerm != "Y") && (charSearchTerm != "y")
                                        && (charSearchTerm != "W") && (charSearchTerm != "w")
                                        && (charSearchTerm != "S") && (charSearchTerm != "s")
                                        && (charSearchTerm != "M") && (charSearchTerm != "m")
                                        && (charSearchTerm != "K") && (charSearchTerm != "k")
                                        && (charSearchTerm != "B") && (charSearchTerm != "b")
                                        && (charSearchTerm != "H") && (charSearchTerm != "h")
                                        && (charSearchTerm != "D") && (charSearchTerm != "d")
                                        && (charSearchTerm != "V") && (charSearchTerm != "v")) {
                                        searchtermWildcard2 =  searchtermWildcard2 + "[" + charSearchTerm + "N]" + "\n?";
                              }
                              else {
                                        searchtermWildcard2 =  searchtermWildcard2 + charSearchTerm + "\n?";
                              }
                    }
                    //alert("searchtermWildcard2=" + searchtermWildcard2);
                    var searchtermWildcard3 = searchtermWildcard2.replace(/R/gi, "[AG]") + "\n?";
                    var searchtermWildcard4 = searchtermWildcard3.replace(/Y/gi, "[CT]") + "\n?";
                    var searchtermWildcard5 = searchtermWildcard4.replace(/W/gi, "[AT]") + "\n?";
                    var searchtermWildcard6 = searchtermWildcard5.replace(/S/gi, "[GC]") + "\n?";
                    var searchtermWildcard7 = searchtermWildcard6.replace(/M/gi, "[AC]") + "\n?";
                    var searchtermWildcard8 = searchtermWildcard7.replace(/K/gi, "[GT]") + "\n?";
                    var searchtermWildcard9 = searchtermWildcard8.replace(/B/gi, "[GCT]") + "\n?";
                    var searchtermWildcard10 = searchtermWildcard9.replace(/H/gi, "[ACT]") + "\n?";
                    var searchtermWildcard11 = searchtermWildcard10.replace(/D/gi, "[AGT]") + "\n?";
                    var searchtermWildcard12 = searchtermWildcard11.replace(/V/gi, "[AGC]") + "\n?";

                    //alert("searchtermWildcard12=" + searchtermWildcard12);
```

Figure 80: Javascript code used to query the XML database file.

```
                    for (var i=0;i<alldna.length;i++) {
                              // see if the XML entry matches the search term,
                              // and (if so) store it in an array
                              var dna = alldna[i].lastChild.nodeValue;
                              //alert("alldna[i].lastChild.nodeValue=" + alldna[i].lastChild.nodeValue);
                              var exp = new RegExp(searchtermWildcard12,"gi");
                              //alert("exp=" + exp);
                              var dnaHighlighted = "";
                              var dnaWrapped = dna.wordWrap(50, "\n", 2);
                              if (dna.match(exp) != null) {
                                        resultsSpecies.push(allspecies[i]);
                                        dnaHighlighted = dnaHighlighted + dnaWrapped.replace(dnaWrapped.match(exp), "<font
                                        color='red'>" + dnaWrapped.match(exp) + "</font>");
                                        resultsDNA.push(dnaHighlighted);
                              }
          }
          // send the results to another function that displays them to the user
          showResults(resultsSpecies, resultsDNA, searchterm);
    }
}


// Handle search results
function showResults(resultsSpecies, resultsDNA, searchterm) {

//alert("showresults");
//alert("resultsSpecies=") + resultsSpecies;
//alert("resultsDNA=") + resultsDNA;

    if ((resultsSpecies.length > 0) && (resultsDNA.length > 0)) {
          // if there are any results, put them in a list inside the "resultshere" div
                    var txtOutput = "<b>Your sequence is consistent with the following taxa. Exact matches to the reference
sequence(s) are displayed in red.</b><ul>";
                    for (var i=0;i<resultsSpecies.length;i++) {
                              //alert(resultsDNA[i]);
                              txtOutput = txtOutput + "<li>" + "<p>" + "<b>" + resultsSpecies[i].lastChild.nodeValue + "</b>"
+ "</p>" + "<p>" + resultsDNA[i].replace(/\n/gi, "<br />") + "</p>" + "</li>";
                    }
                    txtOutput = txtOutput + "</ul>";
                    document.getElementById("resultshere").innerHTML = txtOutput;
//alert(txtOutput);
    }
          else {
          // else tell the user no matches were found
                    document.getElementById("resultshere").innerHTML = "<h5>Your sequence is not consistent with any taxa in
the database. Please make sure the sequence does not have any return characters, including at the end. If you received this
message after ensuring that your sequence is formatted correctly and verified, your search was inconclusive. Please view the
link at the bottom of this page for suggestions.</h5>";
          }
}
```

Figure 80 (cont.): Javascript code used to query the XML database file.

are allowed in sequence queries, but all ambiguity codes were replaced with N’s in reference sequences to provide faster searches.

An HTML page (Fig. 79) was created to initiate search queries. Search code is written in Javascript (Fig. 80), and additional code incorporated into the HTML page to validate query (input) sequences, which must meet the following criteria:

1) The sequence must be between 200 and 700 nucleotides long.
2) The sequence must not contain forward or reverse primers used to amplify the COI barcode region (list in Table 113).
3) The sequence must not contain long strings of ambiguous data (long strings of N’s). Standard nucleotide ambiguity codes other than N’s are allowed in any quantity.

If the above criteria are not met, the user will receive a warning with a specific error message, although the user will not be prevented from continuing the query. After acknowledging the sequence verification message, the user is returned an HTML page with all matching haplotypes and the exact sequence match(es) highlighted in red (Fig. 81).

DNA sequence search pages were incorporated into *LBAM ID* (for *E. postvittana* only; Gilligan and Epstein 2009), and *TortAI* (for several tortricid pests; Gilligan and Epstein 2012). These search tools can be accessed at the following URLs:

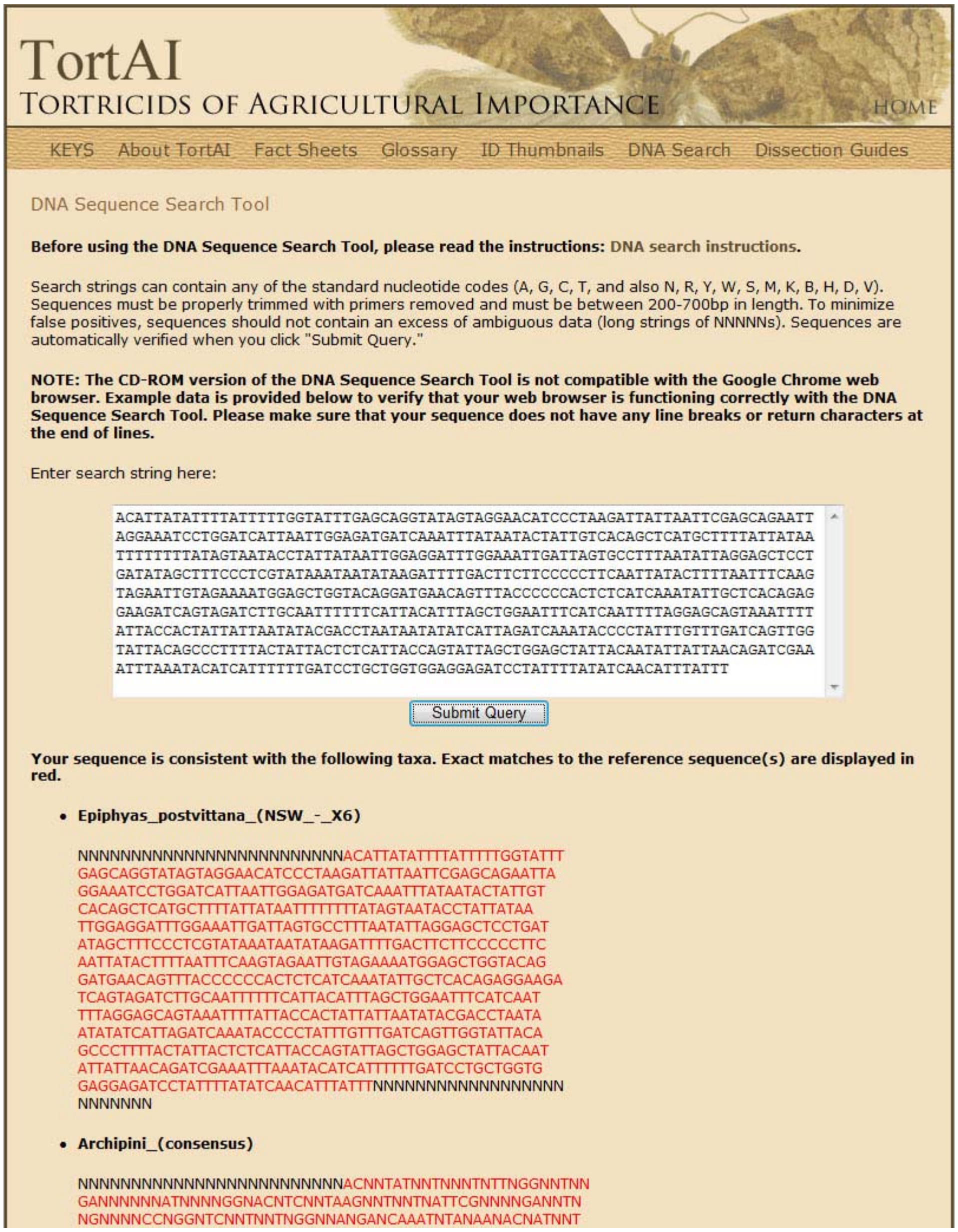
LBAM ID: http://itp.lucidcentral.org/id/lep/lbam/dna_search.html

TortAI: http://idtools.org/id/leps/tortai/dna_search.html

Instructions

This section outlines the instructions for using the DNA sequence search tool. Most text is modified from the version incorporated into *TortAI* (Gilligan and Epstein 2012).

The DNA Sequence Search Tool is designed to assist users in verifying the identity of tortricid specimens, primarily larvae. The tool searches for an exact match between the input sequence and the reference database, which contains more than 700 COI haplotypes of over 300 target and non-target species from around the world. Each haplotype consists of a 657 bp sequence of the region of COI commonly known as the DNA barcode region. The search tool assumes that the input sequence was generated using primers designed to amplify the COI barcode region and that the sequence has been properly trimmed and the primer sequences removed. A sequence that is too long will not return a match regardless of the specimen identity. Input sequences must be verified to be of sufficient length, to be free of primers, and to not contain excessive ambiguous data. Sequences are automatically verified when the user clicks the "Submit Query" button.



DNA Sequence Search Tool

Before using the DNA Sequence Search Tool, please read the instructions: DNA search instructions.

Search strings can contain any of the standard nucleotide codes (A, G, C, T, and also N, R, Y, W, S, M, K, B, H, D, V). Sequences must be properly trimmed with primers removed and must be between 200-700bp in length. To minimize false positives, sequences should not contain an excess of ambiguous data (long strings of NNNNNs). Sequences are automatically verified when you click "Submit Query."

NOTE: The CD-ROM version of the DNA Sequence Search Tool is not compatible with the Google Chrome web browser. Example data is provided below to verify that your web browser is functioning correctly with the DNA Sequence Search Tool. Please make sure that your sequence does not have any line breaks or return characters at the end of lines.

Enter search string here:

```
ACATTATATTTTATTTTTGGTATTTGAGCAGGTATAGTAGGAACATCCCTAAGATTATTAATTCGAGCAGAATT
AGGAAATCCTGGATCATTAATTGGAGATGATCAAATTTATAATACTATTGTCACAGCTCATGCTTTTATTATAA
TTTTTTTTATAGTAATACCTATTATAATTGGAGGATTTGGAAATTGATTAGTGCCTTTAATATTAGGAGCTCCT
GATATAGCTTTCCCTCGTATAAATAATATAAGATTTTGACTTCTTCCCCCTTCAATTATACTTTTAATTTCAAG
TAGAATTGTAGAAAATGGAGCTGGTACAGGATGAACAGTTTACCCCCCACTCTCATCAAATATTGCTCACAGAG
GAAGATCAGTAGATCTTGCAATTTTTTCATTACATTTAGCTGGAATTTCATCAATTTTAGGAGCAGTAAATTTT
ATTACCACTATTATTAATATACGACCTAATAATATATCATTAGATCAAATACCCCTATTTGTTTGATCAGTTGG
TATTACAGCCCTTTTACTATTACTCTCATTACCAGTATTAGCTGGAGCTATTACAATATTATTAACAGATCGAA
ATTTAAATACATCATTTTTTGATCCTGCTGGTGGAGGAGATCCTATTTTATATCAACATTTATTT
```

Your sequence is consistent with the following taxa. Exact matches to the reference sequence(s) are displayed in red.

- **Epiphyas_postvittana_(NSW_-_X6)**

```
NNNNNNNNNNNNNNNNNNNNNNNNNACATTATATTTTATTTTTGGTATTT
GAGCAGGTATAGTAGGAACATCCCTAAGATTATTAATTCGAGCAGAATTA
GGAAATCCTGGATCATTAATTGGAGATGATCAAATTTATAATACTATTGT
CACAGCTCATGCTTTTATTATAATTTTTTTTATAGTAATACCTATTATAA
TTGGAGGATTTGGAAATTGATTAGTGCCTTTAATATTAGGAGCTCCTGAT
ATAGCTTTCCCTCGTATAAATAATATAAGATTTTGACTTCTTCCCCCTTC
AATTATACTTTTAATTTCAAGTAGAATTGTAGAAAATGGAGCTGGTACAG
GATGAACAGTTTACCCCCCACTCTCATCAAATATTGCTCACAGAGGAAGA
TCAGTAGATCTTGCAATTTTTTCATTACATTTAGCTGGAATTTCATCAAT
TTTAGGAGCAGTAAATTTTATTACCACTATTATTAATATACGACCTAATA
ATATATCATTAGATCAAATACCCCTATTTGTTTGATCAGTTGGTATTACA
GCCCTTTTACTATTACTCTCATTACCAGTATTAGCTGGAGCTATTACAAT
ATTATTAACAGATCGAAATTTAAATACATCATTTTTTGATCCTGCTGGTG
GAGGAGATCCTATTTTATATCAACATTTATTTNNNNNNNNNNNNNNNNN
NNNNNNN
```

- **Archipini_(consensus)**

```
NNNNNNNNNNNNNNNNNNNNNNNNNACNNTATNNTNNNTNTTNGGNNTNN
GANNNNNNATNNNNGGNACNTCNNTAAGNNTNNTNATTCGNNNNGANNTN
NGNNNNCCNGGNTCNNTNNTNGGNNANGANCAAATNTANAANACNATNNT
```

Figure 81: Results of a successful query using the DNA sequence search tool. Exact sequence matches are showing in red.

The search tool will return the name and description of haplotype(s) if a match is found. A message that the sequence is consistent with one or more haplotypes means that the input sequence exactly (100%) matches one of the reference sequences. This information can be used to assist with specimen identity, but the results should be used with caution. It is possible that sequencing or identification errors in the reference database could lead to misidentifications; however, the possibility of false positives is greatly reduced with an exact matching algorithm. A message that the sequence is not consistent with any haplotypes in the database means that the input sequence does not exactly match any of the reference sequences. This could be due to errors in the input sequence or a failure to sample all haplotypes of a particular species in the reference database, and the lack of a match should be treated as an inconclusive result. Alternative molecular databases (GenBank or BOLD) can be consulted if no match is found.

Search sequences should be generated with one of three common primer sets used to target the DNA barcode region of Cytochrome oxidase I (COI). Acceptable primer sets are listed in Table 113 and sequences should be trimmed between the primer sets. While the recommended primers generate sequences approximately 650 bp long, a shorter fragment can be used if the sequence is degraded (probably requiring different primers). Final sequence length should be between 200–700 bp. Cut and paste the sequence into the DNA sequence search tool query window. Make sure the sequence contains no blank spaces, line breaks, or return characters. Click "Submit Query" to submit the sequence to the search tool. The search tool will automatically check the sequence for any problems (length, primers, long strings of NNN's). If the sequence verifies, you will receive a message stating "Sequence is verified to be of the correct length and to not contain any primer sequences or excessive ambiguous data. Click OK to search the database." If the sequence does not verify, you will receive a message stating the reason, and be returned to the query window to correct the problem.

Successful searches will return the following message: "Your sequence is consistent with the following taxa. Exact matches to the reference sequence(s) are displayed in red." Complete sequences will be displayed below the query window with matching regions highlighted in red. Failed searches will return the following message: "Your sequence is not consistent with any taxa in the database. Please make sure the sequence does not have any return characters, including at the end. If you received this message after ensuring that your sequence is formatted correctly and verified, your search was inconclusive. Please view the link at the bottom of this page for suggestions." Further information on steps to take if your sequence was not consistent with any reference sequences in the database is listed below:

The DNA Search Sequence Tool performs an "exact matching" algorithm against sequences in the reference database. Failure to match any reference sequences could be due to one of the following reasons:

1. The search sequence is not formatted correctly

Search sequences must be properly trimmed between the primers. Three sets of primers are commonly used to generate COI barcode sequences (LepF1-LepR1, LCO1490-HCO2198, and TYJ1460-C1N2191). Sequences should be trimmed between these primer regions so that they are approximately 650bp long. Sequences should also be continuous strings of bases (letters) with no spaces, line breaks, or return characters, including at the end of the sequence.

2. The taxon is not included in the reference database

While the reference database contains more than 300 taxa, it does not contain representatives of all 9,500 described tortricid species. It is also possible that the search sequence is not from a tortricid.

3. The haplotype for the queried specimen is not in the reference database

Although the reference database contains more than 800 unique COI haplotypes, it does not contain all possible haplotypes for all species. Some species, such as *Epiphyas postvittana*, have more than 40 unique haplotypes for the COI DNA barcode region.

4. Sequencing or PCR errors exist in the search or reference sequences

It is possible that sequencing or PCR problems could have led to errors in the search or reference sequences. Without resampling all taxa these errors would be difficult or impossible to detect.

Conclusions

The DNA sequence search tool described here has been successfully used to identify both adults and larvae of tortricid pest species. By using a local database with heavy sampling of pest species identified by a taxonomic expert, many of the problems associated with BOLD and GenBank are avoided. This is a working example of pest identification using DNA barcodes following the “negative identification” model of Virgilio et al. (2010).

Although DNA barcoding can be used for species identification, examination of the issues associated with public DNA barcode databases reinforce that barcoding should not be a replacement for proper taxonomy. There is a temptation to revise the reference database based on novel haplotypes that potentially represent new species because of arbitrary percentage cutoffs implemented in the database. These divergent sequences may point to a problem in the alpha taxonomy of a group (cryptic species or species complexes), or they may simply represent the genetic diversity within a given taxon. While barcoding may illuminate taxonomic errors or cryptic species, it is not a substitute for proper morphological study and phylogenetic analyses by an expert in the group. In addition, current concepts of quarantine pests do not necessarily coincide with perfect alpha taxonomy (e.g., in the case of species complexes), and this should not be a requirement of the identification system. Taxonomists should be allowed to dictate what defines a species, and this information should be used to define species within the DNA barcoding universe, not vice-versa.

References

Altschul, S. F., W. Gish, W. Miller, E. W. Myers and D. J. Lipman. 1990. Basic local alignment search tool. Journal of Molecular Biology. 215: 403–410.

Armstrong, K. F. and S. L. Ball. 2005. DNA barcodes for biosecurity: invasive species identification. Philosophical Transactions of the Royal Society B. 360: 1813–1823.

Ball, S. L. and K. F. Armstrong. 2006. DNA barcodes for insect pest identification: a test case with tussock moths (Lepidoptera: Lymantriidae). Canadian Journal of Forest Research. 36: 337–350.

Barcenas, N. M., T. R. Unruh and L. G. Neven. 2005. DNA diagnostics to identify internal feeders (Lepidoptera: Tortricidae) of pome fruits of quarantine importance. Journal of Economic Entomology. 98: 299–306.

Barr, N. B., L. A. Ledezma, R. E. Farris, M. E. Epstein and T. M. Gilligan. 2011. A multiplex real-time polymerase chain reaction assay to diagnose *Epiphyas postvittana* (Lepidoptera: Tortricidae). Journal of Economic Entomology. 104(5): 1706–1719.

Benson, D. A., I. Karsch-Mizrachi, D. J. Lipman, J. Ostell and E. W. Sayers. 2010. GenBank. Nucleic Acids Research. 38. DOI:10.1093/nar/gkp1024

Brower, A. V. Z. 2006. Problems with DNA barcodes for species delimitation: ‘ten species’ of *Astraptes fulgerator* reassessed (Lepidoptera: Hesperiidae). Systematics and Biodiversity. 4(2):127–132.

Dugdale, J. S., D. Gleeson, L. H. Clunie and P. W. Holder. 2005. A diagnostic guide to Tortricidae encountered in field surveys and quarantine inspections in New Zealand: Morphological and molecular characters. National Plant Pest Reference Laboratory. 161 pp.

Eddy, S. R. 1996. Hidden Markov models. Current Opinion in Structural Biology. 6: 361–365.

Folmer, O., M. Black, W. Hoeh, R. Lutz and R. Vrijenhoek. 1994. DNA primers for amplification of mitochondrial cytochrome c oxidase I from diverse metazoan invertebrates. Molecular Marine Biology and Biotechnology. 3: 294–299.

Gilligan, T. M. and M. E. Epstein. 2009. LBAM ID: Tools for diagnosing light brown apple moth and related western U.S. leafrollers (Tortricidae: Archipini). CD-ROM. Center for Plant Health Science and Technology, USDA/APHIS/PPQ, Raleigh, NC.

Gilligan, T. M., M. E. Epstein, S. C. Passoa, J. A. Powell, O. C. Sage & J. W. Brown. 2011. Discovery of *Lobesia botrana* ([Denis & Schiffermüller]) in California: an invasive species new to North America (Lepidoptera: Tortricidae). Proceedings of the Entomological Society of Washington. 113(1): 14–30.

Gilligan, T. M. and M. E. Epstein. 2012. TortAI, Tortricids of Agricultural Importance to the United States (Lepidoptera: Tortricidae). CD-ROM. Identification Technology Program (ITP), USDA/APHIS/PPQ/CPHST, Fort Collins, CO.

Hajibabaei, M., D. H. Janzen, J. M. Burns, W. Hallwachs and P. D. N. Hebert. 2006. DNA barcodes distinguish species of tropical Lepidoptera. Proceedings of the National Academy of Sciences of the United States of America. 103: 968–971.

Hebert, P. D. N., A. Cywinska, S. L. Ball and J. R. deWaard. 2003. Biological identifications through DNA barcodes. Proceedings of the Royal Society of London, Series B. 270: 313–321.

Hebert, P. D. N., E. H. Penton, J. M. Burns, D. H. Janzen and W. Hallwachs. 2004a. Ten species in one: DNA barcoding reveals cryptic species in the neotropical skipper butterfly *Astraptes fulgerator*. Proceedings of the National Academy of Sciences of the United States of America. 101: 14812–14817.

Hebert, P. D. N., M. Y. Stoeckle, T. S. Zemlak and C. M. Francis. 2004b. Identification of birds through DNA barcodes. PLoS Biology. 2(10):e312.

Janzen, D. H., M. Hajibabaei, J. M. Burns, W. Hallwachs, E. Remigio and P. D. N. Hebert. 2005. Wedding biodiversity inventory of a large and complex Lepidoptera fauna with DNA barcoding. Philosophical Transactions of the Royal Society B: Biological Sciences. 360: 1835–1845.

Johnson, M., I. Zaretskaya, Y. Raytselis, Y. Merezhuk, S. McGinnis and T. L. Madden. 2008. NCBI BLAST: a better web interface. Nucleic Acids Research. 36: W5–W9.

Kaila, L. and G. Ståhls. 2006. DNA barcodes: evaluating the potential of COI to differentiate closely related species of *Elachista* (Lepidoptera: Gelechioidea: Elachistidae) from Australia. Zootaxa. 1170: 1–26.

Katoh, Kazutaka, K. Misawa, K. Kuma and T. Miyata. 2002. MAFFT: a novel method for rapid multiple sequence alignment based on fast Fourier transform. Nucleic Acids Research. 30(14): 3059–3066.

Korning, P. G., S. M. Hebsgaard, P. Rouze and S. Brunak. 1996. Cleaning the GenBank Arabidopsis thaliana data set. Nucleic Acids Research. 24: 316–320.

Kumar, S., J. Dudley, M. Nei and K. Tamura. 2008. MEGA: A biologist-centric software for evolutionary analysis of DNA and protein sequences. Briefings in Bioinformatics. 9: 299–306.

Kwong, S., A. Srivathsan and R. Meier. 2012. An update on DNA barcoding: low species coverage and numerous unidentified sequences. Cladistics. DOI: 10.1111/j.1096-0031.2012.00408.x

Little, D. P. and D. W. Stevenson. 2007. A comparison of algorithms for identification of specimens using DNA barcodes: examples from gymnosperms. Cladistics. 23: 1–21.

Moritz, C. and C. Cicero. 2004. DNA barcoding: promise and pitfalls. PLoS Biology. 2: 1529–1531.

Nagoshi, R. N., J. Brambila and R. L. Meagher. 2011. Use of DNA barcodes to identify invasive armyworm *Spodoptera* species in Florida. Journal of Insect Science. 11: 154 (1–11).

Pennisi E. 1999. Keeping genome databases clean and up to date. Science. 286(5439): 447–450.

Ratnasingham, S. and P. D. N. Hebert. 2007. BOLD: The barcode of life data system. Molecular Ecology Notes. DOI: 10.1111/j.1471-8286.2006.01678.x

Rubinoff, D. 2006. Utility of mitochondrial DNA barcodes in species conservation. Conserv Biol. 20: 1026–1033.

Rubinoff, D., S. Cameron and K. Will. 2006a. A genomic perspective on the shortcomings of mitochondrial DNA for "barcoding" identification. Journal of Heredity. 97: 581–594.

Rubinoff, D., S. Cameron and K. Will. 2006b. Are plant DNA barcodes a search for the Holy Grail? Trends Ecol Evol. 21: 1–2.

Scheffer, S. J., M. L. Lewis and R. C. Joshi. 2006. DNA barcoding applied to invasive leafminers (Diptera: Agromyzidae) in the Philippines. Annals of the Entomological Society of America. 99: 204–210.

Simon, C., F. Frati, A. Beckenbach, B. Crespi, H. Liu and P. Flook. 1994. Evolution, weighting and phylogenetic utility of mitochondrial gene sequences and a compilation of conserved polymerase chain reaction primers. Annals of the Entomological Society of America. 87: 651–701.

Song, H., J. E. Buhay, M. F. Whiting and K. A. Crandall. 2008. Many species in one: DNA barcoding overestimates the number of species when nuclear mitochondrial pseudogenes are coamplified. Proceedings of the National Academy of Sciences of the United States of America. 105: 13486–13491.

Sundberg, P., E. Thuroczy Vodoti and M. Strand. 2010. DNA barcoding should accompany taxonomy – the case of *Cerebratulus* spp (Nemertea). Molecular Ecology Resources. 10: 274–281.

Swofford, D. L. 2002. PAUP*. Phylogenetic analysis using parsimony (*and other methods). Sinauer Associates, Sunderland, MA.

Tooman, L. K., C. J. Rose, C. Carraher, D. M. Suckling, S. R. Paquette, L. A. Ledezma, T. M. Gilligan, M. Epstein, N. B. Barr and R. D. Newcomb. 2011. Patterns of mitochondrial haplotype diversity in the invasive pest *Epiphyas postvittana* (Lepidoptera: Tortricidae). Journal of Economic Entomology. 104(3): 920–932.

Virgilio, M., T. Backeljau, B. Nevado and M. De Meyer. 2010. Comparative performances of DNA barcoding across insect orders. BMC Bioinformatics. 11: 206 (1–10).

Ward, R. D., T. S. Zemlak, B. H. Innes, P. R. Last and P. D. N. Hebert. 2005. DNA barcoding Australia's fish species. Philosophical Transactions of the Royal Society B: Biological Sciences. 360: 1847–1857.

Wesche, P. L., D. J. Gaffney and P. D. Keightley. 2004. DNA sequence error rates in GenBank records estimated using the mouse genome as a reference. Mitochondrial DNA. 15: 362–364.

Will, K. W. and D. Rubinoff. 2004. Myth of the molecule: DNA barcodes for species cannot replace morphology for identification and classification. Cladistics. 20: 47–55.

Will, K. W., B. D. Mishler and Q. D. Wheeler. 2005. The perils of DNA barcoding and the need for integrative taxonomy. Systematic Biology. 54: 844–851.

Chapter 8

The type species of *Eucosma* Hübner (Lepidoptera: Tortricidae: Eucosmini)

Introduction

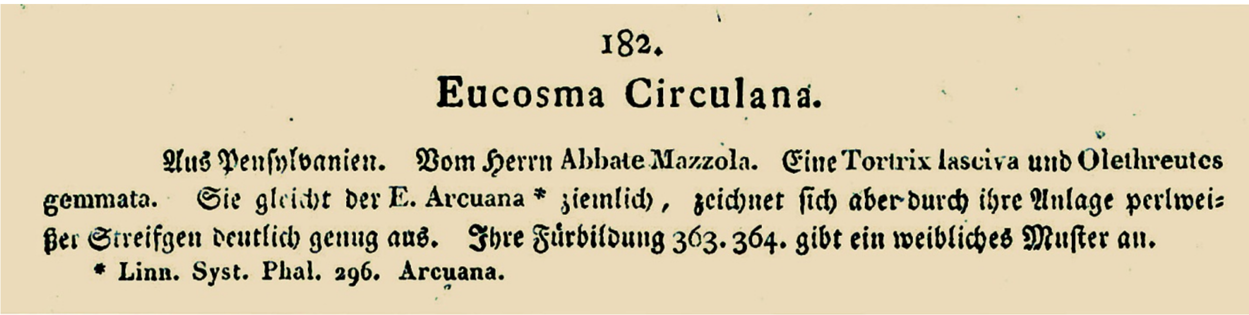
Eucosma Hübner is the largest genus in the Tortricidae, consisting of some 290 described species (Gilligan et al. 2012). Its distribution is Holarctic, with the highest level of species diversity occurring in western North America. There are several descriptive accounts of the genus in the literature (e.g. Heinrich 1923; Obraztsov 1968; Razowski 1989, 2003; Gilligan et al. 2008), but no autapomorphies have been reported for *Eucosma* that would distinguish it from closely related genera such as *Pelochrista* Lederer. The resulting ambiguity, which has persisted for nearly two centuries and resulted in different interpretations of these taxa in the Palearctic and Nearctic, is due in part to the lack of a type specimen for *Eucosma circulana* Hübner.

The name *Eucosma* first appeared with the description of *E. circulana* in the second volume of Hübner's *Zuträge zur Sammlung exotischer Schmettlinge* [sic] (1823). The "*Zuträge*" was published in five parts (the last two authored by Carl Geyer) between 1818 and 1837 as a supplement to *Sammlung exotischer Schmetterlinge*, Hübner's (1806–[1832], [1819–1832]) prominent work on exotic (non-Palearctic) Lepidoptera (Hemming 1937a, b). The description of *E. circulana* consists of a short paragraph of text (Fig. 82) and hand painted illustrations (numbered 363 and 364) of dorsal and ventral views, respectively, of a female from Pennsylvania (Fig. 83). The fate of the illustrated specimen is unknown, and consequently the

---

*This chapter will be submitted for publication as:

Gilligan, T. M. & D. J. Wright. The type species of *Eucosma* Hübner (Lepidoptera: Tortricidae: Eucosmini).



182.

Eucosma Circulana.

Aus Penſylvanien. Vom Herrn Abbate Mazzola. Eine Tortrix lasciva und Olethreutes gemmata. Sie gleicht der E. Arcuana * ziemlich, zeichnet ſich aber durch ihre Anlage perlweißer Streifgen deutlich genug aus. Ihre Fürbildung 363. 364. gibt ein weibliches Muſter an.

* Linn. Syst. Phal. 296. Arcuana.

Figure 82. Original description of *E. circulana* from Hübner (1823).

identity of *E. circulana* has never been satisfactorily established. The Hübner types were acquired by Vincenz Abbate Edler von Mazzola in the early part of the 19th century, and the European material was deposited in the “Hof-Naturalien-Kabinett” at the Hofburg Imperial Library in 1823 (Calhoun 2003). There is no record of the disposition of the exotic material. If it also went to Hofburg, then the *E. circulana* type may have perished in a fire during the Vienna Rebellion of 1848 that destroyed many of the Hübner types (Calhoun 2003). The remaining Hübner specimens currently reside in the Naturhistorisches Museum Wien in Vienna (NMW). In response to an inquiry to NMW, we were informed that there are no specimens labeled “*Eucosma circulana*” in their collections (S. Gaal, pers. comm. 2012). For these reasons we conclude that the *E. circulana* type either is lost or was destroyed.

Prior to Heinrich (1923), there was no consensus as to the application of *Eucosma* and related generic names (such as *Paedisca* Treitschke). Apart from the description of *E. circulana*, the earliest literature references to *Eucosma* are Hübner ([1816–1825]), who listed *E. circulana* in his *Verzeichniss bekannter Schmettlinge* [sic], and Geyer (1832), who described *Eucosma tuberculana* (determined by Fernald (1882) to be a pyralid) in the fourth volume of the “*Zuträge.*” Mid-19th century authors (e.g. Wilkinson 1859, Heinemann 1863) utilized *Paedisca* for Palearctic species, although Walker (1863) listed two species of *Eucosma* (including *E. circulana*) as present in the British Museum. Fernald (1882) placed *E. circulana* under *Paedisca*, presumably under the assumption that it was the same species as *Callimosema* [= *Pelochrista*] *scintillana* Clemens (1865) and *Paedisca dodecana* [= *Pelochrista scintillana*] Zeller (1875), which he treated as synonyms. This is the first instance of North American taxonomists interpreting the

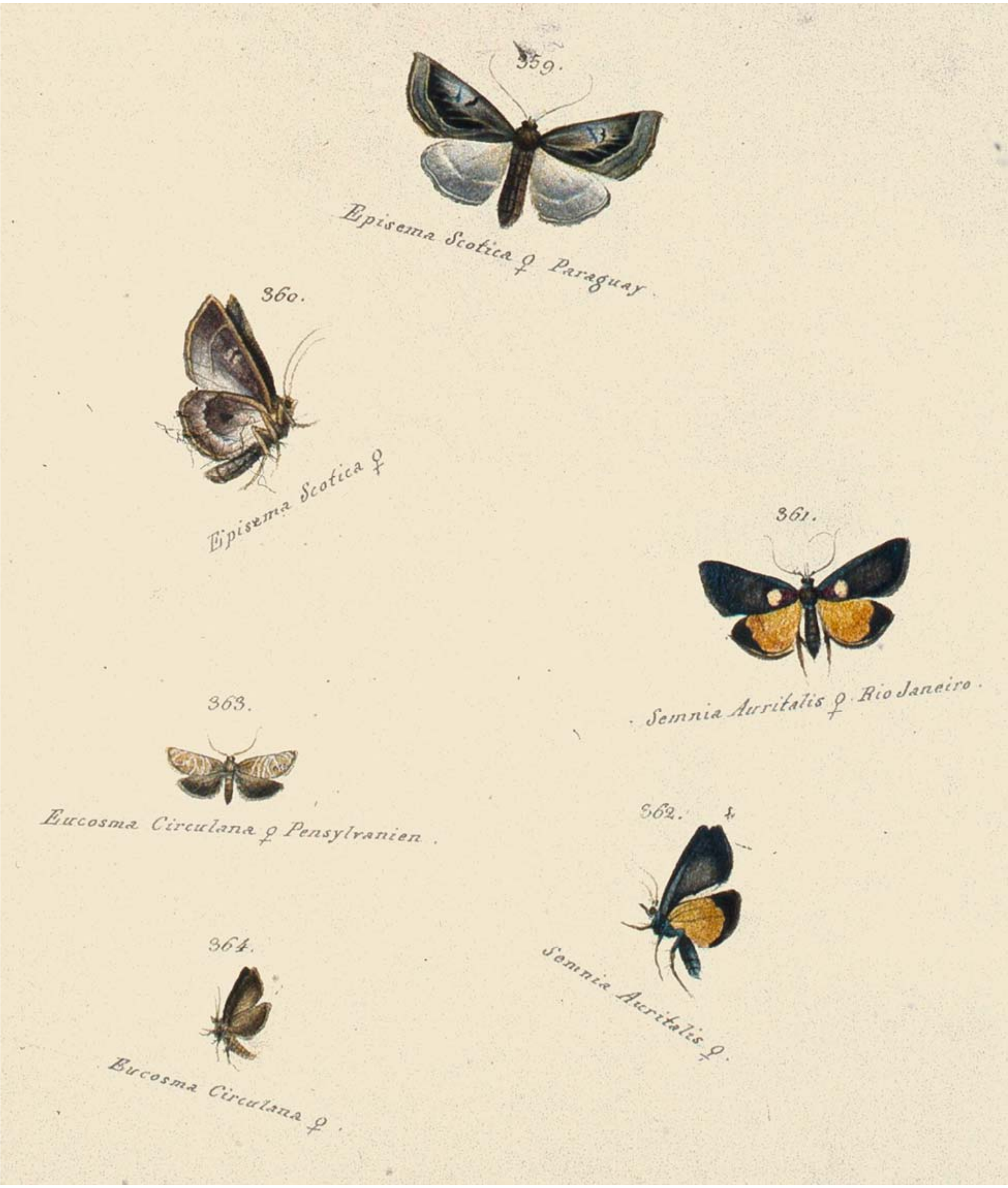


Figure 83. Original illustrations of *E. circulana* by Hübner (Figs. 363, 364; Plate [63]).

species now known as *Pelochrista scintillana* (Clemens) as Hübner's *E. circulana*. Fernald (1891) repeated this arrangement in Smith's *List of Lepidoptera of Boreal America*, but in his list of North American Lepidoptera (Fernald 1903), he followed Walsingham (1897) and treated *Paedisca* as a junior synonym of *Eucosma*. In the same publication he also stated that *E. circulana* occurs in the "So[uth] Atl[antic] States." In his last contribution on this subject, Fernald (1908) treated 25 generic names as synonyms of *Eucosma* and designated "*Eucosma circulana* Hübner" as the type species of the genus. Walsingham (1914) expanded on Fernald's list and included other genera such as *Crocidosema* Zeller, *Notocelia* Hübner, *Spilonota* Stephens, *Strepsicrates* Meyrick, and *Thiodia* Hübner as synonyms of *Eucosma*, in essence combining together all of the genera at the time that would currently be considered Eucosmini. Pierce and Metcalfe (1922) incorporated genitalic characters into their classification of British Tortricidae. They claim to have examined a male *E. circulana*, although the specimen was almost certainly a male *P. scintillana*, as no *E. circulana* are present in the BMNH (K. Tuck, pers. comm.) and the two species were synonymous at the time. As a result, Pierce and Metcalfe assigned the species that we would today consider *Eucosma* to *Pelochrista*.

The genus-level confusion addressed by these early authors was to a large extent resolved with the 1923 publication of Heinrich's *Revision of the North American moths of the subfamily Eucosminae of the family Olethreutidae*. Heinrich was the first to use features of the male genitalia to characterize the Nearctic genera, and he resurrected many of the generic names synonymized by Fernald and Walsingham. His concept of *E. circulana* was based on specimens from Florida and Louisiana with genitalia that differ substantially from those of *P. scintillana* (which had been accepted by North American taxonomists as synonymous with *E. circulana* for approximately 40 years). He also described the "variety" *E. circulana gemellana*, which closely resembles *E. circulana* in forewing appearance but differs sufficiently in genitalia to justify the use of a subspecies (or species) designation. Heinrich's interpretation of the type species was accepted by subsequent authors such as Obraztsov (1968), Powell (1983), and Brown (2005).

Heinrich's *E. circulana* is poorly represented in collections, and literature records from the late 19th and early 20th centuries (e.g., Walsingham 1884, Kearfott 1905) are unreliable because they most likely refer to *P. scintillana*. Consequently, we know very little about the range of *E. circulana* and even less about its historical distribution. There are no verified modern records of it from Pennsylvania, the type locality reported by Hübner. Engel (1908) reported collecting *E. circulana* from Pennsylvania; however, we examined these specimens, currently housed in the CMNH, and determined them all to be *P. scintillana*. Moreover, there are several other North American species that are similar in forewing pattern and coloration to Hübner's illustration of *E. circulana*, and in recent years there has been informal discussion as to which might be best suited to bear the *E. circulana* name (R. Brown, pers. comm.). Here we evaluate the various candidates, ranking them according to morphological features illustrated in Hübner's figures. We designate a Neotype for *E. circulana*, elevate *E. gemellana* to species status, describe a new species previously confused with *E. gemellana*, and provide redescriptions of *E. circulana*, *E. fraudabilis*, and *E. gemellana*. This work is a preliminary step toward a forthcoming revision of *Eucosma*, *Pelochrista*, and *Phaneta* based on molecular and morphological data.

Materials and Methods

One hundred and seventy-five adult specimens (146 ♂, 29 ♀) were examined in this study, together with 37 associated genitalia preparations. Genitalia dissection methodology follows Brown and Powell (1991) except that some preparations used Euparal (Bioquip Products, Rancho Dominguez, Calif.) as the mounting medium. Adult photographs were taken using a Canon EOS digital SLR (Canon U.S.A., Lake Success, N.Y.) and edited using Adobe Photoshop CS5 Extended (Adobe Systems Inc., San Jose, Calif.).

Morphological terms and wing pattern descriptions follow Horak (2006) and Gilligan et al. (2008). Positions of strigulae labeled in Fig. 84 are estimated from wing markings and venation of the candidate *E. circulana* species along with a generalized olethreutine wing pattern

(Baixeras 2002; Gilligan et al. 2008). The following abbreviations and symbols are utilized: FW = forewing; FWL = forewing length, measured from base to apex including fringe; AR = forewing aspect ratio = FWL divided by medial forewing width; NR = neck ratio = minimal neck width divided by valval width near saccular corner; SA = saccular angle = angle formed at juncture of ventral margins of sacculus and neck; ca. = circa (approximately); HW = hindwing; n = number of specimens examined. Abbreviations of institutional and private collections providing study material are as follows: AMNH, American Museum of Natural History, New York, N. Y.; BMNH, The Natural History Museum, London; CMNH, Carnegie Museum of Natural History, Pittsburgh, Pennsylvania; CNC, Canadian National Collection of Insects, Arachnids, and Nematodes, Ottawa, Ontario, Canada; DJW, Donald J. Wright collection, Cincinnati, Ohio; FSCA, Florida State Collection of Arthropods, Gainesville, Florida; LDG, Loran D. Gibson collection, Florence, Kentucky; MEM, Mississippi Entomological Museum, Mississippi State, Mississippi; TMG, Todd M. Gilligan collection, Loveland, Colorado; USNM, National Museum of Natural History, Smithsonian Institution, Washington, D.C.

It was noted by Riley (1886) that Hübner's figures are not necessarily uniform from one copy of the "*Zuträge*" to another, the colors sometimes varying with the colorist and/or the effects of aging. We examined the plate containing *E. circulana*, numbered [63], (Fig. 83) in copies of the "*Zuträge*" located at the USNM, the CNC, and the Cyril F. dos Passos literature collection at Wittenberg University Library, Springfield, Ohio. We found them to be reasonably consistent, with the pattern elements unchanged from copy to copy.

Discussion and Results

Roughly translated from the German, Hübner's description of *E. circulana* (Fig. 82) reads: "From Pensilvania [Pennsylvania]. Discovered by [From] Mr. Abbate Mazzola. A *Tortrix* [= *Phalanx* = Superfamily] *lasciva* [= *Tribus* = Family] and *Olethreutes* [= *Stirps* = Subfamily] *gemmata* [= *Familia* = Tribe]. It resembles *E. Arcuana** [= *Olethreutes arcuella* (Clerck)] but can

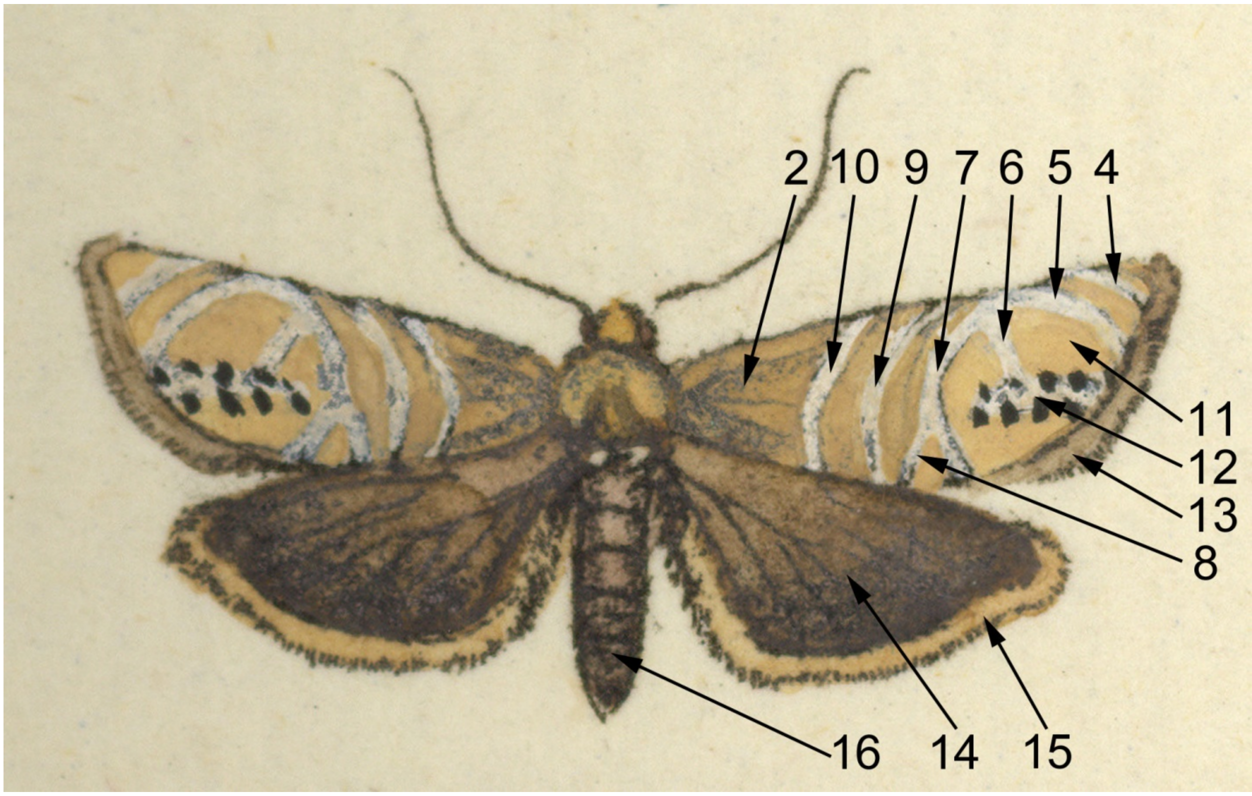


Figure 84. Pattern elements in Hübner's figure 363.

be differentiated by its pearly white stripes/the pearly white strips are outstanding. [Figures] 363.364. illustrate a female. *Lina. Syst. Phal. 296. *Arcuana*." The description provides little in the way of morphological information except for the resemblance to *O. arcuella* and emphasis on the "white stripes," which are interpreted as metallic-gray striae emanating from various paired costal strigulae.

Hübner's illustration shows a yellowish-brown forewing with darker suffusion at the base, a circular ocellular region with a prominent ocellus, and a rather extensive system of metallic-gray striae. We identified 17 morphological elements that could be used to compare the illustration with the *Eucosma* neotype candidates (Fig. 84, Table 114): 1: FW ground color yellowish-brown (not labeled); 2: basal portion of FW suffused with dark brown; 3: FW termen slightly concave (not labeled); 4: stria from strigula 8 or 9 to termen; 5: stria from strigula 7 to termen; 6: stria from strigula 6 or 7 to center of ocellular region; 7: stria from strigula 6 to dorsum (which, in combination with element 5, partially “encircles” ocellular region); 8: “branch” from element 7 to dorsum; 9: stria from strigula 5 to dorsum; 10: stria from strigula 4 to dorsum; 11: ocellular region unicolorous golden yellow apart from ocellus and element 6; 12: ocellus consisting of two rows of 4–5 black dashes or dots; 13: FW fringe pale brown; 14: HW dark brown, lighter towards base; 15: HW fringe contrastingly paler than HW ground color; 16: abdomen dark brown; 17: AR ca. 2.83 (not labeled).

Table 114. Pattern elements present in Hübner’s *E. circulana* illustration scored for candidate species (*Pe*. = *Pelochrista*; *Ph*. = *Phaneta*).

	1 (FW ground yellowish-brown)	2 (basal portion of FW dark brown)	3 (FW termen weakly concave)	4 (stria from strigula 8 or 9)	5 (stria from strigula 7)	6 (stria to center of ocellular region)	7 (stria from strigula 6)	8 (branch from element 7)	9 (stria from strigula 5)	10 (stria from strigula 4)	11 (ocellular region unicolorous golden yello	12 (ocellus with two rows of 4-5 marks)	13 (FW fringe pale brown)	14 (HW dark brown)	15 (HW fringe paler than ground color)	16 (abdomen dark brown)	**Total**	17 (FW aspect ration)
Hübner Fig. 363	1	1	1	1	1	1	1	1	1	1	1	1	1	1	1	1	16	2.83
E. circulana	1	1	1	1	1	0	1	1*	1*	0	1	1	1	1	1	1	14	2.79
E. gemellana	1	1*	1	1	1	0	1	0	1	0	0	1	1	1	1	1	12	2.93
E. paragemellana	1	1*	1	1	1	0	1	0	1	0	0	1	1	1	1	1	12	2.70
E. fraudabilis	1	0	0	1	1	0	1*	0	1	1	0	0	1	1	1	1*	10	2.76
Pe. scintillana	1	1	0	1	1	0	1	0	1	0	0	0	1	1	1	1	10	3.06
Pe. fratruelis	1	1	0	1	1	0	1	0	1	0	0	0	1	1	0	1	9	2.61
Ph. autumnana	1	1	1	1	1	0	1	0	1	0	0	1	1	1	1	1	12	3.12
Ph. verna	1	1	1	1	1	0	1	0	1	0	0	1	1	0	1	0	10	3.15
0 = element absent																		
1 = element present																		
1* = element remnant in some specimens																		

The North American species most closely resembling Hübner’s illustration are: *E. circulana* (*sensu* Heinrich) (Figs. 85.5–7), *E. gemellana* Heinrich (Figs. 85.8–11), *E.*

paragemellana Gilligan and Wright (Figs. 85.12–13), *E. fraudabilis* Heinrich (Figs. 85.14–15), *Pelochrista scintillana* (Clemens) (Figs. 86.16–18), *P. fratruelis* (Heinrich) (Fig. 86.19), *Phaneta verna* Miller (Figs. 86.20–21), and *P. autumnana* (McDunnough) (Figs. 86.22–24). Table 114 summarizes the extent to which each agrees with Hübner's illustration based on the 17 elements cited above. By those criteria, the best match is *E. circulana* (*sensu* Heinrich). It has the disadvantage that element 10 is not present and element 9 is only partially expressed. *Eucosma fraudabilis* is the only species with elements 9 and 10 conspicuously present, but its ocellus is least like that of Hübner's illustration (four rows of black dots vs. two), and the rest of its ocellular region is not golden yellow but is covered with blackish-brown scales with whitish apices. Curiously, the stria projecting into the ocellular region (element 6) is not found in any of the candidate species. All things considered, we conclude that *E. circulana* (*sensu* Heinrich) is the best available choice for the type species. Confirmed records of this species nearest to the type locality of Pennsylvania come from western Kentucky. Per Article 75 of the International Code of Zoological Nomenclature (International Commission on Zoological Nomenclature 1999), we have determined the name-bearing type of *E. circulana* to be lost or destroyed, and we have selected a specimen from Kentucky for the neotype. The formal designation occurs below in the description of *E. circulana*.

Descriptions and redescriptions

Eucosma circulana Hübner

(Figs. 85.5–7, 87.26, 88.30)

Eucosma circulana Hübner 1823: 28, Figs. 363–364.

Eucosma circulana; Hübner [1816–1825]: 374; Fernald [1903]:455; Fernald 1908: 4; Walsingham 1914: 226; Barnes & McDunnough 1917:169; Heinrich 1923:95; McDunnough 1939: 46; Obraztsov 1968:1; Powell 1983:34; Razowski 1989: 175; Razowski 2003: 84; Brown 2005:317.

Paedisca circulana: Fernald 1882:36; Walsingham 1884: 136; Fernald 1891: 91.

Diagnosis. *Eucosma* circulana (Figs. 85.5–7) is similar in forewing appearance to *E.gemellana* (Figs. 85.8–11) and *E. paragemellana* (Figs. 85.12–13) but lacks the whitish subcostal and basal streaking found in these two species. The three species are readily separated by male valval shape (Figs. 87.26–27, 29). In *E. circulana* the uncus is wider and more strongly developed, the neck is wider (NR = 0.41 vs. 0.28 and 0.20), the saccular angle is obtuse instead of acute (SA 101° vs. 89° and 75°), and the cucullus is much more elongate dorsally. Females are separated by the structure of sternum 7, which is emarginated to the length of the sterigma in *E. circulana*, and to 1¼ the length of the sterigma in the other two species (Figs. 88.30–31, 33).

Description. *Head*: Frons white; vertex creamy white medially, brownish laterally; labial palpus with medial surface creamy white, lateral surface brownish; antenna tan; ventral surface of scape creamy white. *Thorax*: Dorsal surface brown; fore- and mid-legs with anterior surfaces pale brown, posterior surfaces creamy white; hind-legs creamy white; tarsi with weakly contrasting white annulations. *Forewing* (Figs. 85.5–7): ♂ FWL 6.7–9.6 mm (mean =8.2, n = 26), AR = 2.80; ♀ FWL 8.0–10.7 mm (mean = 8.9, n = 8), AR = 2.77; costal margin nearly straight; apex acute; termen weakly concave; male with costal fold on dorsal surface from base to ca. 1/3 FWL; dorsal surface with proximal one-half brown to brownish yellow, distal one-half golden yellow, the latter edged with brown along costa, dorsum, and termen and containing a circular ocellular region that extends from tornus nearly to costa; ocellular region with golden yellow central field and metallic-gray circular boundary, the anterior semi-circle largely intact but with up to three short interruptions, the posterior semi-circle fragmentary, consisting of two or three dots and a short arc; ocellus (ca. between M_2 and CuA_1) consisting of two rows (and usually a partial third) of three to five black dots separated by metallic-gray scaling, the primary rows often at least partially confluent; costal strigulae not expressed; subcostal portion of golden yellow region marked with several lustrous gray striae and/or fragments of striae, the most

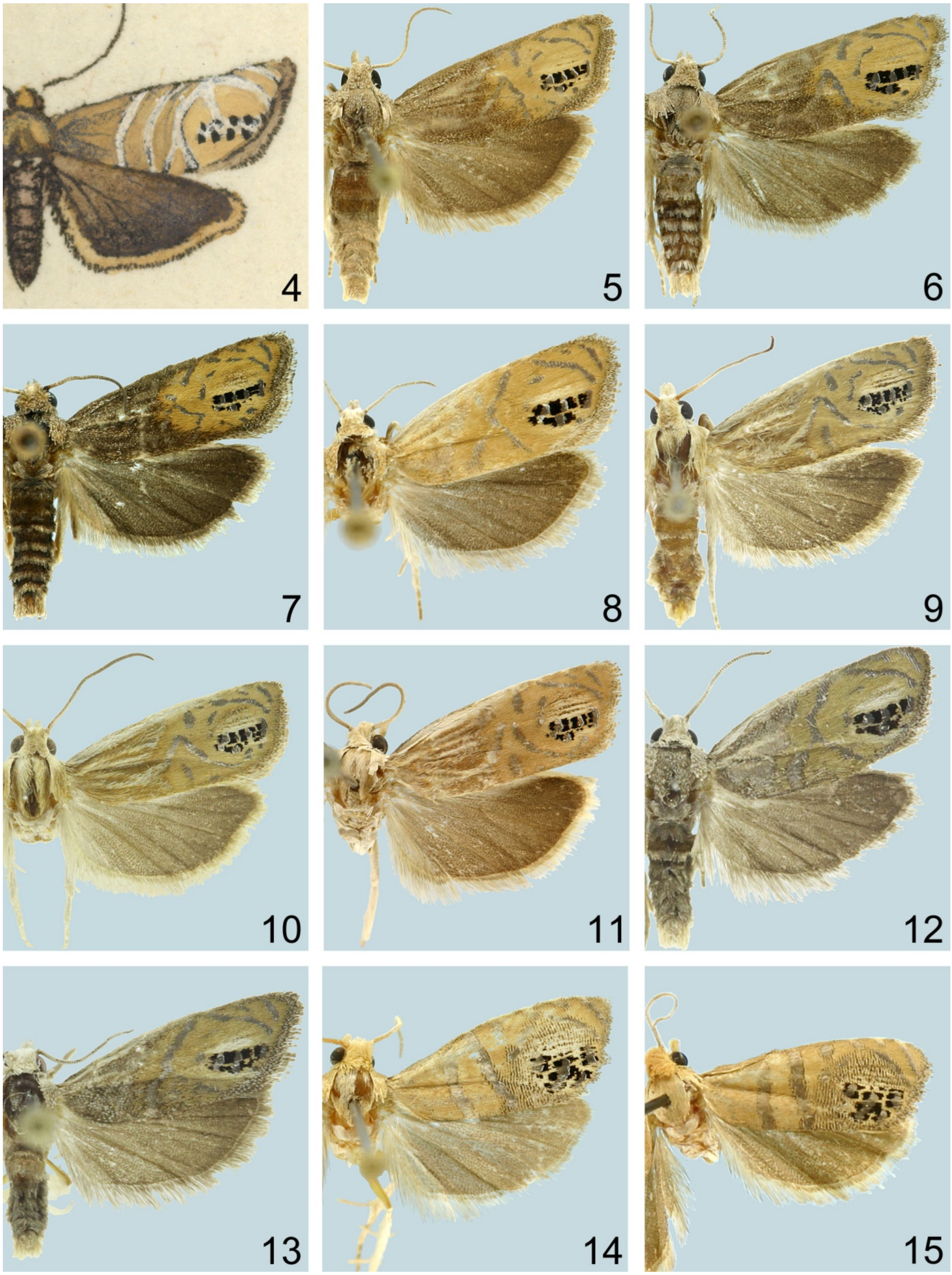


Figure 85. *Eucosma circulana* and closely related species; 4: Hübner’s illustration; 5–7: *E. circulana sensu* Heinrich; 8–11: *E. gemellana*; 12–13: *E. paragemellana*; 14–15: *E. fraudabilis*.

clearly defined being those arising at the positions of strigulae 6 and 9, with stria 6 often extending to inner margin after an interruption in the cell; fringe brown. *Hindwing*: brownish gray. *Male genitalia* (Fig. 87.26) (n = 5): Uncus moderately developed, divided medially by shallow indentation, and well differentiated from dorsolateral shoulders of tegumen; socii fingerlike; phallus stout, moderately long, and tapering distally; anellus approximate to phallobase; vesica with 25–40 (mean = 31, n = 3) deciduous cornuti; valva with costal margin concave, ventral emargination moderate, NR = 0.35–0.46 (mean = 0.41, n = 4), ventrolateral margin of neck scooped out, saccular corner angular, SA slightly obtuse, 96–108° (mean = 101°, n = 4); cucullus with apex semicircular, distal margin nearly straight, anal angle acute and moderately produced, setation of medial surface coarse toward distal margin and anal angle grading to fine toward costal margin and apex; vertex of anal angle with one ventrally projecting spiniform seta that is marginally stouter than adjacent setae on medial surface of cucullus. *Female genitalia* (Fig. 88.30) (n =2): papillae anales laterally facing and moderately setose; lamella postvaginalis rectangular (length ≈ width) and microspinulate; lamella antevaginalis ringlike; posterior margin of sternum 7 emarginated to length of sterigma and approximate to lamella antevaginalis and to lateral margins of lamella postvaginalis; scaling of sternum 7 dense on posterior projections and anterolateral margins, relatively sparse elsewhere; ductus bursae with sclerotized ring posterior to juncture with ductus seminalis, the latter arising midway between ostium and corpus bursae; corpus bursae with two large signa of approximately equal size.

Neotoype here designated (Fig. 85.5). ♂, USA, Kentucky, McCracken County, Paducah, Zip Track, D. J. Wright, 12 August 2008, 37° 03' 43.4" N, 88° 35' 57.9" W. Deposited in USNM.

Material Examined (26 ♂; 8 ♀). FLORIDA: Archer (1 ♀), MEM; Hastings, 8 June (1 ♂, slide USNM 70325, illustrated in Heinrich 1923, Fig. 148), USNM. KENTUCKY: same location as neotype, D. J. Wright, 12 August 2008 (12 ♂; 4 ♀), DJW, TMG; L. D. Gibson, 9 September 2006 (3 ♂, slides DJW 1700, 2004; 1 ♀), LDG; L. D. Gibson, 7 September 2007 (1 ♂), TMG; L. D. Gibson, 16 June 2007 (1 ♂), TMG. MISSISSIPPI: Hinds Co., Jackson, 31 March 1931 (1 ♀), MEM; Clinton, B. Mather, 27 February 1962 (1 ♂), MEM; Oktibbeha Co., 6 mi. SW Starkville, R. L. Brown, 18 April 1987 (1 ♂), MEM; nr. Hattiesburg, C.D.M., 1–15 August 1944, (1 ♂, slide DJW 1178), AMNH. TEXAS: Harris Co., Houston, A. & M. E. Blanchard, 5 June 1968 (1 ♂), 6 June 1968 (1 ♂, slide USNM 134150), 8 June 1968 (1 ♀, slide USNM 134151), 7 September 1965 (1 ♂), USNM.

Distribution (Fig. 89): Northeastern Florida, western Kentucky, Mississippi, east Texas; late February – early September.

Eucosma gemellana **Heinrich, new status**

(Figs. 85.8–11, 87.27, 88.31)

Eucosma circulana gemellana Heinrich 1923:96, Fig. 150.

Eucosma circulana gemellana; McDunnough 1939: 46; Powell 1983:34; Brown 2005:317.

Diagnosis. *Eucosma gemellana* (Figs. 85.8–11) is similar in forewing appearance to *E. paragemellana* (Figs. 85.12–13), and the two species cannot be reliably separated by forewing pattern. Males are separated by costal fold length, which is nearly twice as long in *E. gemellana* (0.38 FWL) as in *E. paragemellana* (0.21 FWL), and male genitalia. In *E. gemellana*, the anal angle of the cucullus is rounded, the area dorsal to the anal angle is free of setae, and the neck is wider (NR = 0.28 vs. 0.20). Females of the two species cannot be distinguished from each other. *Eucosma circulana* is similar to both *E. gemellana* and *E. paragemellana*; details for separating *E. circulana* are found under that species' diagnosis.

Description. *Head*: Frons and vertex creamy white; labial palpus creamy white with some pale brown tinting on lateral surface; antenna creamy white. *Thorax*: Dorsal surface creamy white to pale brown; fore- and mid-legs with anterior surfaces pale brown, posterior surfaces creamy white; hind-legs creamy white; tarsi with weakly contrasting white annulations. *Forewing* (Fig. 85.8–11): ♂ FWL 7.2–11.0 mm (mean = 9.1, n = 50), AR = 2.92; ♀ FWL 8.1–11.9 mm (mean = 10.2, n = 12), AR = 2.93; costal margin weakly arched; apex acute; termen weakly

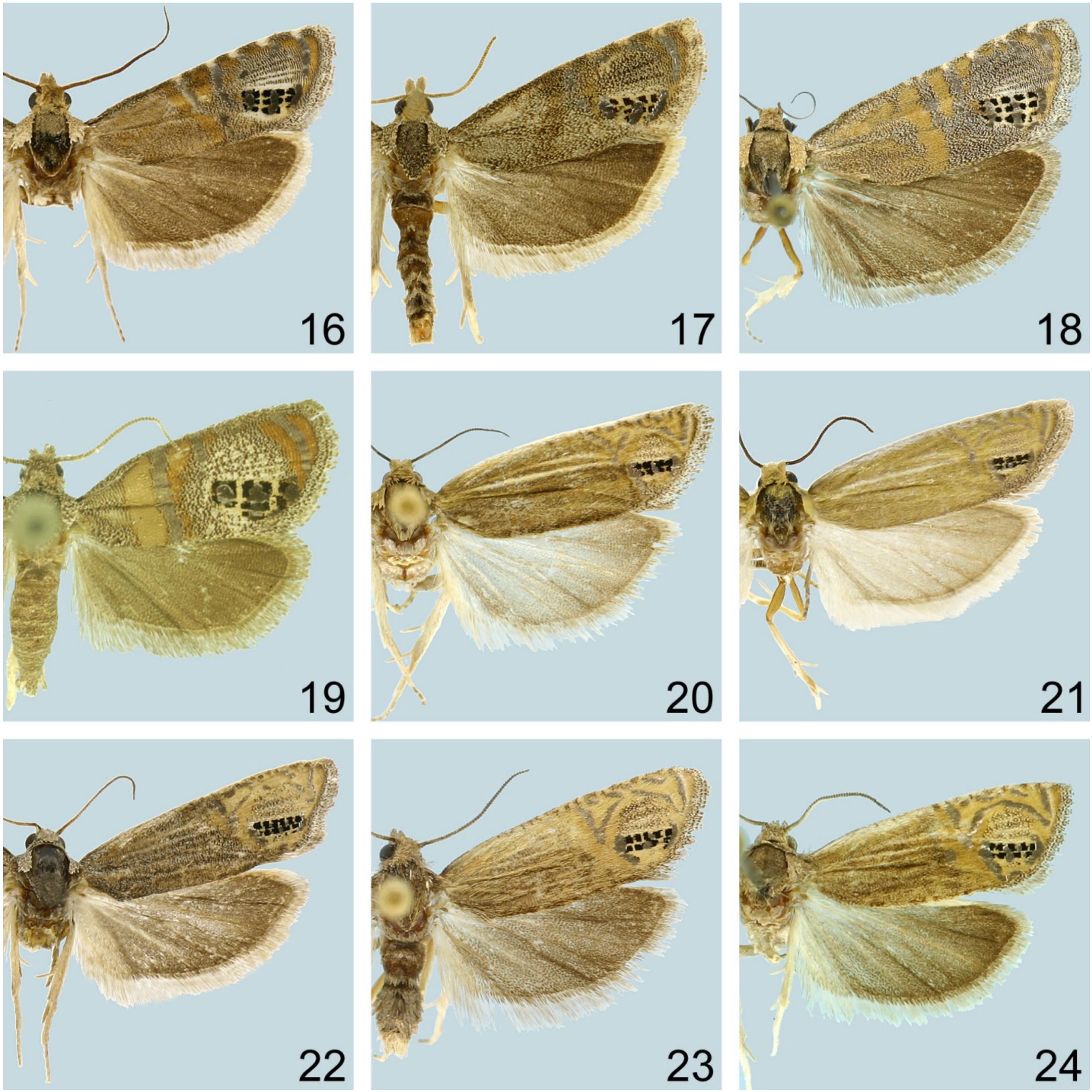


Figure 86. Species easily confused with *E. circulana*; 16–18: *Pelochrista scintillana*; 19: *P. fratruelis*; 20–21: *Phaneta verna*; 22–24: *Phaneta autumnana*.

concave; male with costal fold on dorsal surface from base to 0.33–0.43 FWL (mean = 0.38, n = 15); proximal one-third of dorsal surface brown to brownish yellow, with a creamy white subcostal band and sometimes with creamy white streaks in cell and on CuP and A_{1+2}; males with gray-brown edging on costa from base to end of fold; distal two-thirds of dorsal surface golden yellow, edged with brown along costa, dorsum, and termen, and containing a circular ocellular region; ocellular region similar to that of *E. circulana* except the circular metallic-gray boundary is reduced to a few small fragments, the central field has whitish scaling anterior to ocellus, the distal two columns of black dots in the ocellus are not separated by metallic-gray scaling, and the anterior one-half is sometimes marked by dark streaks along the medial veins; stria 6 forks at the cubitus, with both branches extending to dorsum. *Hindwing*: brownish gray. *Male genitalia* (Fig. 87.27) (n = 7): Uncus weakly produced but moderately well differentiated from dorsolateral shoulders of tegumen; socii finger-like, phallus stout, moderately long, and tapering distally; anellus approximate to phallobase; vesica with 20–40 (mean = 28, n = 4) deciduous cornuti; valva with costal margin weakly concave, ventral emargination deeply concave with nearly uniform curvature, NR = 0.23–0.36 (mean = 0.28, n = 7), ventrolateral margin of neck weakly scooped out, saccular corner angular, SA 85–91° (mean = 89°, n = 7); cucullus with apex rounded, distal margin weakly convex of nearly uniform curvature, anal angle rounded and moderately produced, setation of medial surface coarse toward distal margin and anal angle grading to fine toward costal margin and apex; anal angle with one spiniform seta that is well separated from adjacent setae on medial surface of cucullus. *Female genitalia* (Fig. 88.31) (n =3): papillae anales laterally facing and moderately setose; lamella postvaginalis microspinulate, consistently tapering towards ostium; lamella antevaginalis ring-like; posterior margin of sternum 7 emarginated to ca. 1¼ length of sterigma and closely approximate to lamella antevaginalis and fused to lateral margins of lamella postvaginalis; scaling of sternum 7 dense on posterior projections and anterolateral margins, relatively sparse elsewhere; ductus bursae with sclerotized ring posterior to juncture with ductus seminalis, the latter arising midway between ostium and corpus bursae; corpus bursae with two large signa of approximately equal size.

Holotype. ♂, Florida, [Pinellas Co.], St. Petersburg, USNM.

Paratypes. FLORIDA: [Pinellas Co.], St. Petersburg, R. Ludwig, 11 April 1914, USNM; Morrison, 1884 [determined as *Paedisca circulana* by Walsingham].

Additional Material Examined. **FLORIDA**: Charlotte Co., Punta Gorda, H. Ramstadt, 1–10 May 1903 (1 ♂), USNM; Citrus Co., Sandhill Crane NWR, H. D. Baggett, 2 June 1984 (1 ♂), MEM; Hwy 480 W Floral City, H. D. Baggett, 2 June 1984 (1 ♂), FSCA; Lecanto, J. Glaser, 1 October 1996 (1 ♂, slide TMG 601), USNM; Hernando Co., Croom Wildlife Preserve, W. L. Adair, 8 February 1989 (1 ♂), FSCA; Highlands Co., Highlands Hammock St. Park, G. J. Balogh, 5 November 1987 (1 ♂, slide DJW 330), DJW; Hillsborough Co., USF Ecology Area, W. L. Adair, 23 May 1992 (1 ♀), FSCA; Tampa, H. D. Baggett, 27 April 1984 (1 ♂), FSCA, (1 ♂, slide RLB 1781), MEM, 27 April 1989 (1 ♂), MEM; 9 October 1933, (1 ♂), USNM; Stemper, G. Krautwurm, 14 February 1911 (1 ♂), 7 October 1911 (1 ♂), CMNH, 1–7 July (1 ♂), USNM; Lee Co., Ft. Myers, 22 April 1912 (1 ♀), AMNH; Levy Co., Goethe State Forest, J. E. Hayden & G. E. Lee, 15 June 2012 (1 ♂), FSCA; Liberty Co., Wilma, J. Glaser, 30 September 1996 (2 ♂, slide DJW 2772), USNM; Marion Co., Ocala NF, Lake Delancy, J. S. Kutis, 10 April 1991 (1 ♂), 23 June 1991 (1 ♀), 28 June 1991 (1 ♂, slide TMG 606), 11 September 1991 (3 ♂), 21 September 1991 (1 ♂), 9 October 1991 (1 ♂, slide RLB 1783), 18 October 1990 (1 ♂), MEM; H. D. Baggett, 2 March 1985 (1 ♂), 10 April 1991 (1 ♂), 16 April 1990 (1 ♂), 22 June 1991 (2 ♂), 28 July 1991 (1 ♀), 11 August 1990 (1 ♂), 17 September 1991 (1 ♀, slide RLB 1782), 16 October 1990 (3 ♂), 18 October 1990 (1 ♂, slide TMG 611), 19 October 1991 (2 ♂), MEM; W. L. Adair, 31 October 1992, (1 ♂, 1 ♀), FSCA; Orange Co., Winter Park, A. B. Klots, May 1946 (1 ♂; 1 ♀, slide DJW 1179), AMNH; Pinellas Co., St. Petersburg, 16–23 February (2 ♂, slide TMG 597), 1–7 March (1 ♂, slide TMG 598), 11 April 1914 (1 ♂), 16–23 October (2 ♂), USNM; Putnam Co., U. of Florida Preserve, Welaka, H. D. Baggett, 19 September 1987 (1 ♀); Welaka, D. C. Ferguson, 17 April 1973 (1 ♂, slide USNM 124081), USNM; Santa Rosa Co., FAMU Biological Station nr. Holt, J. B. Heppner, 25–28 August 1986 (1 ♀), FSCA; Volusia Co., Cassadaga, S. V. Fuller, 3 May 1965 (1 ♂), 1 July 1964 (1 ♂), 3 September 1996 (1 ♀), 9

October 1962 (1 ♀), 15 December 1965 (1 ♂), FSCA. **MISSISSIPPI**: Forrest Co., Hattiesburg, C. D. M., 1–15 August 1944 (1 ♀, slide DJW 1345), AMNH.

Distribution (Fig. 89): Florida, southern Mississippi; mid-February – December.

Eucosma paragemellana **Gilligan & Wright, new species**

(Figs. 85.12–13, 87.29, 88.33)

Diagnosis. *Eucosma paragemellana* (Figs. 85.12–13) is similar in forewing appearance to *E. gemellana* (Figs. 85.8–11), and the two species cannot be separated based on forewing pattern. Males are separated by costal fold length, which is nearly half as long in *E. paragemellana* (0.21 FWL) as in *E. gemellana* (0.38 FWL), and male genitalia. In *E. paragemellana*, the anal angle of the cucullus is narrow and elongate, the entire cucullus from the anal angle to the apex is setose, and the neck is narrower (NR = 0.20 vs. 0.28). Females of the two species cannot be distinguished from each other. *Eucosma circulana* is similar to both *E. gemellana* and *E. paragemellana*; details for separating *E. circulana* are found under that species' diagnosis.

Description. *Head and Thorax*: As in *E. gemellana*. *Forewing* (Figs. 85.12–13): ♂ FWL 5.7–7.5 mm (mean = 6.3, n = 50), AR = 2.66; ♀ FWL 6.6–7.0 mm (mean = 6.7, n = 8), AR = 2.73; costal margin weakly arched; apex acute; termen weakly concave; male with costal fold on dorsal surface from base to 0.16–0.28 FWL (mean = 0.21, n = 21); proximal one-third of dorsal surface brown to brownish yellow, with a creamy white subcostal band and sometimes with creamy white streaks in cell and on CuP and A_{1+2}; distal two-thirds of dorsal surface golden yellow, edged with brown along costa, dorsum, and termen, and containing a circular ocellular region; ocellular region similar to that of *E. circulana* except the circular metallic-gray boundary is reduced to a few small fragments, the central field has whitish scaling anterior to ocellus, the distal two columns of black dots in the ocellus are not separated by metallic-gray scaling, and the anterior one-half is sometimes marked by dark streaks along the medial veins; stria 6 forks

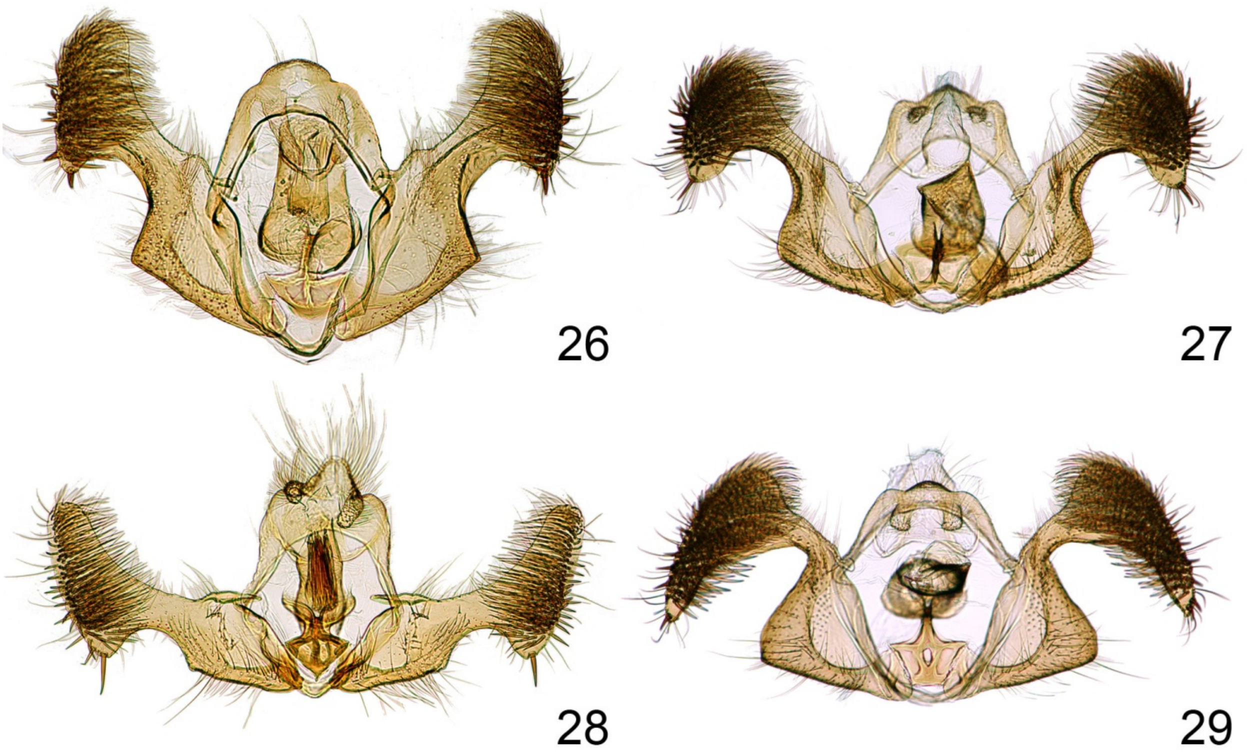


Figure 87. Male genitalia; 26: *E. circulana*; 27: *E. gemellana*; 28: *E. fraudabilis*; 29: *E. paragemellana*.

at the cubitus, with both branches extending to dorsum. *Hindwing*: brownish gray. *Male genitalia* (Fig. 87.29) (n = 7): Uncus weakly produced but moderately well differentiated from dorsolateral shoulders of tegumen; socii finger-like, phallus stout, moderately long, and tapering distally; anellus approximate to phallobase; vesica with ca. 25 deciduous cornuti (n = 1); valva with costal margin weakly concave, ventral emargination deep, NR = 0.17–0.25 (mean = 0.20, n = 7), ventrolateral margin of neck weakly scooped out, saccular corner angular, SA acute 65–86° (mean = 75°, n = 7); cucullus with apex rounded, distal margin weakly convex, anal angle acute and strongly produced, cucullus setose from anal angle to apex, setation of medial surface coarse toward distal margin and anal angle grading to fine toward costal margin and apex; anal angle with one or two spiniform seta. *Female genitalia* (Fig. 88.33) (n =3): As in *E. gemellana*.

Holotype (Fig. 85.12). ♂, Alabama, Baldwin Co., Weeks Bay Estuarine Reserve, D. J. Wright, 22 June 2008. Deposited in USNM.

Paratypes. **ALABAMA**: Baldwin Co., Weeks Bay Estuarine Reserve, R. L. Brown, 20 June 2001 (2 ♂), MEM; D. J. Wright, 21 June 2008 (5 ♂, slide DJW 2050), 22 June 2008 (6 ♂; 2 ♀, slide DJW 2773), DJW; T. M. Gilligan, 21 June 2008 (5 ♂), 22 June 2008 (2 ♂; 1 ♀), TMG. **MISSISSIPPI**: Harrison Co., Pass Christian, R. Kergosien, 6 August 1994 (1 ♂, slide TMG 612), MEM; Jackson Co., 1 mi. W Hwy 90 & 57, R. L. Brown, 5 April 1991 (1 ♂), 12 April 1991 (1 ♂), D. M. Pollock, 5 April 1991 (1 ♂), MEM; I-10 at Escatawpa River, R. L. Brown, 13 April 1991 (1 ♂, slide TMG 607), MEM; I-10 1 mi. W Ala., T. L. Schiefer, 22 July 1989 (1 ♂, slide DJW 2006; 3 ♀), MEM, DJW; Sandhill Crane NWR, J. A. MacGown, 29 August 1995 (3 ♂), 7 September 1994 (3 ♂; 1 ♀), MEM; Grand Bay Savanah, J. A. MacGown, 6 September 1994 (1 ♂), MEM. **NORTH CAROLINA**: Pender Co., Holly Shelter Game Land, J. B. Sullivan, 1 July 1998 (2 ♂, slide TMG 595), 7 August 1997 (4 ♂), 26 August 1997 (9 ♂, slide TMG 603; 1 ♀), 26 September 1995 (1 ♂), USNM.

Distribution (Fig. 89): Gulf Coast of Alabama and Mississippi, North Carolina coast; April – September.

***Eucosma fraudabilis* Heinrich**

(Figs. 85.14–15, 87.28, 88.32)

Eucosma fraudabilis Heinrich 1923:98, Fig. 161.

Eucosma fraudabilis; McDunnough 1939:46; Powell 1983:34; Brown 2005:320.

Diagnosis. *Eucosma fraudabilis* (Figs. 85.14–15) is similar in forewing appearance the three other species described here. It is distinguished by the two metallic striae running from strigula 4 and 5 on the costa to dorsum.

Description. *Head*: Frons creamy white; vertex straw yellow; labial palpus creamy white with pale brown tints on lateral surface of second segment; antenna concolorous with vertex. *Thorax*: Dorsal surface straw yellow; fore- and mid-legs with anterior surfaces pale brown, posterior surfaces creamy white; hind-legs creamy white; tarsi with weakly contrasting white annulations. *Forewing* (Figs. 85.14–15): ♂ FWL 5.1–7.5 mm (mean = 6.9, n = 20), AR = 2.70; ♀

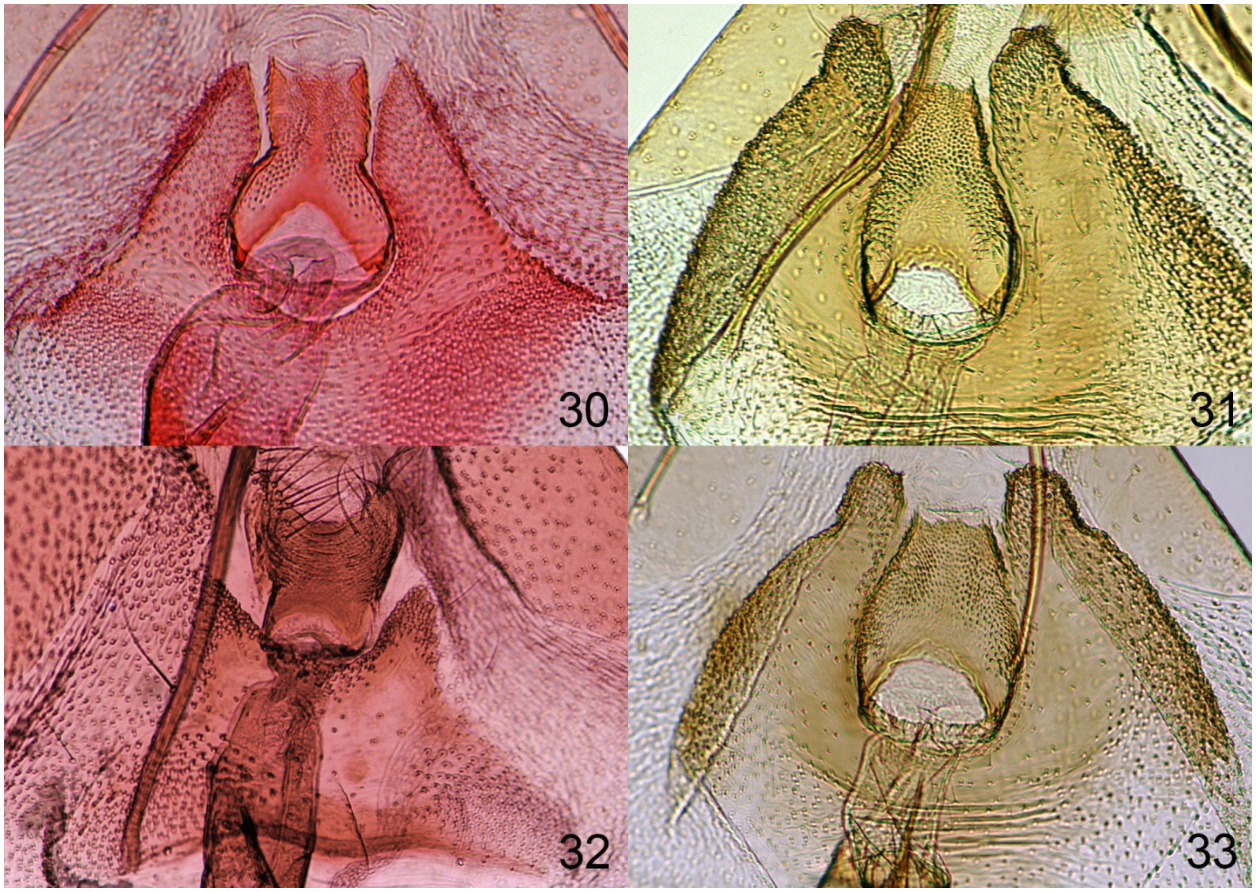


Figure 88. Female genitalia; 30: *E. circulana*; 31: *E. gemellana*; 32: *E. fraudabilis*; 34: *E. paragemellana*.

FWL 7.6 mm (n = 1), AR = 2.81; costal margin weakly arched; apex acute; termen weakly convex; male with costal fold on dorsal surface from base to ca. 1/3 FWL; dorsal surface straw yellow with metallic-gray subbasal and median bands from costa to inner margin and a large circular ocellular region extending from tornus nearly to costa, ocellular region with apical quadrant bounded by metallic-gray arc and with scales in anterior portion gray brown with creamy white apices; ocellus consisting of four obscurely defined rows of black dots on a creamy white field, the rows divided medially by a transverse metallic-gray bar and capped proximally and distally by smaller such bars; subcostal area anterior to ocellular region marked with lustrous gray striae and/or fragments thereof; distal one-half of costa with obscure whitish strigulae; fringe brown. *Hindwing*: brownish gray. *Male genitalia* (Fig. 87.28) (n = 5): Uncus strongly produced, tapering distally, and strongly differentiated from dorsolateral shoulders of tegumen; socii finger-like; phallus stout, moderately long, and tapering distally; anellus approximate to phallobase; vesica with 9–20 (mean = 15, n = 2) deciduous cornuti; valva with costal margin concave, ventral emargination moderate, NR = 0.42–0.59 (mean = 0.52, n = 5), saccular corner rounded, SA obtuse, 127–150° (mean = 139°, n = 5), distal margin of basal excavation produced into tablike pulvinus; cucullus with apex rounded, distal margin weakly convex of nearly uniform curvature, anal angle acute and moderately produced, setation of medial surface coarse toward distal margin and anal angle grading to fine toward costal margin and apex; anal angle with one spiniform seta of ca. twice the size of adjacent setae on medial surface. *Female genitalia* (Fig. 88.32) (n =1): Papillae anales with posterior lobes facing ventrally and anterior lobes produced into laterally facing ventral extensions; margins of posterior lobes with long ventrally curving setae; margins of anal opening with moderately long hook-tipped setae; lamella postvaginalis gradually widening posteriorly, with posterior margin concavely emarginated; lamella antevaginalis ring-like; posterior margin of sternum 7 concavely emarginated to one-half length of sterigma and diverging from lateral margins of sterigma; scaling of sternum 7 dense on posterior projections and anterolateral corners, relatively sparse elsewhere; ductus bursae with elongate sclerotized patch posterior to juncture with ductus seminalis, the latter located closer to ostium than corpus bursae; corpus bursae with two signa, one much larger than the other.

Holotype (Fig. 85.15). ♂, North Carolina, [Moore Co.], Southern Pines, USNM.

Paratypes. North Carolina, [Moore Co.], Southern Pines, 1–7 June (1 ♂), 8–15 June (1 ♂, slide TMG 600), 1–7 July (2 ♂), 8–15 July (3 ♂; 1 ♀, slide USNM 124084), [1 ♀ *P. scintillana*, slide TMG 599], 16–23 July (1 ♂), USNM.

Additional Material Examined. **FLORIDA**: Suwannee Co., H. D. Baggett, 27 June 1981 (1 ♂), MEM; Duval Co., Jax. Police Academy, H. D. Baggett, 20 August 1980 (1 ♂), MEM. **GEORGIA**: Emanuel Co., Ohoopee N. A., R. L. Brown, 16 June 2002 (3 ♂), MEM. **LOUISIANA**:

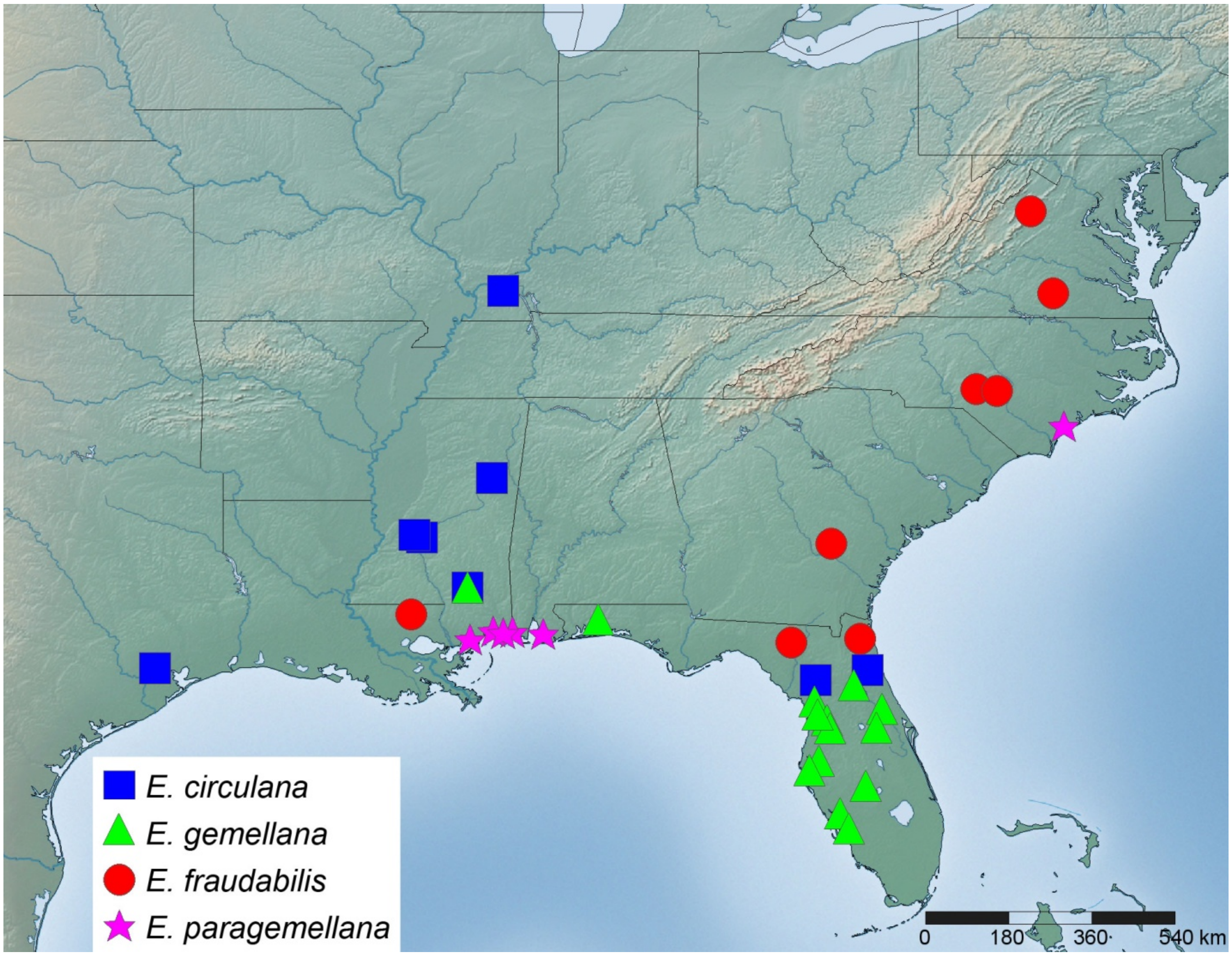


Figure 89. Distribution map for *E. circulana*, *E. gemellana, E. fraudabilis,* and *E. paragemellana*.

Tangipahoa Parish, Sandy Hollow WLMA, D. P. Pashley, 26 May 1993 (1 ♂), MEM, 31 May 1994 (1 ♂, slide DJW 2017), DJW. **NORTH CAROLINA**: Hoke Co., Ft. Bragg, 0.5 mi NE Jct. Blues Rd & Manchester Rd., J. B. Sullivan & S. P. Hall, 20 June 2001 (1 ♂), USNM; Moore Co., Southern Pines, 8–15 July (1 ♂, slide DJW 1329), AMNH. **VIRGINIA**: Nottoway Co., Ft. Pickett Mil. Res. Grassland, 0.8km E. jct. Wilcox & Range Rd., A. C. Chazal, 6 July 2000 (1 ♂, slide USNM 118545), USNM; Page Co., Skyland, H. G. Dyar, 15 July 1911 (1 ♂, slide USNM 70355), USNM.

Distribution (Fig. 89): Florida, Georgia, Louisiana, North Carolina, Virginia; late May – mid-August.

References

Baixeras, J. 2002. An overview of genus level taxonomic problems surrounding *Argyroplace* Hübner (Lepidoptera: Tortricidae), with description of a new species. Annals of the Entomological Society of America. 95: 422–431.

Barnes, W. and J. McDunnough. 1917. Checklist of the Lepidoptera of Boreal America. Decatur, Illinois. 200 pp.

Brown, J. W. 2005. World catalogue of insects. Volume 5: Tortricidae (Lepidoptera). Apollo Books, Stenstrup, Denmark. 741 pp.

Brown, J. W. and J. A. Powell. 1991. Systematics of the *Chryoxena* group of genera (Lepidoptera: Tortricidae: Euliini). University of California Publications in Entomology 111. 87 pp.

Brown, R. L. and J. A. Powell. 1991. Description of a new species of *Epiblema* (Lepidoptera: Tortricidae: Olethreutinae) from coastal redwood forests in California with an analysis of the forewing pattern. Pan-Pacific Entomologist. 67(2): 107–114.

Calhoun, J. V. 2003. The history and true identity of *Melitaea ismeria* (Nymphalidae): A remarkable tale of duplication, misinterpretation, and presumption. Journal of the Lepidopterists' Society. 57(3): 204–219.

Clemens, B. 1865. North American Micro-Lepidoptera. Proceedings of the Entomological Society of Philadelphia. 5: 133–147.

Engel, H. 1908. A preliminary list of the Lepidoptera of western Pennsylvania collected in the vicinity of Pittsburgh. Annals of the Carnegie Museum. 5: 27–136.

Fernald, C. H. 1882. A synonymical catalogue of the described Tortricidae of North America north of Mexico. Transactions of the American Entomological Society. 10: 1–64.

Fernald, C. H. 1891. Tortricina, pp. 88–94. *In*: Smith, J. B. (ed.), List of Lepidoptera of Boreal America. American Entomological Society of Philadelphia.

Fernald, C. H. 1903 (1902). Family Tortricidae, pp. 448–489. *In*: Dyar, H. G. (ed.), A list of North American Lepidoptera. Bulletin of the United States National Museum, No. 52.

Fernald, C. H. 1908. The genera of Tortricidae and their types. Carpenter & Morehouse, Amherst, Massachusetts. 68 pp.

Geyer, C. 1832. pp. 1–48. *In*: Hübner, J., Zuträge zur Sammlung exotischer Schmettlinge [sic]. Augsburg [no publisher listed].

Gilligan, T. M., D. J. Wright and L. D. Gibson. 2008. Olethreutine moths of the midwestern United States, an identification guide. Bulletin of the Ohio Biological Survey, Volume 16(2). 334 pp.

Gilligan, T. M., J. Baixeras, J. W. Brown and K. R. Tuck. 2012. T@RTS: Online World Catalogue of the Tortricidae (Ver. 2.0). http://www.tortricid.net/catalogue.asp.

Heinemann, H. 1863. Die Schmetterlinge Deutschlands und der Schweiz. 2. Abt. Kleinschmetterlinge. Band I. Heft I. Die Wickler. Braunschweig. 248 pp.

Heinrich, C. 1923. Revision of the North American moths of the subfamily Eucosminae of the family Olethreutidae. Bulletin of the United States National Museum. 123: 1–128.

Hemming, F. 1937a. Hübner : a Bibliographical and Systematic Account of the Entomological Works of Jacob Hübner and of the Supplements thereto by Carl Geyer, Gottfried Franz von Frölich and Gottlieb August Wilhelm Herrich-Schäffer. Vol. 1. Royal Entomological Society of London, London. 605 pp.

Hemming, F. 1937b. Hübner: a Bibliographical and Systematic Account of the Entomological Works of Jacob Hübner and of the Supplements thereto by Carl Geyer, Gottfried Franz Von Frölich and Gottlieb August Wilhelm Herrich-Schäffer. Vol. 2. Royal Entomological Society of London, London. 274 pp.

Horak, M. 2006. Olethreutine moths of Australia (Lepidoptera: Tortriacidae). Monographs on Australian Lepidoptera 10. 522 pp.

Hübner, J. 1806–[1832]. Sammlung exotischer Schmetterlinge. Volume 1. Augsburg [no publisher listed]. 36 pp. [unnumbered], 213 pls.

Hübner, J. [1816–1825]. Verzeichniss bekannter Schmettlinge [sic]. Augsburg [no publisher listed]. 431 pp.

Hübner, J. [1819–1832]. Sammlung exotischer Schmetterlinge. Volume 2. Augsburg [no publisher listed]. 44 pp. [unnumbered], 225 pls.

Hübner, J. 1823. Zuträge zur Sammlung exotischer Schmettlinge [sic], bestehend in Betundigung einzeiner Fliegmuster neuer oder rarer Nichteuropäischer Gattungen. Zwentes Hundert. Augsburg [no publisher listed]. 32 pp., 33 pls. published [1819–1822].

International Commission on Zoological Nomenclature. 1999. International Code of Zoological Nomenclature, Fourth Edition. 306 pp.

Kearfott, W. D. 1905. Descriptions of new species of tortricid moths from North Carolina, with notes. Proceedings of the U. S. National Museum. 28: 349–364.

Kearfott, W. D. 1907. New North American Tortricidae. Transactions of the American Entomological Society. 33: 42–43.

McDunnough, J. H. 1939. Check list of the Lepidoptera of Canada and the United States of America. Part II. Microlepidoptera. Memoirs of the California Academy of Sciences. 2: 3–717.

Obraztsov, N. S. 1968. Die Gattungen der Palaearktischen Tortricidae II Die Unterfamilie Olethreutinae. 8 Teil und Schluss. Tijdschrift voor Entomologie. 111: 1–48.

Pierce, F. N. and J. W. Metcalfe. 1922. The genitalia of the group Tortricidae of the Lepidoptera of the British Islands. Oundle, Liverpool, England. 101 pp.

Powell, J. A. 1983. Tortricidae, pp. 31–41. *In*: Hodges, R. W. (ed.), Check list of the Lepidoptera of America north of Mexico. E. W. Classey & Wedge Entomological Research Foundation, London.

Razowski, J. 1989. The genera of Tortricidae (Lepidoptera). Part II: Palaearctic Olethreutinae. Acta Zoologica Cracoviensia. 32: 107–328.

Razowski, J. 2003. Tortricidae of Europe, Volume 2, Olethreutinae. František Slamka. 301 pp.

Riley, C. V. 1886. *Aletia xylina* vs. *A. argillacea*. Entomologica Americana. 1(9): 161–163.

Walker, F. 1863. List of the specimens of lepidopterous insects in the collection of the British Museum. Part XXVIII. Tortricites & Tineites. British Museum (Natural History), London. 287–561 pp.

Walsingham, Lord T. de Gray. 1884. North American Tortricidae. Transactions of the Royal Entomological Society of London. 32: 121–147.

Walsingham, Lord T. de Grey. 1897. Revision of the West-Indian Microlepidoptera with descriptions of new species. Proceedings of the Entomological Society of London. 1897: 54–183.

Walsingham, Lord T. de Grey. 1914 (1909–1915). Insecta. Lepidoptera-Heterocera, Vol. IV. Tineina, Pterophorina, Orneodina, and Pyralidina and Hepialina (part), pp. 1–482. In: Godman, F. D. and O. Salvin (eds.), Biologia Centrali-Americana. R. H. Porter, London.

Wilkinson, S. J. 1859. The British Tortrices. John van Voorst, London. 328 pp.

Zeller, P. C. 1875. Beitrage zur Kenntniss der nordamericanischen Nachtfalter, besonders der Microlepidopteren. Verhandlungen der Zoologisch-Botanischen Gesellschaft in Wien. 25: 205–360.

Chapter 9

Molecular phylogeny of *Eucosma* Hübner and related genera (Lepidoptera: Tortricidae: Eucosmini): A revised classification and updated world checklist*

Introduction

Eucosma Hübner is the largest genus in the Tortricidae, consisting of 298 described species (Gilligan et al. 2012). *Eucosma* is closely related to three other genera: *Pelochrista* Lederer, consisting of 87 described species, *Phaneta* Stephens, consisting of 119 described species, and *Epiblema* Hübner, consisting of 89 described species (Gilligan et al. 2012). The four genera are Holarctic and most exhibit highest levels of species diversification in western North America. With few exceptions, larvae of all four genera are stem- or root-borers in Asteraceae, and this group may represent the largest radiation of Lepidoptera onto hosts in the Asteraceae (R. Brown, pers. comm. 2012).

Historically, there has been confusion in the circumscription of *Eucosma* and related genera. Hübner (1823) described *Eucosma* from a specimen of *E. circulana* collected in Pennsylvania, and the application of *Eucosma* was restricted to the North American type until the end of the 19th century. *Pelochrista* was described by Lederer (1859) as a subgenus of *Grapholitha* with *Paedisca mancipiana* Mann as the type species; Walker (1863) elevated

---

*This chapter will be submitted for publication as:

Gilligan, T. M., D. J. Wright, K. Yakobson, J. Munz & M. P. Simmons. Molecular phylogeny and revised classification of *Eucosma* Hübner and related genera (Lepidoptera: Tortricidae: Eucosmini).

Gilligan, T. M. & D. J. Wright. A revised world checklist of *Eucosma*, *Pelochrista*, and *Phaneta* (Lepidoptera: Tortricidae: Eucosmini).

Pelochrista to generic status several years later. In the latter part of the 19th and early part of the 20th centuries, various authors (e.g., Fernald 1882; Walsingham 1897) treated *Eucosma* as synonymous with *Paedisca*. Fernald (1908), in his seminal publication on tortricid types, synonymized 25 generic names, including *Pelochrista*, with *Eucosma*. This trend continued with Walsingham (1914), who expanded on Fernald's list by including a total of 38 generic names as synonyms under *Eucosma*.

Major advances in tortricid taxonomy in the early part of the 20th Century resulted from the use of genitalic characters, pioneered by Dampf (1908). Pierce and Metcalfe (1922) produced the first comprehensive taxonomic work to include genitalia descriptions and illustrations for many European species. They treated *Eucosma* as the modern-day *Pelochrista*, likely because of misinterpretation of the type species (Gilligan and Wright 2012 [Chapter 8]), and used *Catoptria* Guenée to place the remainder of the species that would today be considered *Eucosma*. Heinrich (1923) relied heavily on male genitalic characters in his revision of the Nearctic Eucosminae, in which he resurrected many of the generic names previously synonymized by Fernald (1908) and Walsingham (1914). He correctly interpreted the type species of *Eucosma* (Gilligan and Wright 2012 [Chapter 8]) but retained the synonymy between *Eucosma* and *Pelochrista* following Fernald (1908). Much of Heinrich's classification system is still in use today.

In the middle of the 20th Century, Nicholas Obraztsov began a revision of the Palearctic Tortricidae. In his initial classification system for the Eucosmini (1946) he attempted to reconcile the differences between the treatment of *Eucosma* and closely related genera in North America and Europe. He synonymized *Thiodia*, *Semasia*, *Phaneta*, and part of Heinrich's (1923) *Eucosma* under *Catoptria* and transferred Pierce and Metcalfe's (1922) *Eucosma* (= *Pelochrista*) species to *Pseudeucosma*. In the next decade, Obraztsov refined his concepts of tortricid classification, thanks, in part, to continued communication with other tortricid experts at the time, such as Bradley, Hannemann, and Kuznetsov. His new Eucosmini classification first appeared in Agenjo's (1955) list of Spanish fauna, where several Palearctic species were listed under *Eucosma*. Hannemann (1961) followed suit, including many species in *Eucosma* and dividing the genus into three subgenera: *Eucosma*, *Phaneta*, and *Pygolopha*. Obraztsov's posthumous publications (1967, 1968) produced the most influential changes to Heinrich's classification system. He treated the subfamily Eucosminae as the tribe Eucosmini and separated *Pelochrista* from *Eucosma* based on differences in the male and female genitalia. Following Hannemann (1961), he included numerous subgenera under *Pelochrista* (*Pelochrista* and *Pseudeucosma*) and *Eucosma* (*Eucosma*, *Palpocrinia*, *Phaneta*, and *Pygolopha*), although the subgenera were not retained by subsequent authors (e.g., Razowski 1989). Powell (1983) followed Obraztsov in assignment of species to genera in the Eucosmini in his catalogue of North American tortricids.

Although Obraztsov contributed greatly towards the understanding of higher olethreutine taxonomy, he also created one of the most significant sources of inconsistency in the assignment of species to the genera *Eucosma* and *Phaneta*. Throughout the 19th century, a large number of Eucosmini were assigned to the genus *Semasia* Stephens (= *Cydia*). Walsingham (1897) recognized that these species did not belong to the same genus as *Cydia* and treated *Semasia* as a synonym of *Thiodia* Hübner, with *Tortrix citrana* Hübner as the type species. Fernald (1903) followed the same convention, and listed the North American taxa treated as *Semasia* in his previous publications (1882, 1891) under *Thiodia*; the genus was subsequently synonymized under *Eucosma* by Walsingham (1914). Heinrich (1923) resurrected *Thiodia* for a group of species previously placed in *Eucosma* in an attempt to better circumscribe this large genus in North America. He used the male costal fold as the defining character, stating that "…use of the [costal] fold enables division and easier handling of what would otherwise be a most unwieldy group" (pg. 33). In this arrangement, species placed under *Eucosma* possessed a costal fold, and those placed under *Thiodia* lacked a costal fold. Heinrich realized at the time that this division was simply "a convenience" and that the type of *Thiodia*, *T.*

citrana, differed from the North America species in the male genitalia. Obraztsov (1952) compared the Nearctic and Palearctic species of *Thiodia* and correctly determined that they did not belong to the same genus. According to Obraztsov: “The American species [of *Thiodia*] are closely related to *Eucosma* Hb. while the Palearctic *Thiodia* have more resemblance to *Rhyacionia* Hb. in their genitalia.” To alleviate this problem, Obraztsov (1952) transferred all of the Nearctic *Thiodia* (as listed in Heinrich 1923) to the genus *Phaneta*, which was monotypic at the time, consisting of only the type species, *Cochylis pauperana* Duponchel. Obraztsov’s action created a large discrepancy between the interpretation of *Phaneta* and *Eucosma* in North America and Europe: Palearctic *Phaneta* consisted of a single species, whereas the Nearctic fauna consisted of close to 100 species, all of which would be placed in *Eucosma* (or *Pelochrista*) if described from Europe. This division has remained in the literature, with North American authors (e.g., Miller 1987; Gilligan et al. 2008) retaining Obraztsov’s expanded definition of *Phaneta*, and European authors (e.g., Razowski 2003) restricting *Phaneta* to one or two species.

Two primary issues have led to the confusion in assignment of species to the genera discussed here: a lack of morphological synapomorphies defining *Eucosma* and *Pelochrista*, and the unfounded importance placed on the presence or absence of the male costal fold in *Phaneta* and *Eucosma* by Obraztsov (1952). The type of *Eucosma*, *E. circulana*, was lost or destroyed and taxonomic confusion has made determination of the type species difficult; the taxonomic history, descriptions of closely related species, and declaration of a Neotype for *E. circulana* are detailed in Gilligan and Wright (2012 [Chapter 8]). Traditional male genitalic characters used to separate *Eucosma* and *Pelochrista* include the presence or absence of a lobe, or pulvinus, on the basal excavation of the valva (Obraztsov 1967, 1968; Razowski 1989), and the presence or absence of a single large spiniform seta on the cucullus (Heinrich 1923; Wright 2007). However, reduction or loss of structures in the male genitalia has resulted in many inconsistencies and exceptions. *Eucosma* has also been confused with *Epiblema* because of loss of the “clasper” on the male valva in some species (Razowski 1989). The male costal fold has been used in tortricid classification since Heinemann (1863), although it had been noted as unreliable by early taxonomists (e.g., Barrett 1885), and by Heinrich himself (1923). There are currently several tortricid genera in which a male costal fold is both present or absent (e.g., *Clepsis*, *Dichrorampha*, *Epinotia*).

Another inconsistency in the current circumscription of *Eucosma* is the inclusion of several Pinaceae-feeding species in the genus. While the overwhelming majority of *Eucosma* are stem- or root-borers in Asteraceae, these species bore into shoots or cones of *Pinus* or other coniferous hosts (Powell 1968; Gilligan et al. 2008). Their inclusion in *Eucosma* is the result of three species descriptions by Kearfott (1907), and subsequent authors (Heinrich 1920, 1931; Powell 1968) continued to describe new species in the same genus. There are currently 12 such Pinaceae-feeders, all restricted to North America.

The goals of this study are to test the monophyly of *Eucosma*, *Pelochrista*, *Phaneta*, and *Epiblema* as they are currently defined, identify morphological character states that can be used to define each genus, examine the relationships of the Pinaceae-feeding *Eucosma* to the rest of the genus, and provide an updated classification and world catalogue for species in these genera. As previously defined morphological characters have been unreliable in separating these genera, a molecular data set consisting of 2692 bp was generated using the mitochondrial gene cytochrome c oxidase 1 (COI; 658 bp) and the nuclear genes carbamoylphosphate synthetase/aspartate transcarbamylase/dihydroorotase (CAD; 638 bp), elongation factor-1a (EF-1α; 568 bp), and 28S ribosomal DNA (28S rDNA; 828 bp). These data were analyzed together with a morphological character set.

Methods

Taxon sampling

A total of 71 taxa were sampled (Appendix II), comprised of 60 ingroup and 11 outgroup taxa. Two specimens were sampled for seven of the ingroup taxa, and three specimens sampled for two of the ingroup taxa, for a total of 82 terminals included in the simultaneous analysis (Kluge 1989; Nixon and Carpenter 1996). All specimens are stored in 95–100% ethanol and will be deposited in the ATOLep alcohol collection at the University of Maryland (Regier et al. 2012; details at http://www.leptree.net/collection).

Ingroup taxa consisted of a broad sample of primarily North American *Eucosma* and *Pelochrista*, including four species in the Pinaceae-feeding *Eucosma* group. Thirteen species of North American *Phaneta* were also included, along with six species of *Epiblema*. We were unable to obtain a specimen of the *Phaneta* type species, *P. pauperana*, although several morphological synapomorphies clearly demonstrate that the European and North American *Phaneta* do not belong to the same genus (Razowski 1989). Outgroup taxa consisted of three Microcorsini, two Olethreutini, and six additional Eucosmini.

Morphological characters

Twenty-seven morphological characters were coded for structures of the male and female genitalia traditionally used to define species in the Eucosmini (Appendix III). When applicable, characters were coded using states defined by Horak (2006). The presence or absence of a male forewing costal fold and two wing venation characters were also coded for all species. Other structures on the head, thorax, legs, and wings (Horak 2006, Gilligan et al. 2008) were examined and determined to be invariant or otherwise parsimony uninformative for the ingroup.

Molecular methods

Total genomic DNA was extracted using a Qiagen DNeasy Blood and Tissue Kit (Qiagen, Valencia, Calif.). One to three legs from an adult moth were crushed, incubated overnight at 56°C, and eluted in 100 µl of AE buffer after following the manufacturer's recommended protocol. All four gene-regions were amplified using conventional PCR on an Eppendorf Mastercycler gradient 5331 thermal cycler (Eppendorf AG, Hamburg, Germany). Reactions were performed with TaKaRa Ex Taq HS polymerase (Takara Bio, Shiga, Japan) in total volumes of 50 µl using the manufacturer's recommended volumes of 10X Ex Taq buffer, dNTP mixture, and water.

Amplifications of COI were performed using the primers LCO-1490 and HCO-2198 (Folmer et al. 1994). PCR conditions included an initial denaturation step of 94°C (3 min), 32 cycles of 94°C (20 sec)/ 50°C (20 sec)/ 72°C (30 sec), and an extension step of 72°C (5 min). Amplifications of 28S rDNA were performed using the primers WF&LD2F and A335R (A. Zwick, pers. comm. 2009) and following the same PCR conditions. To minimize the effects of rDNA secondary structure, 3 µl of water was replaced with dimethylsulfoxide (DMSO) in all 28S rDNA reactions. Amplifications of EF-1α were performed using the primers M51.9tort (A. Zwick, pers. comm. 2009) and rcM4 (Cho et al. 1995). PCR conditions included an initial denaturation step of 94°C (4 min), 5 cycles of 94°C (30 sec)/ 52°C (30 sec)/ 72°C (1 min), 7 cycles of 94°C (30 sec)/ 51°C (1 min)/ 72°C (1 min), 36 cycles of 94°C (30 sec)/ 45°C (20 sec)/ 72°C (1 min, 30 sec), and a final extension step of 72°C (3 min). Amplicons of COI, 28S rDNA, and EF-1α were purified using a Qiaquick PCR Purification Kit (Qiagen, Valencia, Calif.) and eluted into 35 µl of EB buffer. Amplifications of CAD were performed using the primers 791F (Regier et al. 2008) and 1028R (Wahlberg and Wheat 2008). PCR conditions were identical to those used in amplifying EF-1α. Because CAD amplification resulted in multiple PCR products for many taxa, all PCR products were gel extracted using a QIAquick Gel Extraction Kit (Qiagen, Valencia, Calif.). The PCR product and 6 µl of loading dye were loaded into a single well on a 2.0% lowmelt agarose gel. After performing electrophoresis for approximately 3 hours at 75V, individual bands were excised using gel cutting pipette tips (BioExpress, Kaysville, Utah), dissolved in 500 µl of buffer QC, and eluted into 35 µl of EB buffer after following the manufacturer's recommended gel extraction protocol.

All purified PCR products were sequenced by the University of Chicago Cancer Research Center DNA Sequencing Facility using an Applied Biosystems 3730XL DNA sequencer (Applied Biosystems, Foster City, Calif.). The same primers used for PCR were also used for sequencing. Individual contigs were assembled and trimmed using Geneious Pro 5.4.6 (Drummond et al. 2012). All DNA sequences generated by this study were submitted to GenBank under accession numbers [to be submitted in manuscript] (Appendix II).

Data analysis

Individual gene regions were aligned using MAFFT ver. 6 (Katoh et al. 2002). The Q-INS-i algorithm was used for 28S rDNA sequences as it considers secondary structure of rRNA (Katoh and Toh 2008), while the G-INS-i algorithm was used for all other gene regions. Parameters for all gene regions used the “1PAM / k=2” scoring matrix, a gap opening penalty of 1.53, and gap offset value of 0.1. Single-read CAD sequences for six specimens that failed to assemble were aligned to the 5'-end or 3'-end of the initial CAD alignment and manually assembled into a single sequence.

With the exception of the six manually assembled CAD sequences, no gaps were present in the COI, EF-1α, or CAD alignments. Manual adjustments to the 28S rDNA alignment were performed in MacClade ver. 4.08 (Maddison and Maddison 2005) using the similarity method described in Simmons (2004) following Zurawski and Clegg (1987). A total of 26 ambiguously-aligned positions in three regions were excluded from the 28S rDNA alignment. Five gap characters were scored using modified complex indel coding (Simmons and Ochoterena 2000) from unambiguously-aligned regions and included in the parsimony analyses.

Several separate process partitions (Bull et al. 1993) were analyzed as a method of data exploration. Each of the four gene regions were analyzed as separate coalescent genes (Doyle 1995) and their gene trees compared for well-supported topological incongruencies that may be a sign of introgression, lineage sorting, or unrecognized paralogy (Doyle 1992). All genes were analyzed in a combined molecular matrix and a simultaneous parsimony analysis was performed using all molecular and morphological data. Individual and simultaneous-analysis data matrices are posted a supplemental online data.

Equally weighted parsimony tree searches were conducted for each data matrix using TNT ver. 1.1 (Goloboff et al. 2008). Five thousand random addition tree-bisection-reconnection (TBR) searches were performed with a maximum of 50 trees held per replicate, and the ratchet (Nixon 1999) set for 100 iterations with a 10% probability of upweighting a character and a 5% probability of downweighting a character. Parsimony jackknife (JK; Farris et al. 1996) analyses were conducted using TNT with the removal probability set to 0.37. One thousand JK replicates were performed with 100 random addition TBR searches and a maximum of 50 trees held per replicate. PartitionFinder ver. 1.0.1 (Lanfear et al. 2012) was used to estimate the best-fit partitioning scheme and nucleotide substitution model for likelihood analyses (Felsenstein 1973). A total of 28 substitution models were considered; invariant-site models were excluded because models containing the gamma distribution were evaluated (Yang 1993, 2006). The Akaike information criterion (AIC; Akaike 1974) was used to select the best model with linked branch lengths between subsets. Data blocks were partitioned into genes and codon position for each coding gene and all search schemes were considered (exhaustive search). A total of ten subsets (each codon position in each gene for the coding genes + 28S rDNA) were chosen for the combined molecular matrix, with Q-matrices alternately consisting of GTR, TVM, TIM, TrN, HKY, and F81. All models except F81 incorporated the gamma distribution. Three subsets (each codon position) were chosen for each individual gene, with the same set of models as in the combined matrix.

Table 115. Data matrix and tree statistics. CI = ensemble consistency index (Kluge and Farris, 1969) on the most parsimonious trees for the parsimony-informative characters. RI = ensemble retention index (Farris 1989). *Bootstrap statistics calculated from the combined molecular likelihood tree.

Matrix	# terminals	# characters analyzed	# of parsimony informative characters	% missing/ inapplicable	Most parsimonious tree length	# of most parsimonious trees	# of jackknife/ bootstrap clades ≥ 50%	Average jackknife/ bootstrap support (%)	CI	RI
28S rRNA	73	828	105	5.5	383	532	16/16	76/75	0.54	0.79
COI	82	658	236	0.2	1928	9	28/23	85/86	0.22	0.50
EF-1a	71	568	117	1.3	461	62300	17/18	77/77	0.43	0.66
CAD	60	638	207	3.1	812	480	26/28	82/81	0.49	0.72
Simultaneous*	82	2719	691	14.8	4095	14	41/45	86/85	0.31	0.57

Likelihood analyses of nucleotide characters were performed as tests for long-branch attraction (Felsenstein 1978; but see Siddall 1998) using GARLI ver. 2.0 (Zwickl 2006). Optimal likelihood trees were searched for using 1000 independent searches. The partitioning schemes and models suggested by PartitionFinder were specified along with different models and different subset rates in the GARLI configuration file. Likelihood bootstrap (BS; Felsenstein 1985) values were obtained using at least 1000 replicates and ten searches per replicate for each matrix.

Results

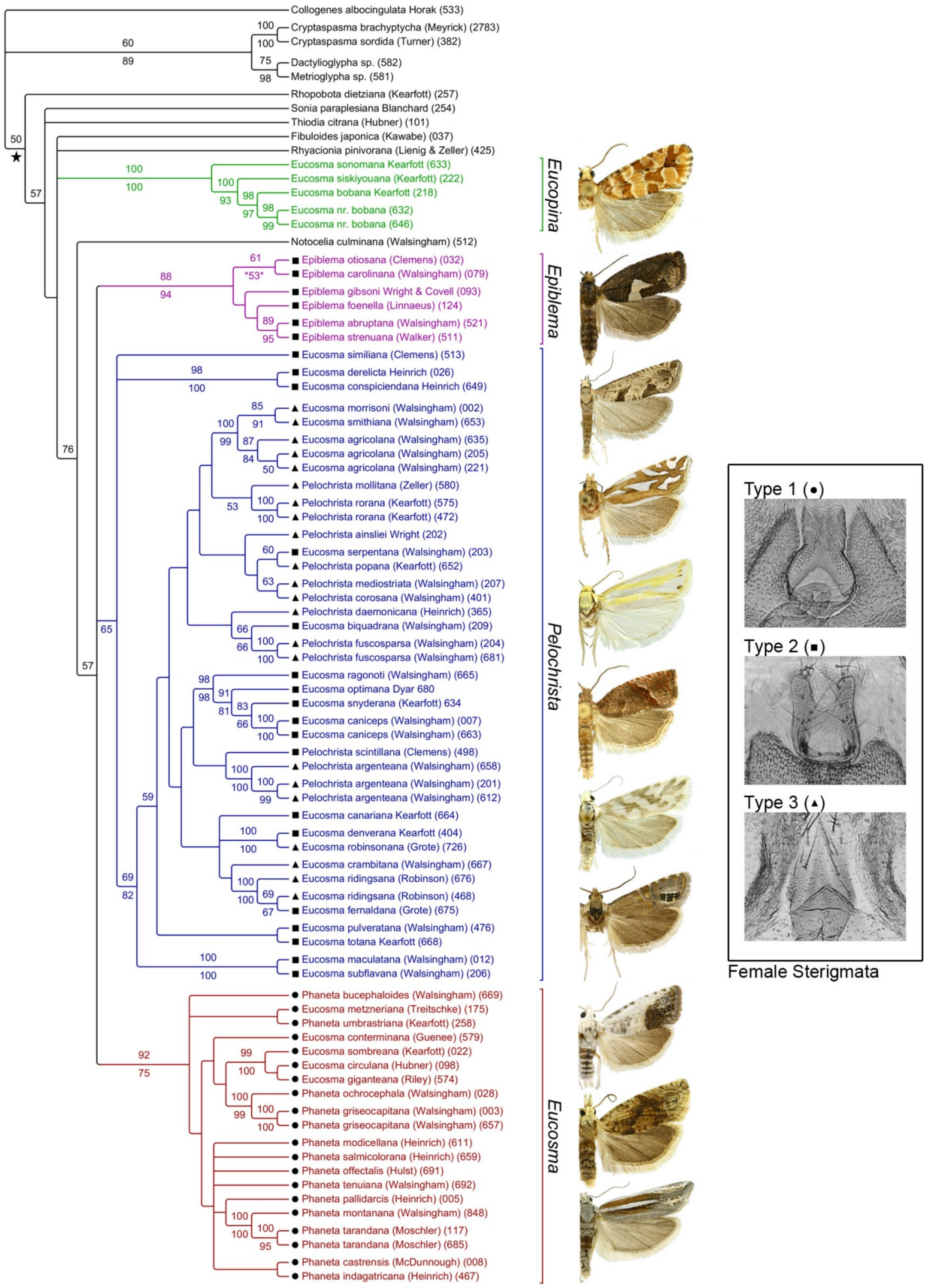
The simultaneous analysis parsimony strict consensus tree is presented in Fig. 90 with parsimony JK values ≥ 50% above each branch and likelihood BS values ≥ 50% below each branch from the combined molecular likelihood analysis. Equivalent trees for each individual gene and the combined molecular likelihood tree are presented in Appendix IV (Figs. 102–106).

All trees were created using TreeGraph 2 (Stöver and Müller 2010), and SumTrees from the DendroPy 3.11 package (Sukumaran and Holder 2010) was used to map support values. Support values were mapped onto the parsimony strict consensus tree instead of being presented on the majority-rule-consensus tree to avoid frequency-within-replicates and undersampling-within-replicates JK and BS artifacts (Davis et al. 1998; Simmons and

Freudenstein 2011). Unambiguously-supported clades refer to those with JK/BS values = 100%, highly-supported clades refer to those with JK/BS values ≥ 90%, well-supported clades refer to those with JK/BS values 89–70%, and weakly-supported clades refer to those with < 69% JK/BS values. Statistics for data matricies and corresponding trees are presented in Table 115.

Process partitions

No mutually well-supported incongruencies were resolved between the parsimony and likelihood trees for the individual gene data matrices or between the simultaneous analysis parsimony strict consensus tree (Fig. 90) and the combined molecular likelihood tree (Fig. 106). Minor incongruencies were resolved between the COI and nuclear gene trees, likely as a result of introgression, lineage sorting, or unrecognized parology (Doyle 1992; Maddison 1997) in closely related taxa. The COI gene tree conflicts with the three other genes in the arrangement of taxa in the *Eucosma agricolana* clade. While all gene trees resolve the clade with high support (≥ 88% JK/ ≥ 85% BS), *E. agricolana* (635) and *E. smithiana* (653) are resolved as sisters (85% JK/85% BS) in the COI tree, while *E. smithiana* + *E. morrisoni* are weakly supported as sister to the *E. agricolana* individuals in the three other gene trees (< 69% JK/BS). Similarly, *E. ridingsana* (TOR-DNA-468) and *E. fernaldana* are resolved as sister taxa in the COI gene tree (66% JK/85% JK), while the other *E. ridingsana* specimen (TOR-DNA-0676) is resolved as sister to *E. fernaldana* in the the EF1a gene tree (83% JK/ < 50% BS).



[Caption next page]

Figure 90. Simultaneous analysis parsimony strict consensus tree. Parsimony JK values are above each branch and likelihood BS are below each branch. The single clade that was contradicted by ≥ 50% BS support is indicated by *XX* with BS support for the contradictory clade. The base of the Eucosmini is indicated with a star. Ingroup taxa are labeled with generic names proposed in this revision and color-coded as follows: *Eucopina* green; *Epiblema* purple; *Pelochrista* blue; *Eucosma* red. Female sterigmata types are mapped onto the tree for the *Epiblema-Eucosma-Pelochrista* clade and indicated with the following symbols: type 1, circle; type 2, square; type 3, triangle. Moth photos represent typical wing patterns found in each ingroup clade.

Systematic implications

In agreement with other studies (Regier et al. 2009, 2012), Eucosmini are resolved as monophyletic, although weakly supported (50% JK/ < 50% BS). The single synapomorphy for the tribe is the stalked M_3-CuA_1 vein in the hindwing. Because Eucosmini are a large tribe consisting of 227 genera (Gilligan et al. 2012), taxon sampling was not sufficient to adequately test sister relationships between the several outgroup Eucosmini taxa and the ingroup clades. As such, the remainder of the results and discussion will focus only on the ingroup clades as resolved in the simultaneous analysis parsimony strict consensus tree (Fig. 90), which is preferred because it includes additional data (morphological and gap characters) not sampled in the likelihood analysis.

Eucosma, as currently defined, consists of at least three separate lineages. The first is an unambiguously supported (100% JK/100% BS) clade of Pinaceae-feeding *Eucosma*: *E. sonomana*, *E. siskiyouana*, *E. bobana*, and a likely undescribed species "*E.* nr. *bobana*." This group is well separated from all other *Eucosma*-containing clades and is distinguished by a medial ridge on the neck of the male valva. The remainder of the ingroup is contained in a single clade (57% JK/ < 50% BS) consisting of *Epiblema*, *Eucosma* + *Pelochrista*, and *Eucosma* + *Phaneta*, although the relationships between these three groups are unresolved. *Epiblema* is well supported as a single lineage (88% JK/94% BS) and is distinguished by the presence of a clasper on the male valva.

The second *Eucosma* lineage consists of a weakly-supported (< 50% JK/65% BS) clade of *Eucosma* + *Pelochrista*. Possible morphological synapomorphies include the presence of hook-tipped setae on the papillae anales, and one or more spiniform setae on the distal end of the neck or apex of the anal angle in the male valva. *Eucosma similana* and the clade of *E. derelicta* + *E. conspiciendana* are in a polytomy with a clade consisting of all other members of this lineage (69% JK/82% BS). Higher-level relationships within this larger subclade are weakly supported, with *Pelochrista* sorting into four separate subclades. Unambiguously or highly supported clades containing more than one species include: *E. morrisoni*, *E. smithiana*, and *E. agricolana* (100% JK/99% BS); *E. ragonoti*, *E. optimana*, *E. snyderana*, and *E. caniceps* (98% JK/98% BS); *E. denverana* and *E. robinsonana* (100% JK/100% BS); *E. ridingsana* and *E. fernaldana* (100% JK/100% BS); and *E. maculatana* and *E. subflavana* (100% JK/100% BS).

The third *Eucosma* lineage is a well-supported (92% JK/75% BS) clade of *Eucosma* + North American *Phaneta*. The morphological synapomorphy for this clade is the shape of the posterior margin of sternum 7, which is U-shaped and approximate to or fused laterally with the sterigma. Relationships within the clade are largely unresolved, with the exception of high support (100% JK/≥ 99% BS) for three subclades consisting of: *E. sombreana*, *E. circulana*, and *E. giganteana*; *P. ochrocephala* and *P. griseocapitana*; and *P. montanana* and *P. tarandana*.

Discussion

This is the first study to test the monophyly and infer intraspecific relationships in the largest of the olethreutine genera using a molecular phylogentic framework. *Eucosma* is divided into three separate lineages, including a monophyletic group of Pinaceae-feeders. *Epiblema* is inferred as monophyletic, and *Pelochrista* and North American *Phaneta* are divided among the other two *Eucosma* lineages. Changes in classification are required to redefine these genera as natural groups.

The Pinaceae-feeding species of *Eucosma* are a well-defined group consisting of 12 described species, all Nearctic in distribution. Whereas most *Eucosma* are borers in Asteraceae, larvae of this group feed on *Pinus*, *Picea*, *Abies*, and *Pseudotsuga* (Pinaceae) (Powell 1968). Two species, *E. gloriola* and *E. sonomana*, bore into shoots and stems and feed inside, while the remaining species bore into cones and feed on the seeds (Powell 1968). The single morphological synapomorphy for this group is a raised ridge on the medial surface of the neck of the male valva. Other characters that can be used to distinguish *Eucopina* include: male with strong overlap of the cucullus with the neck of the valva, and a row of short spiniform setae along the distal margin of the cucullus; female with long ovipositor, and flat, triangular, ventrally facing papillae anales. Wing maculation is also consistent, with most species having red or orange forewings similar those found in other Pinaceae-feeding olethreutine genera, such as *Rhyacionia* and *Retinia*. The Pinaceae-feeding *Eucosma* clade is resolved well outside of the main *Epiblema-Eucosma-Pelochrista-Phaneta* clade, contradicting the hypothesis that these species are derived from an Asteraceae-feeding lineage within *Eucosma* (Powell 1968, Powell and Opler 2009). For these reasons, we describe this group as a new genus, *Eucopina* (formal description below). Interestingly, other unpublished analyses of Eucosmini using molecular data consistently recover many of the olethreutine Pinaceae-feeding genera in the same clade, suggesting a single origin of Pinaceae-feeding within the family (J. Baixeras, pers. comm. 2012). There is evidence of this trend here, as *R. pinivorana* is well supported as sister to *Eucopina* in the combined likelihood analysis.

Epiblema has been previously described as being similar to *Eucosma*, but distinguished from the latter by a clasper on the distal margin of the basal excavation on the male valva (Heinrich 1923, Gilligan et al. 2008). Razowski (1989) stated that the absence of this structure in *Eucosma* could be the result of a reduction, suggesting that the two genera could be congeneric. In the present study, we find *Epiblema* to be a well-supported monophyletic group, resolved in the same polytomy as the two *Eucosma* + *Phaneta* / *Eucosma* + *Pelochrista* clades, and find no reason to modify the circumscription of *Epiblema* based on these results. In the few species where the clasper is reduced or absent, the female sterigma can be used to separate *Epiblema* from *Eucosma* (see below).

Pelochrista was traditionally defined as having a pulvinus on the basal excavation of the male valva (Obraztsov 1967, Razowski 1989). In the genera included in this study, the pulvinus *sensu* Obraztsov (1967) is variable, ranging in a continuum from absent to strongly developed. It serves better as a species-level character, as it is strongly developed in many of the *Eucosma* + *Pelochrista* species. In North America, a large spiniform seta ("spike") on the anal angle of the cucullus or neck of the valva was often used to distinguish *Pelochrista* from related genera such as *Eucosma* (Wright 2007). Although this character is also variable, large spiniform setae are only present in the *Eucosma* + *Pelochrista* clade and are absent from the *Eucosma* + *Phaneta* clade. In cases where spiniform setae are present on the male valva of taxa in the *Eucosma* + *Phaneta* clade, they are numerous and of varying size. We suggest that a single or small group of large spiniform setae on the male valva may serve as a synapomorphy for the *Eucosma* + *Pelochrista* clade, and the absence of these setae in many species may be the result of a secondary loss. The male costal fold, used by Obraztsov (1952) to separate North American *Phaneta* from *Eucosma*, is present in all of the *Eucosma* and *Pelochrista*, and absent (by definition) in all of the *Phaneta* included in this study. As such, the *Eucosma* + *Pelochrista* clade is supported by the presence of the costal fold; this character state conflicts with the *Eucosma* + *Phaneta* grouping.

Female characters, specifically the structure of the sterigma and its interaction with sternum VII, are more congruent than the male characters among the ingroup taxa in the simultaneous-analysis tree. Razowski (2003) hinted at the importance of these female characters when he stated that the North American *Phaneta* could be "characterized chiefly by a short sterigma terminating at the level of the posterior parts of subgenital sternite" (pg. 77). The sterigmata found in *Eucosma*, *Pelochrista*, and *Phaneta* can be divided into three types: 1) lamella postvaginalis rectangular, lamella antevaginalis ringlike, posterior margin of sternum VII deeply emarginated (usually to full length of sterigma) and approximate to or fused with lateral margins of lamella postvaginalis; 2) lamella postvaginalis well developed, variable, with posterior margin often invaginated medially, posterior margin of sternum VII diverging laterally from sterigma with ringlike lamella antevaginalis separating ostium from sternum VII; and 3) lamella postvaginalis well developed, variable, with posterior margin often invaginated medially, anterior margin of ostium fused with sternum VII and lamella antevaginalis absent. These three types are illustrated and mapped onto the simultaneous analysis tree in Fig. 90; type 1 is a synapomorphy for members of the *Eucosma* + *Phaneta* clade, while types 2 and 3 are found in all members of the *Eucosma* + *Pelochrista* clade (and other genera). Additional illustrations are provided in Fig. 93 (type 1 sterigma), Fig. 94 (entire female genitalia), and Fig. 97 (types 2 and 3 sterigmata). A possible synapomorphy for species in the *Eucosma* + *Pelochrista* clade is the presence of hook-tipped setae arising from the female papillae anales. These setae are presumed to be used to prepare the substrate for oviposition by the female. This type of seta is not present in any species in the *Eucosma* + *Phaneta* clade.

Although not included in this study, we are confident, based on the long, tapering, laterally sclerotized socii in the male and the partially sclerotized ventral invaginations in the postsegmental membrane in the female, that the type species of *Phaneta* (*P. pauperana*) is not congeneric with North American *Phaneta*. Gilligan and Wright (2012 [Chapter 8]) designated a neotype for *E. circulana*, the type species of *Eucosma*; this is the same species sampled for this study. Because the type of *Eucosma* shares the same female morphology (type 1) as North American *Phaneta*, and these taxa are resolved in the same well-supported (92% JK/75% BS) clade, we transfer all of these species into *Eucosma*, and redefine the genus using these characters (formal redescription below). We also transfer all *Pelochrista* species with similar female morphology into *Eucosma*.

The clade of *Eucosma* + *Pelochrista* includes many species that would be considered *Pelochrista* both in North America and Europe. All taxa in this clade are characterized by the female having a type 2 or 3 sterigma, as defined above. Sterigma type 2 is likely plesiomorphic within the Eucosmini and the two sterigma types do not segregate into separate lineages within *Pelochrista*. Other distinguishing characters include the presence of hook-tipped setae on the female papillae anales and spiniform setae on the male valva. Additional taxon sampling is needed to adequately resolve intrageneric relationships, as is evident by the varying levels of support for groups within the genus, and by the low support values (< 50% JK/65% BS) for the *Pelochrista* clade itself. Higher support (69% JK/82% BS) is found for the clade that contains all *Eucosma* + *Pelochrista* species except *Eucosma similiana*, *E. derelicta*, and *E. conspiciendana*; however, these three species do not differ morphologically from the rest of the genus. It is likely that future studies will allow for division of *Pelochrista* into separate genera, but in the interim we transfer all *Eucosma* with type 2 or 3 female genitalia into *Pelochrista* and redescribe the genus using these characters (formal redescription below).

A complete list of the redefined *Eucosma*, *Pelochrista*, and *Phaneta* is presented in Appendix V. All new combinations, synonymies, and corrections to the current world catalogue (Gilligan et al. 2012) are noted. The changes presented here affect primarily North American taxa, with most Palearctic taxa retaining their placement according to Razowski (1999, 2003), with the exception of *E. guentheri* and *E. lugubrana*, which are transferred to *Pelochrista* based on female genitalia. When possible, female specimens or illustrations were examined to determine generic placement. If a female was unavailable for a Nearctic species, it is placed if there was compelling evidence for assignment using other characters (usually male genitalia). If a female was unavailable for a Palearctic species, the placement by Razowski (1999, 2003) is retained. All North American *Phaneta* are placed in *Eucosma* with the following exceptions: *P. cinereolineana* is placed in *Pelochrista* because of the large spiniform setae on the male valva, and the female is unknown; *P. delphinoides* and *P. delphinus* are tentatively placed in *Eucosma* under “Eucosmini unplaced” due to their unusual male and female genitalia. *Eucosma gomonana* was also placed under “Eucosmini unplaced” because its genitalia does not match the typical *Eucosma*. All taxa distributed outside of the Holarctic were transferred to “Eucosmini unplaced,” along with species for which a female could not be examined, and those species listed as “unplaced” in Razowski (1999). *Eucosma liturana* was transferred to *Zeiraphera* based on examination of the genitalia, and *Eucosma fulminana* was moved to *Sonia* based on examination of the genitalia and fused $R_4 + R_5$ veins in the forewing. In the appendix, species names in BOLD are used to designate new combinations. A total of 704 names (including synonyms) are listed, including approximately 320 new combinations.

Generic descriptions and redescriptions

EUCOSMA Hübner

(Figs. 91–93, 94.a)

Eucosma Hübner, 1823, Zutr.Samml.exot.Schmett. 2: 28. Type species: *Tortrix circulana* Hübner, 1823.

Affa Walker, 1863, List Specimens lepid. Insects Colln. Br. Mus. 27: 202. Type species: *Affa bipunctella* Walker, 1863.

Ascelodes Fletcher, 1929, Mem. Dep. Agric. India (Ent.) 11: 25; nomen nudum.

Calosetia Stainton, 1859, Man. Br. Butterflies Moths 2: 271. Type species: *Tortrix nigromaculana* Haworth, [1811].

Catoptria Guenée, 1845, Annls Soc. ent. Fr (2) 3: 187. Type species: *Tortrix cana* Haworth, [1811]; preoccupied by *Catoptria* Hübner [1825], Pyralidae. [corrected from Gilligan et al. 2012]

Exentera Grote, 1877, Can. Ent. 9: 227. Type species: *Exentera apriliana* Grote, 1877.

Exenterella Grote, 1883, Can. Ent. 15: 23; unnecessary replacement name for *Exentera*.

Ioplocama Clemens, 1860, Proc. Acad. Nat. Sci.Philad. 12: 360. Type species: *Ioplocama formosana* Clemens, 1860; **new synonymy**.

Joplocama Walker, 1864, List Specimens lepid. Insects Colln. Br. Mus. 30: 994; mispelling of *Ioplocama*; **new synonymy**.

Palpocrinia Kennel, 1919, Mitt. mnch. ent. Ges. 8: 66. Type species: *Palpocrinia ottoniana* Kennel, 1919.

Diagnosis. *Eucosma* can be separated from *Epiblema*, *Eucopina*, *Pelochrista*, and *Phaneta* by a combination of the following female character states: rectangular lamella postvaginalis, ringlike lamella antevaginalis, and posterior margin of sternum VII deeply emarginated and approximate to or fused with the lateral margins of the lamella postvaginalis. In addition, most *Eucosma* females have two signa of approximately equal size in the corpus bursae and lack hook-shaped setae arising from the papillae anales. Male character states are less diagnostic and cannot be used to separate *Eucosma* from *Pelochrista*, although most *Eucosma* lack the large spiniform seta that is present on the anal angle of the cucullus in many

Pelochrista species. *Eucosma* males also lack the clasper found on the distal margin of the basal excavation found in most *Epiblema* species.

Wings (Fig. 91). Forewing with R_4 and R_5 separate, M_2 and M_3 separate, chorda weak; males with (occasionally) or without (usually) a forewing costal fold. Forewing maculation is variable, although many species exhibit a well developed ocellus. Hindwing with Rs and M_1 approximate, M_2 and M_3 approximate, M_3 and Cu_1 stalked or united.

Male genitalia (Fig. 92). Uncus weakly to moderately developed, rounded apically or with shallow medial indentation, often well differentiated from dorsolateral shoulders of tegumen; socii fingerlike, moderately elongate and setose; phallus stout, often tapering distally, caulis short; cornuti present in vesica as a group of long deciduous spines; valva with costal margin moderately concave to nearly straight, neck well-defined, ventral margin broadly to deeply emarginate, basal excavation sometimes with weakly developed pulvinus; saccular corner angle usually well-defined, obtuse to slightly acute; cucullus densely setose, apex rounded and weakly to moderately produced, distal margin weakly to moderately convex, anal angle well-developed, occasionally with one or more rows of spiniform setae.

Female genitalia (Figs. 93, 96.a). Papillae anales flat, laterally facing, moderately to densely setose, without ventral extensions; lamella postvaginalis usually rectangular; lamella antevaginalis ringlike; posterior margin of sternum VII deeply emarginated (usually to full length of sterigma) and approximate to or fused with lateral margins of lamella postvaginalis; ductus bursae with sclerotized ring approximate to juncture with ductus seminalis; corpus bursae with two well developed signa, usually of approximately equal size.

Biology. Larvae feed almost exclusively on Asteraceae. Feeding behaviors include webbing of terminal leaves (particularly in early instars) and boring into seeds, flower heads, stems and roots. Most species appear to complete one annual generation, with adults present in mid-April through mid-October.

Distribution. Approximately 230 species are described, all from the Holarctic.

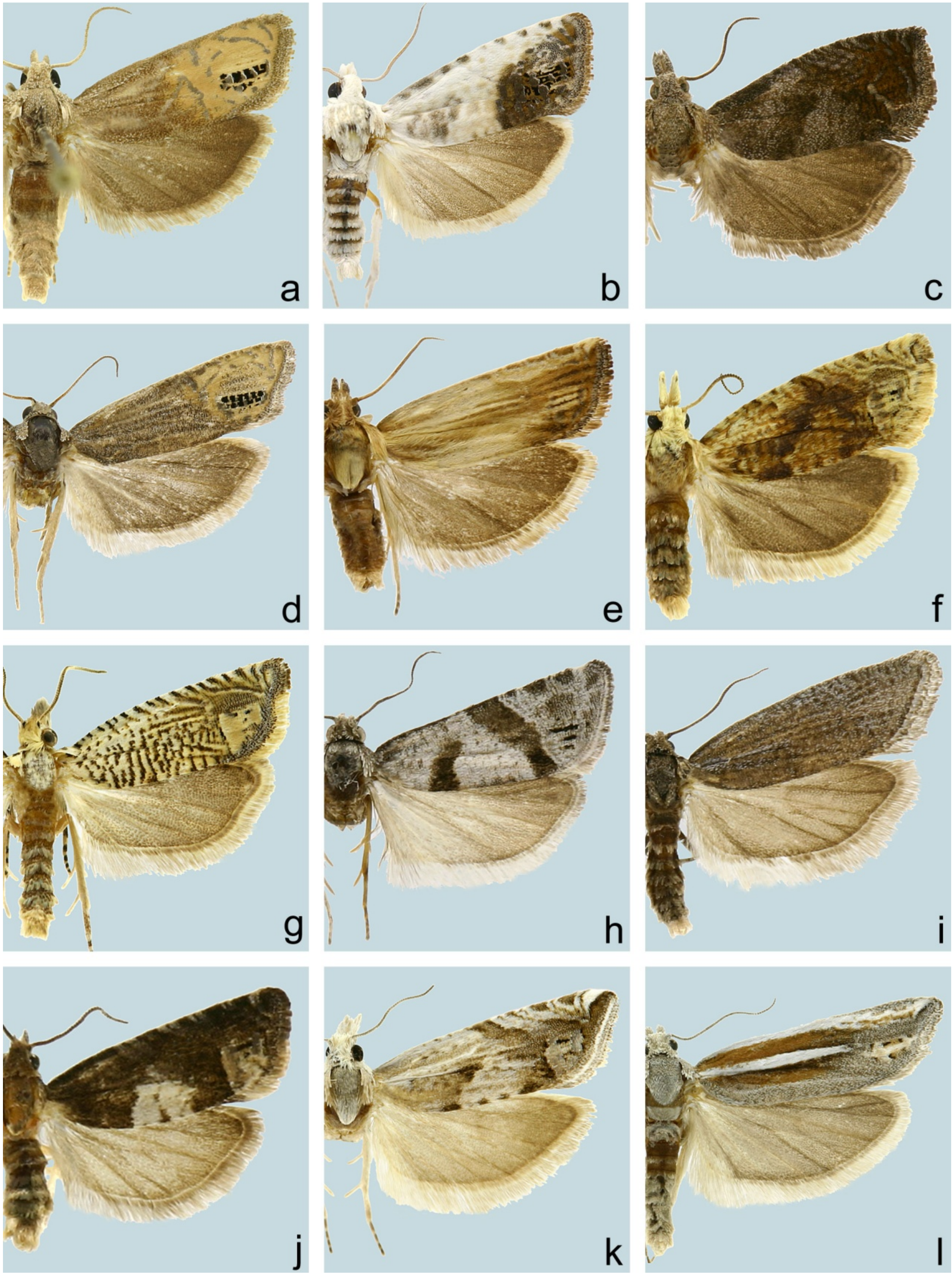


Figure 91. Examples of *Eucosma* adults; a: *E. circulana*; b: *E. giganteana*; c: *E. sombreana*; d: *E. autumnana*; e: *E. radiatana*; f: *E. ochrocephala*; g: *E. argutipunctana*; h: *E. convergana*; i: *E. canusana*; j: *E. parmatana*; k: *E. influana*; l: *E. argenticostana*.

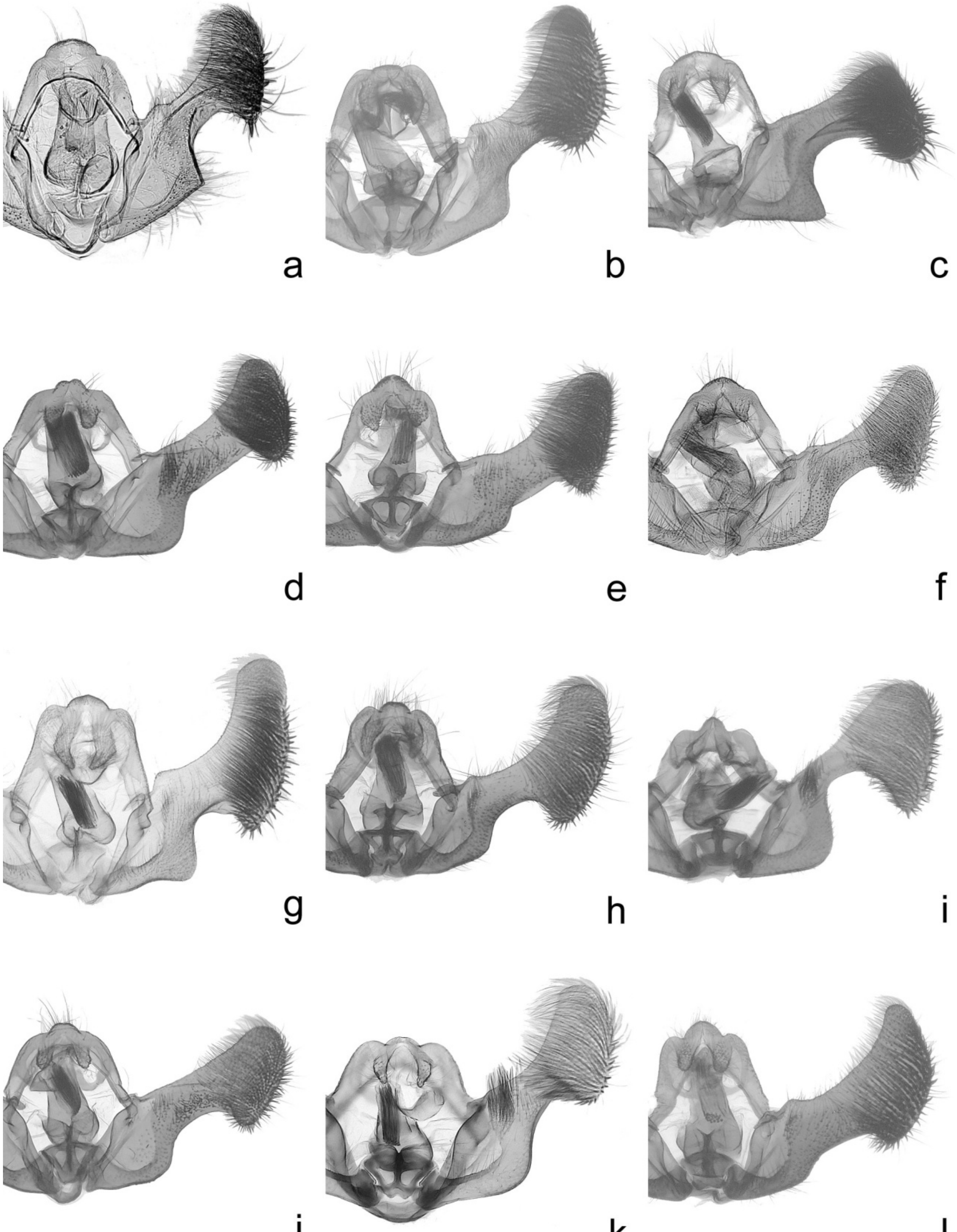


Figure 92. Examples of *Eucosma* male genitalia; a: *E. circulana*; b: *E. giganteana*; c: *E. sombreana*; d: *E. radiatana*; e: *E. convergana*; f: *E. montanana*; g: *E. rusticana*; h: *E. ochrocephala*; i: *E. autumnana*; j: *E. awemeana*; k: *E. canusana*; l: *E. argutipunctana*.

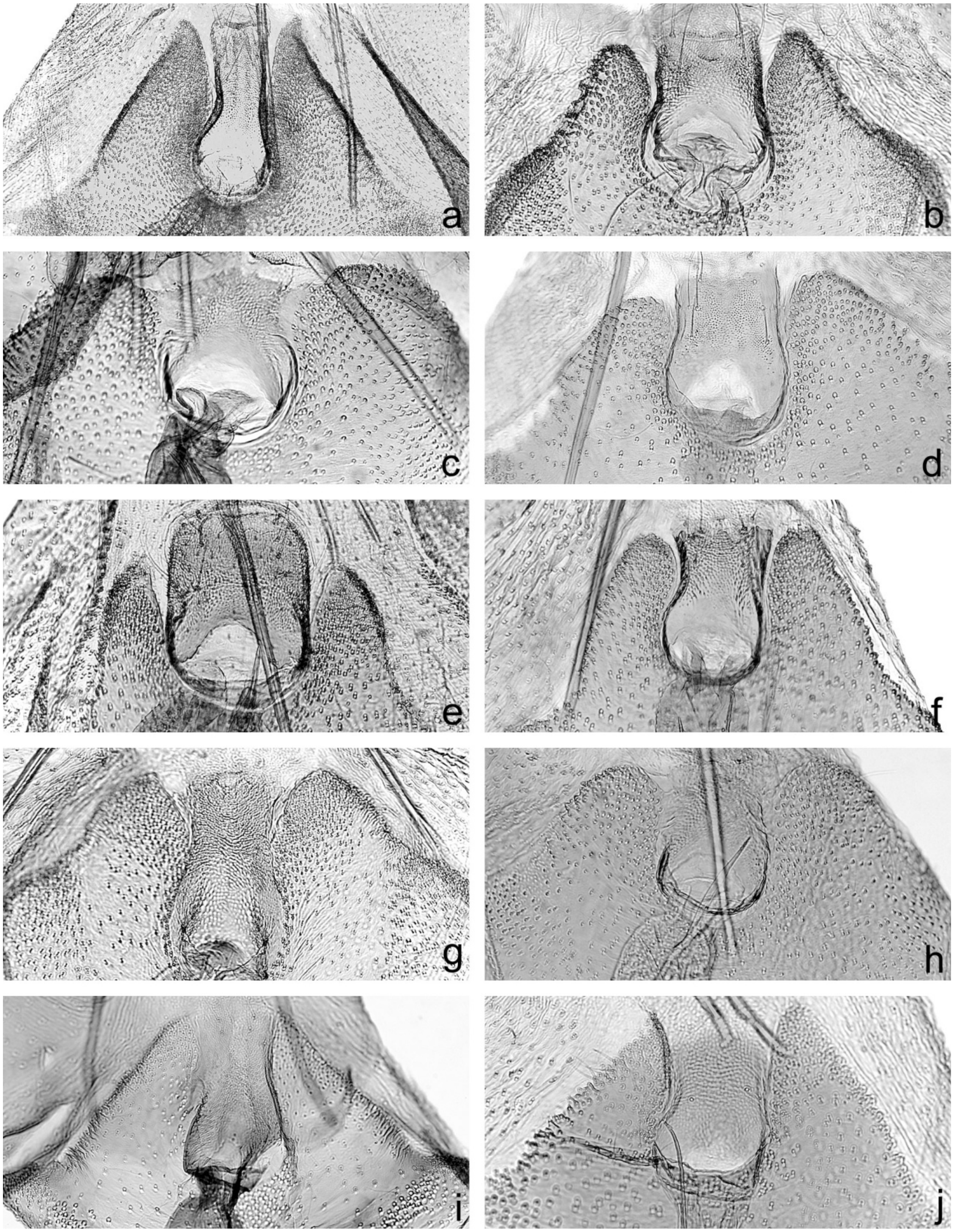


Figure 93. Examples of *Eucosma* sterigmata; a: *E. giganteana*; b: *E. sombreana*; c: *E. radiatana*; d: *E. montanana*; e: *E. rusticana*; f: *E. ochrocephala*; g: *E. autumnana*; h: *E. awemeana*; i: *E. canusana*; j: *E. argutipunctana*.

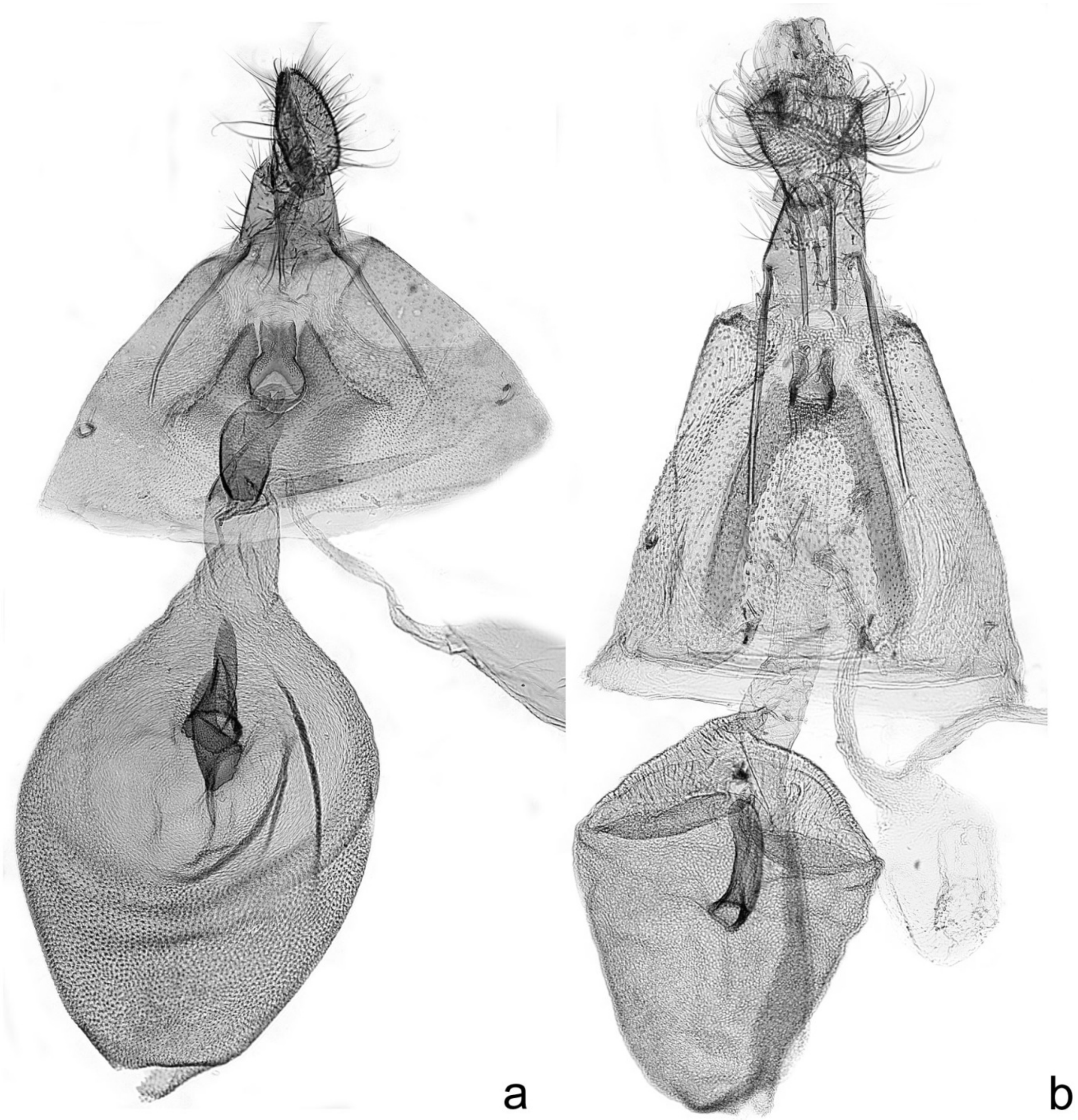


Figure 94. Comparison of *Eucosma* and *Pelochrista* females; a: *E. circulana*; b: *P. scintillana*.

PELOCHRISTA Lederer

(Figs. 94.b, 95–97)

Pelochrista Lederer, 1859, Wien. ent. Monatschr. 3: 331. Type species: *Paedisca mancipiana* Mann, 1855.

Callimosema Clemens, 1865, Proc. ent. Soc. Philad.5 141. Type species: *Callimosema scintillana* Clemens, 1865.

Eucosmoides Obraztsov, 1946, Z. Wien. ent. Ges. 30: 38. Type species: *Paedisca decolorana* Freyer, 1842.

Pseudeucosma Obraztsov, 1946, Z. Wien. ent. Ges. 30: 37. Type species: *Tortrix caecimaculana* Hübner, [1776-1799].

Pygolopha Lederer, 1859, Wien. ent. Monatschr. 3: 123. Type species: *Pygolopha trinacriana* Lederer, 1859; **new synonymy**.

Diagnosis. *Pelochrista* can be separated from *Eucosma* by the structure of the female sterigma, which is divided into two forms: posterior margin of sternum VII diverging laterally from sterigma with ringlike lamella antevaginalis separating ostium from sternum VII or anterior margin of ostium fused with sternum VII and lamella antevaginalis absent. In addition, some *Pelochrista* females have large ventrally facing papillae anales with ventral extensions and many species have hook-tipped setae arising from the papillae anales. Males of the two genera cannot be consistently separated, although many *Pelochrista* have a large spiniform seta on the anal angle of the cucullus or on the neck of the valva that is lacking in *Eucosma*. Details for separating *Pelochrista* from *Epiblema*, *Eucopina*, and *Phaneta* are found under those generic descriptions.

Wings (Fig. 95). Forewing with R_4 and R_5 separate, M_2 and M_3 separate, chorda weak; males with a forewing costal fold. Maculation variable, as in *Eucosma*. Hindwing with Rs and M_1 approximate, M_2 and M_3 approximate, M_3 and Cu_1 usually stalked.

Male genitalia (Fig. 96). Uncus weakly to moderately developed, rounded apically or with shallow medial indentation, often well differentiated from dorsolateral shoulders of tegumen; socii fingerlike, moderately elongate and setose; phallus stout to elongate, tapering distally, caulis often elongate; cornuti present in vesica as a group of long deciduous spines; valva with costal margin moderately concave to nearly straight, neck usually well-defined, ventral margin straight to deeply emarginate, basal excavation with weakly to strongly developed pulvinus; saccular corner angle variable, nearly straight to strongly acute; cucullus densely setose, apex usually rounded and weakly to strongly produced, distal margin weakly to strongly convex, occasionally with one or more rows of large spiniform setae, anal angle variable, if strongly developed, often with one or more larger spiniform setae at vertex.

Female genitalia (Figs. 94.b, 97). Papillae anales moderately to densely setose, flat and laterally facing or ventrally facing with ventral extensions, often with long hook-tipped setae along lateral margins; lamella postvaginalis well developed, variable, with posterior margin often invaginated medially; posterior margin of sternum VII diverging laterally from sterigma with ringlike lamella antevaginalis separating ostium from sternum VII or anterior margin of ostium fused with sternum VII and lamella antevaginalis absent; ductus bursae with sclerotized ring approximate to juncture with ductus seminalis; corpus bursae with one or two signa or signa absent; when two signa, usually of different sizes, occasionally with sclerotization of the burase around the smaller signum.

Biology. Larval biology is identical to that of *Eucosma*.

Distribution. Approximately 200 species are known, all from the Holarctic.

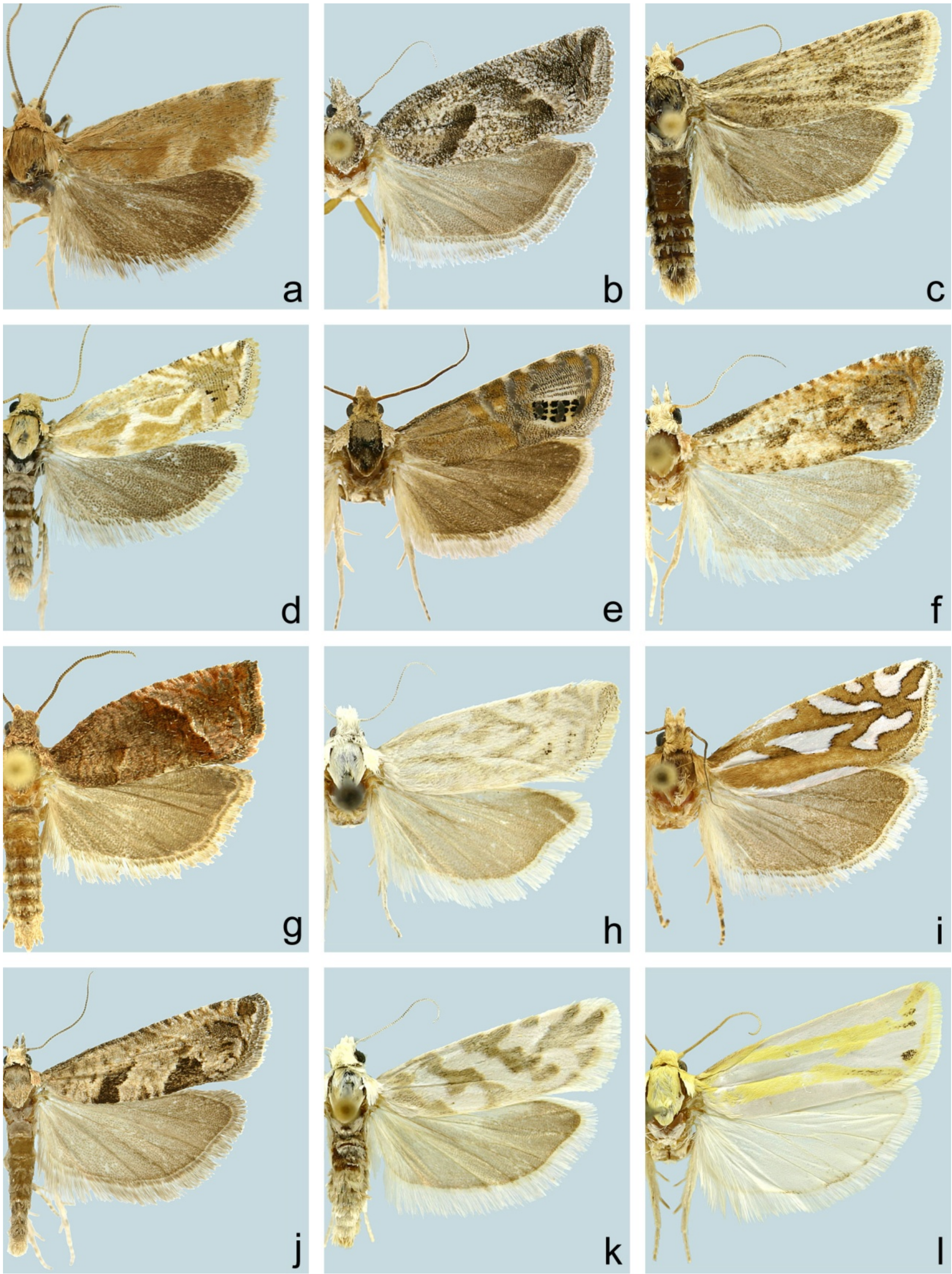


Figure 95. Examples of *Pelochrista* adults; a: *P. mancipiana*; b: *P. corosana*; c: *P. fuscosparsa*; d: *P. gilligani*; e: *P. scintillana*; f: *P. kingi*; g: *P. derelicta*; h: *P. agricolana*; i: *P. ridingsana*; j: *P. pulveratana*; k: *P. ragonoti*; l: *P. canariana*.

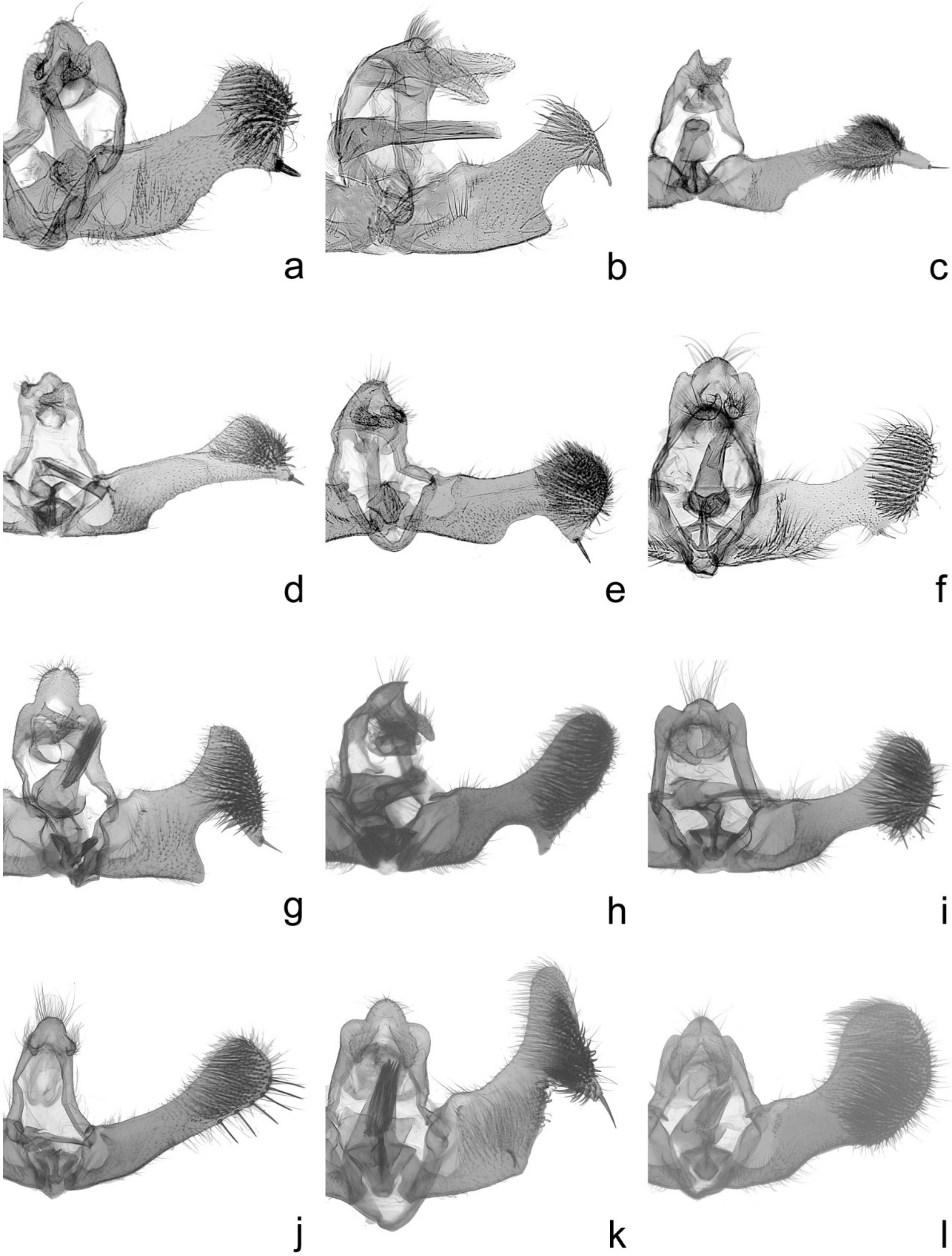


Figure 96. Examples of *Pelochrista* male genitalia; a: *P. scintillana*; b: *P. rorana*; c: *P. argenteana*; d: *P. corosana*; e: *P. milleri*; f: *P. pallidipalpana*; g: *P. agricolana*; h: *P. matutina*; i: *P. ridingsana*; j: *P. robinsonana*; k: *P. derelicta*; l: *P. similiana*.

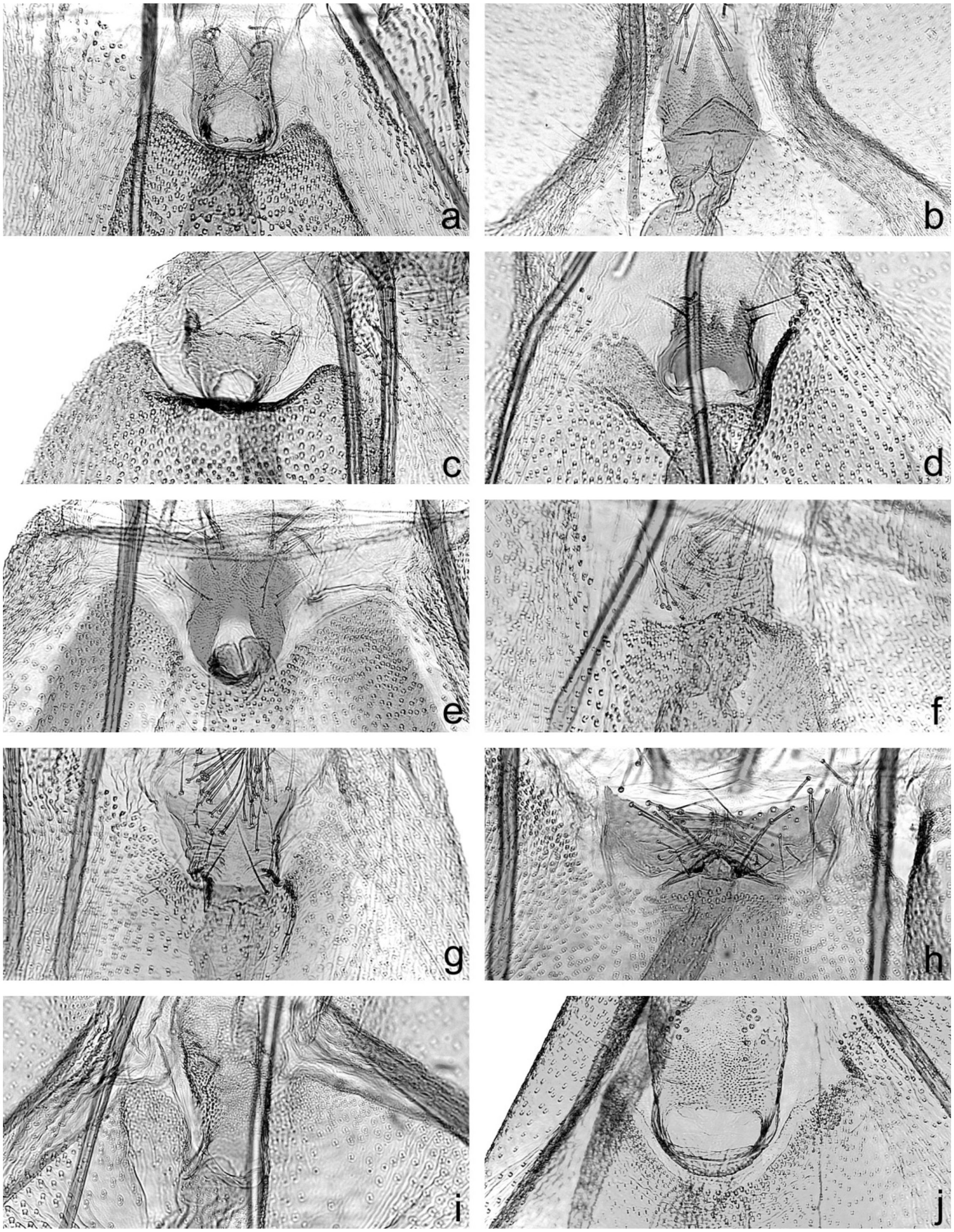


Figure 97. Examples of *Pelochrista* sterigmata; a: *P. scintillana*; b: *P. rorana*; c: *P. argenteana*; d: *P. pallidipalpana*; e: *P. milleri*; f: *P. agricolana*; g: *P. ridingsana*; h: *P. robinsonana*; i: *P. derelicta*; j: *P. similiana*.

***EUCOPINA*, new genus**

(Figs. 98.a–h, 99)

Type species: *Eucosma bobana* Kearfott, 1907

Diagnosis. *Eucopina* can be distinguished from other genera treated here by the long ovipositor and flat, triangular, ventrally facing papillae anales in the female and the male valva, with a ridge on the medial surface, strong overlap of the cucullus with the neck, and a row of short spiniform setae along the distal margin of the cucullus. Forewing pattern is similar to that of other Pinaceae-feeding olethreutines in the genera *Rhyacionia* and *Retinia*; however males of these genera lack the forewing costal fold found in *Eucopina* and are easily separable using genitalic characters (illustrations in Gilligan et al. 2008).

Wings (Figs. 99.a–h). Venation as in *Pelochrista*; males with a forewing costal fold. Wing maculation is consistent, with most species having an orange or red forewing with silver, gray, white, or tan markings.

Male genitalia (Figs. 99.a–c). Uncus weakly to moderately developed, rounded apically or with shallow medial indentation, often well differentiated from dorsolateral shoulders of tegumen; socii fingerlike, moderately elongate and setose; phallus moderately stout, tapering distally; cornuti present in vesica as a group of long deciduous spines; valva with costal margin weakly concave, neck well-defined, medial surface of valva with ridge midway between distal margin of basal excavation and neck; saccular corner angle usually well-defined, obtuse; cucullus densely setose, apex rounded and strongly produced, distal margin weakly convex with series of stout spiniform setae extending from anal angle to ca. ¾ distance to apex, anal angle well-developed with ventral ¼ to ½ of cucullus overlapping neck.

Female genitalia (Figs. 99.d–f). Papillae anales small, flat, triangular, ventrally facing; apophyses anteriores and apophyses posteriores long; tergum VIII long, semi-rectangular; sterigma consisting of well-developed lamella postvaginalis and ringlike lamella antevaginalis or platelike sterigma surrounding ostium; sternum VII strongly sclerotized along margins, weakly sclerotized medially; ductus bursae elongate, often with sclerotized ring posterior to juncture with ductus seminalis; corpus bursae with single thornlike signum.

Biology. Larvae feed on *Abies*, *Picea*, *Pinus*, and *Pseudotsuga* (Pinaceae). Larvae of *E. gloriola* and *E. sonomana* bore into shoots and stems, while those of the other species bore into cones and feed on seeds (Powell 1968).

Distribution. Twelve species are described, all from the Nearctic.

Etymology. The generic name is derived from “Euco + pina,” referring to *Eucosma* as the former genus and Pinaceae as the larval host.

Remarks. Included in the analysis are two specimens of an *Eucopina* species “near *bobana*” collected in Wyoming. These individuals were not collected near any pinyon pine, the larval host of *E. bobana*, and they are also much larger than most *E. bobana* in collections. They could represent a new species, or simply a population of *E. bobana* that has expanded its host range onto other species of pine.

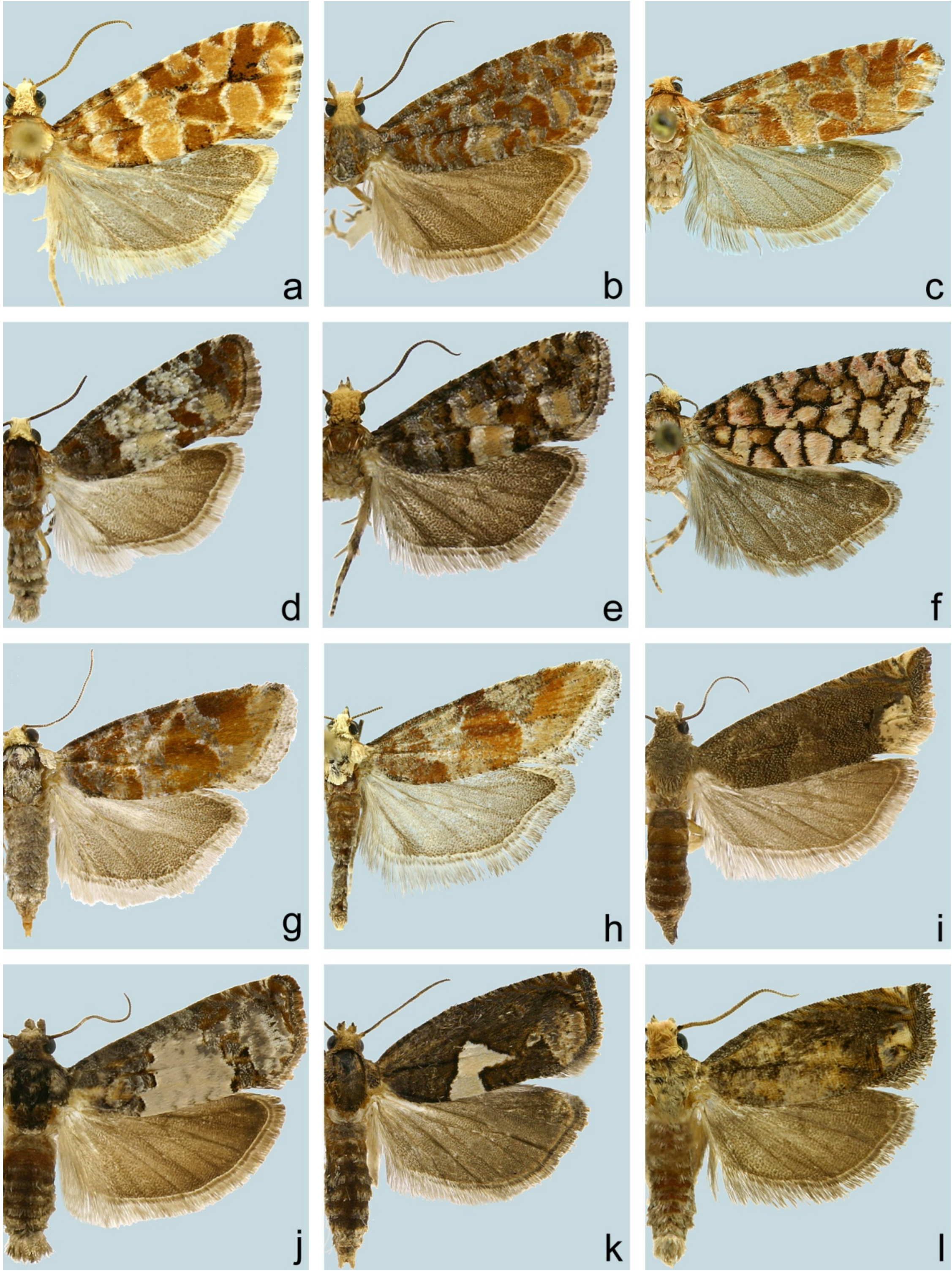


Figure 98. Examples of *Eucopina* (a–h) and *Epiblema* (i–l) adults; a: *Eucopina bobana*; b: *E. cocana*; c: *E. ponderosa*; d: *E. monitorana*; e: *E. tocullionana*; f: *E. siskiyouana*; g: *E. gloriola*; h: *E. sonomana*; i: *Epiblema strenuana*; j: *E. carolinana*; k: *E. otiosana*; l: *E. abruptana*.

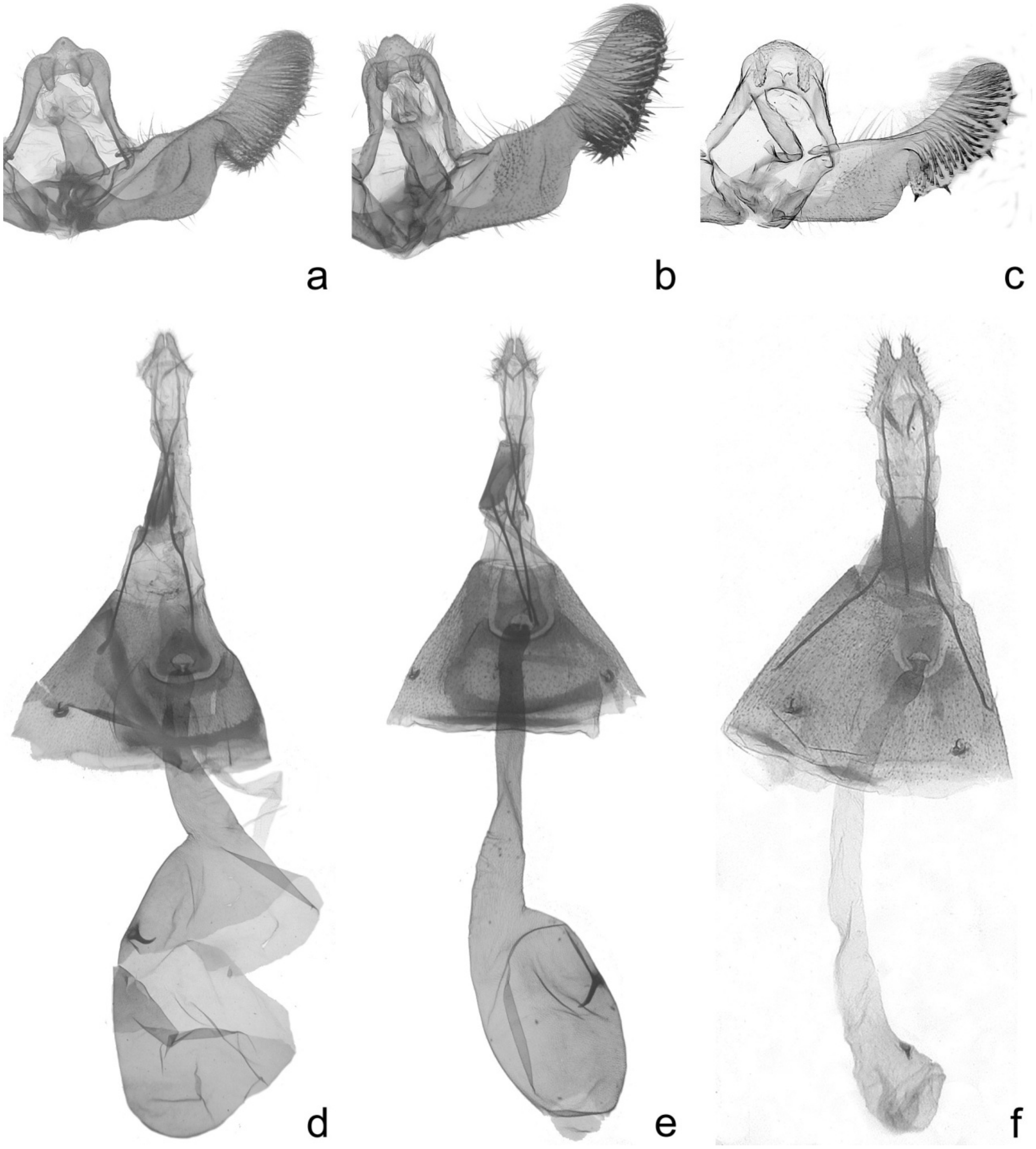


Figure 99. Examples of *Eucopina* male (a–c) and female (d–f) genitalia; a, d: *E. cocana*; b, e: *E. tocullionana*; c, f: *E. gloriola*.

PHANETA Stephens

(Fig. 100)

Phaneta Stephens, 1852, List Specimens Br. Animals Colln. Br. Mus. 10: 32. Type species: *Cochylis pauperana* Duponchel, 1842.

Astenodes Kuznetzov, 1966, Trudy Zool. Inst. Leningrad 37: 196. Type species: *Astenodes bimaculata* Kuznetzov, 1966.

Diagnosis. *Phaneta* can be distinguished from the other genera treated here by the long, tapering, laterally sclerotized socii in the male and the partially sclerotized ventral invaginations in the postsegmental membrane in the female.

Wings (Fig. 100.a). Venation same as in *Eucosma*; males without a forewing costal fold. Forewings are grayish brown with dark brown fasciate markings.

Male genitalia (Fig. 100.b). Uncus undeveloped; socii long, tapering, laterally sclerotized, setose on medial surface; phallus short, tapering distally; valva with costal margin weakly concave, neck reduced and poorly defined, ventral margin weakly emarginate; saccular corner angle obtuse; cucullus with apex tapering, rounded, strongly produced, distal margin weakly concave, anal angle rounded, with several rows of small stout seta along the margin; long stout setae extending from anal angle to mid-cucullus, becoming small and thin near apex.

Female genitalia (Fig. 100.c). Papillae anales flat, ventrally facing, moderately setose, without ventral extensions; apophyses posteriors fused basally; postsegmental membrane with partially sclerotized ventral invaginations; lamella postvaginalis semi-rectangular, divided into two convex lobes posteriorly; lamella antevaginalis ringlike; posterior margin of sternum VII deeply emarginated to ca. ¾ length of sterigma, closely approximate to lateral margins of lamella postvaginalis; ductus bursae with sclerotized ring at juncture with ductus seminalis; corpus bursae with two well developed signa, one ca. ¾ the size of the other.

Biology. Larvae feed in webbed buds, flowers, and fruit of *Rosa* (Rosaceae). Pupation occurs in webbed leaves on the ground. One generation per year, with adults present in April and May (Razowski 2003).

Distribution. Palearctic; two species. *Phaneta pauperana* occurs from western Europe to Asia Minor (Razowski 2003). *P. bimaculata* is recorded from Europe, Russia, China, and Japan (Zhang and Li 2005).

Remarks. As revised, *Phaneta* contains two species: *P. pauperana* and *P. bimaculata*. All North American species are transferred to *Eucosma* with the exception of *P. delphinoides* and *P. delphinis*, which are tentatively transferred to *Eucosma* under "Eucosmini unplaced" based on their aberrant male and female genitalia, and *P. cinereolineana*, which is placed in *Pelochrista* because of the large spiniform setae on the male valva (the female is unknown). Gilligan et al. (2012) lists four additional *Phaneta* occurring outside North America: *P. autochthones* (Walsingham), *P. cetratana* (Kennel), *P. pylonitis* (Meyrick), and *P. sardoensis* (Rebel). Razowski had previously (1999, 2003) moved *P. cetratana* and *P. sardoensis* into *Eucosma*, and this arrangement is preserved here. Clarke (1958) illustrated a male of *P. pylonitis*, and this species is placed in *Eucosma* pending the examination of a female specimen. *Phaneta autochthones* was described from the U. S. Virgin Islands, and it is unplaced under *Eucosma* pending examination of a female specimen.

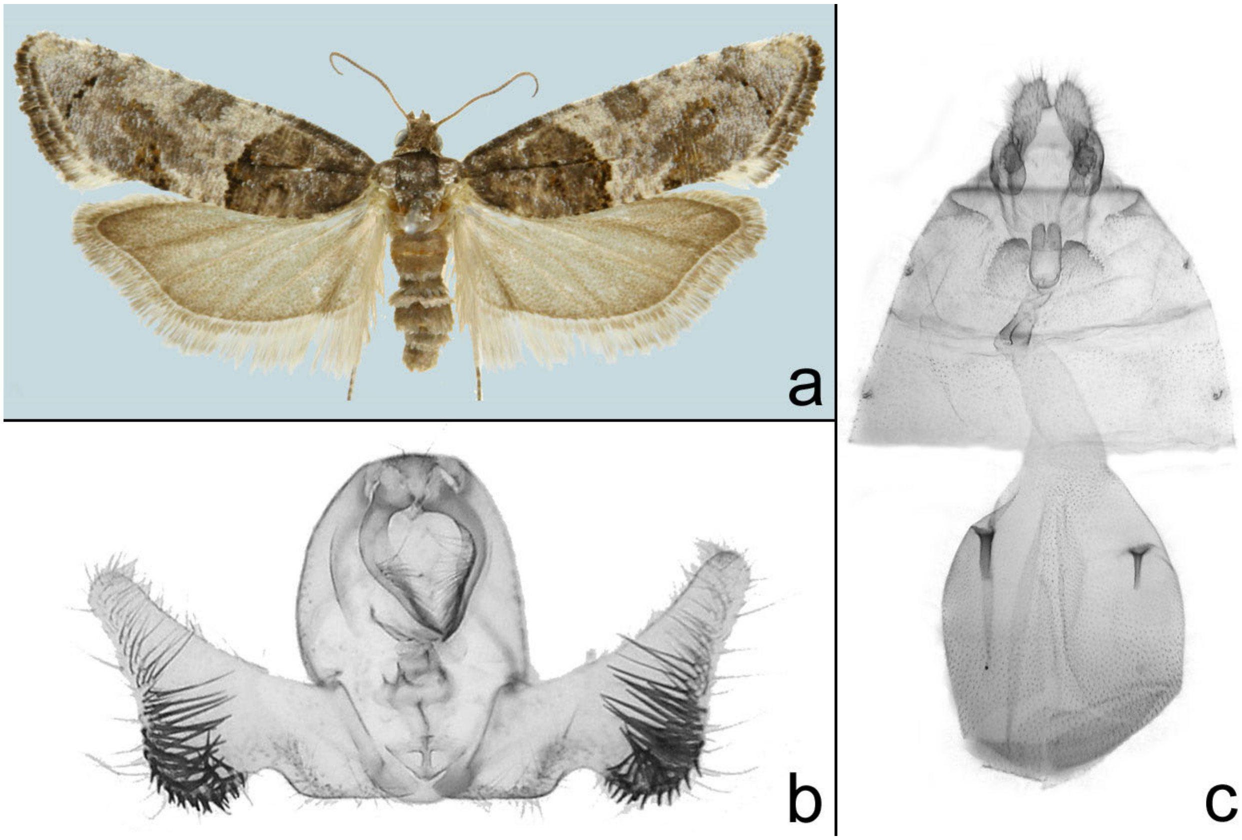


Figure 100. *Phaneta pauperana*; a: adult male; b: male genitalia; c: female genitalia.

EPIBLEMA Hübner

(Figs. 98.i–l, 101)

Epiblema Hübner, [1825] 1816, Verz. bekannter Schmett. 375. Type species: *Phalaena* (*Tinea*) *foenella* Linnaeus,1758.

Cacochroea Lederer, 1859, Wien. ent. Monatschr. 3: 331. Type species: *Paedisca grandaevana* Lienig & Zeller, 1846.

Epiblemma Hübner, [1825] 1816, Verz. bekannter Schmett. 63; misspelling of *Epiblema*.

Euryptychia Clemens, 1865, Proc. ent. Soc. Philad. 5: 140. Type species: *Euryptychia saligneana* Clemens, 1865.

Eurytychia Heinrich, 1923, Bull. U.S. natn. Mus. 123: 137; misspelling of *Euryptychia*.

Monosphragis Clemens, 1860, Proc. Acad. Nat. Sci.Philad. 12: 354. Type species: *Monosphragis otiosana* Clemens, 1860.

Diagnosis. *Epiblema* can be separated from the other genera treated here by the clasper on the distal margin of the basal excavation of the male valva. In the few species where this structure is reduced or absent, the male genitalia most closely resemble those found in *Eucosma*. The female sterigma is similar to that found in *Pelochrista* and can be used to separate female *Epiblema* from *Eucosma*.

Wings (Figs. 98.i–l). Venation as in *Eucosma*; males with a forewing costal fold. Many species have a distinctive interfascial spot on the dorsal margin of the forewing between the subbasal and median fasciae.

Male genitalia (Figs. 101.b–c). Uncus weakly to moderately developed, rounded apically or with shallow medial indentation, often well differentiated from dorsolateral shoulders of tegumen; socii fingerlike, moderately elongate and setose; phallus stout, tapering distally, caulis short; cornuti present in vesica as a group of long deciduous spines; valva with costal margin weakly to moderately concave, neck well-defined, ventral margin weakly emarginated to nearly straight, basal excavation often with weakly developed pulvinus and prominent tetrahedron-shaped clasper on distal margin; saccular corner angle weakly defined or absent, obtuse when present; cucullus densely setose, apex rounded and moderately to strongly produced, distal margin weakly to moderately convex, occasionally with one or more rows of small spiniform setae, anal angle well-developed, large spiniform setae at vertex absent.

Female genitalia (Fig. 101.a). Papillae anales moderately to densely setose, flat and laterally facing or ventrally facing, ventral extensions absent; lamella postvaginalis well developed, variable, with posterior margin often invaginated medially; posterior margin of sternum VII diverging laterally from sterigma with ringlike lamella antevaginalis separating ostium from sternum VII; ductus bursae often with sclerotized ring approximate to juncture with ductus seminalis; corpus bursae with one or two signa; when two signa, usually of different sizes.

Biology. Larvae are stem- or root-borers in Asteraceae. Larvae overwinter and pupate in the spring, often in an elongate gall. Adults fly in late April through September.

Distribution. Holarctic, approximately 90 species are described.

Remarks. The synapomorphy for *Epiblema* is the clasper on the male valva. We define clasper as the “prominent tetrahedron-shaped structure on distal margin of the basal excavation” (Gilligan et al. 2008, pg. 120). This is not to be confused with a pulvinus, which is defined as the “basal lobe of inner wall of valva” (Horak 1984). The pulvinus is sometimes referred to as a “clasper” in *Eucosma* and *Pelochrista* (Wright 2011), but the two structures should be differentiated to avoid taxonomic confusion.

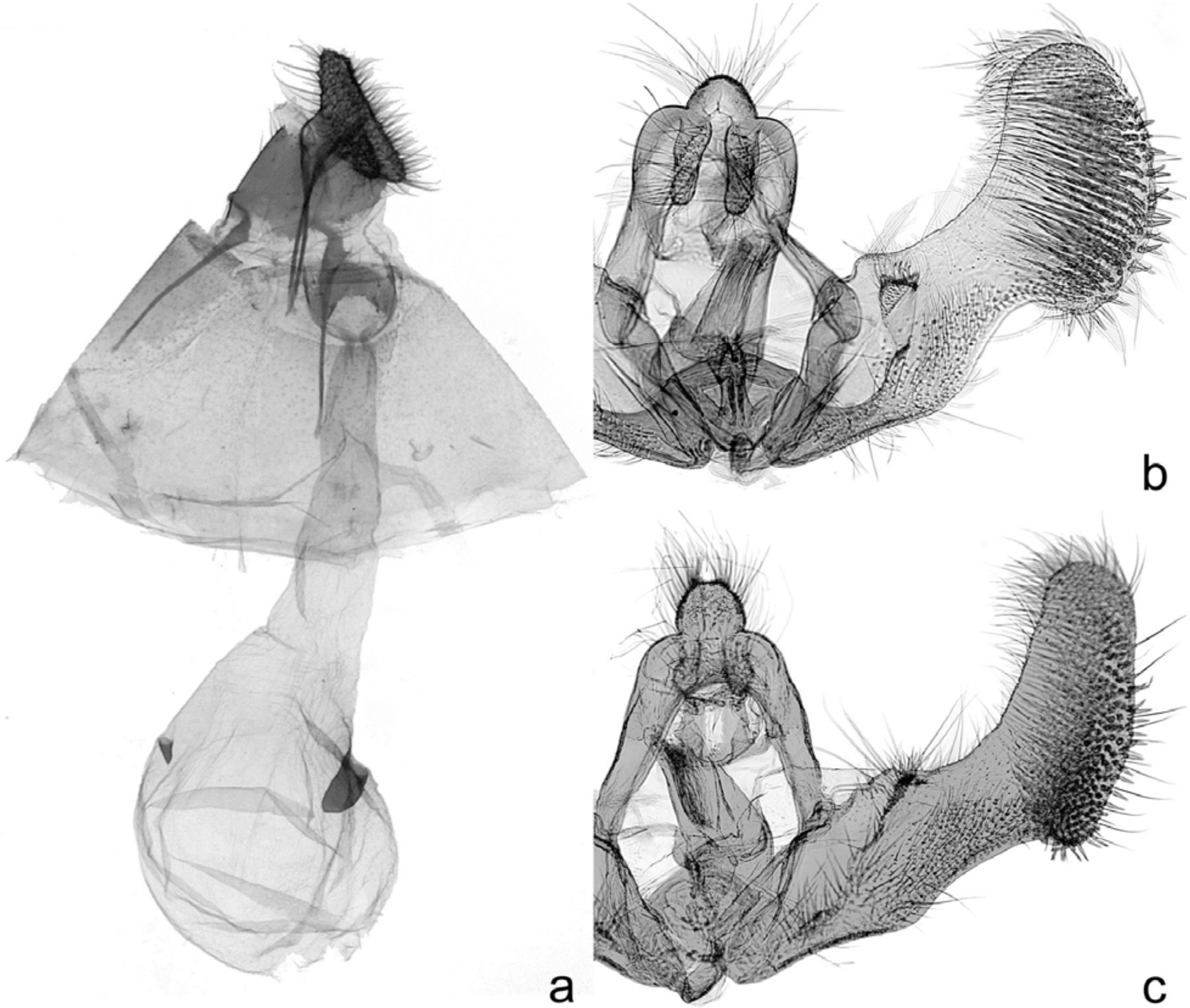


Figure 101. Examples of *Epiblema* genitalia; a: *E. walsinghami* female; b: *E. strenuana* male; c: *E. obfuscana* male.

References

Agenjo, R. 1955. Catálogo ordenador de los lepidópteros de España: Carposinidae, Tortricidae and Phaloniidae. Graellsia 13 (suppl): 14 pp. (unnumbered).

Akaike, H., 1974. A new look at the statistical model identification. IEEE Transactions on Automatic Control. 19: 716–723.

Barrett, C. G. 1885. On the value of the costal fold in the classification of *Tortrices*. The Entomologist's Monthly Magazine. 22: 1–6.

Bull, J. J., J. P. Huelsenbeck, C. W. Cunningham, D. L. Swofford and P. J. Waddell. 1993. Partitioning and combining data in phylogenetic analysis. Systematic Biology. 42: 384–397.

Cho, S., A. Mitchell, J. C. Regier, C. Mitter, R. W. Poole, T. P. Friedlander and S. Zhao. 1995. A highly conserved nuclear gene for low-level phylogenetics: elongation factor-1a recovers morphology-based tree for heliothine moths. Molecular Biology and Evolution. 12: 650–656.

Dampf, A. 1908. Über den Genitalapparat von *Rhopobota naevana* Hb. (Lep., Tortricidae) nebst Bemerkungen zur Systematik der Olethreutinae. Deutsche Entomologische Zeitschrift, Iris. 21: 304–329.

Davis, J. I., M. P. Simmons, D. W. Stevenson and J. F. Wendel. 1998. Data decisiveness, data quality, and incongruence in phylogenetic analysis: an example from the monocotyledons using mitochondrial atpA sequences. Systematic Biology. 47: 282–310.

Doyle, J.J., 1992. Gene trees and species trees: molecular systematics as one character taxonomy. Systematic Botany. 17: 144–163.

Doyle, J.J., 1995. The irrelevance of allele tree topologies for species delimitation, and a non-topological alternative. Systematic Botany. 20: 574–588.

Drummond, A. J., B. Ashton, S. Buxton, M. Cheung, A. Cooper, C. Duran, M. Field, J. Heled, M. Kearse, S. Markowitz, R. Moir, S. Stones-Havas, S. Sturrock, T. Thierer and A. Wilson. 2012. Geneious v5.6, available from http://www.geneious.com.

Farris, J. S. 1989. The retention index and the rescaled consistency index. Cladistics. 5: 417–419.

Farris, J. S., V. A. Albert, M. Källersjö, D. Lipscomb and A. G. Kluge. 1996. Parsimony jackknifing outperforms neighbor-joining. Cladistics. 12: 99–124.

Felsenstein, J., 1973. Maximum likelihood and minimum-steps methods for estimating evolutionary trees from data on discrete characters. Systematic Zoology. 22: 240–249.

Felsenstein, J., 1978. Cases in which parsimony or compatibility methods will be positively misleading. Systematic Zoology. 27: 401–410.

Felsenstein, J., 1985. Confidence limits on phylogenies: an approach using the bootstrap. Evolution. 39: 783–791.

Fernald, C. H. 1882. A synonymical catalogue of the described Tortricidae of North America north of Mexico. Transactions of the American Entomological Society. 10: 1–64.

Fernald, C. H. 1891. Tortricina, pp. 88–94. *In*: Smith, J. B. (ed.), List of Lepidoptera of Boreal America. American Entomological Society of Philadelphia.

Fernald, C. H. 1903 (1902). Family Tortricidae, pp. 448–489. *In*: Dyar, H. G. (ed.), A list of North American Lepidoptera. Bulletin of the United States National Museum, No. 52.

Fernald, C. H. 1908. The genera of Tortricidae and their types. Carpenter & Morehouse, Amherst, Massachusetts. 68 pp.

Folmer, O., M. Black, W. Hoeh, R. Lutz and R. Vrijenhoek. 1994. DNA primers for amplification of mitochondrial cytochrome c oxidase I from diverse metazoan invertebrates. Molecular Marine Biology and Biotechnology. 3: 294–299.

Gilligan, T. M., J. Baixeras, J. W. Brown and K. R. Tuck. 2012. T@RTS: Online World Catalogue of the Tortricidae (Ver. 2.0). http://www.tortricid.net/catalogue.asp.

Gilligan, T. M. and D. J. Wright. 2012. The type species of *Eucosma* Hübner (Lepidoptera: Tortricidae: Eucosmini). [Chapter 8 of this dissertation].

Gilligan, T. M., D. J. Wright and L. D. Gibson. 2008. Olethreutine moths of the midwestern United States, an identification guide. Bulletin of the Ohio Biological Survey, Volume 16(2). 334 pp.

Goloboff, P.A., J. S. Farris and K. C. Nixon. 2008. TNT, a free program for phylogenetic analysis. Cladistics. 24: 774–786.

Hannemann, H. J. 1961. Kleinschmetterlinge oder Microlepidoptera I. Die Wickler (s.str.) (Tortricidae). In: Dahl, F. (ed), Die Tierwelt Deutschlands 48. Gustav Fischer, Jena. 233 pp.

Heinemann, H. 1863. Die Schmetterlinge Deutschlands und der Schweiz. 2. Abt. Kleinschmetterlinge. Band I. Heft I. Die Wickler. Braunschweig. 248 pp.

Heinrich, C. 1920. On some forest Lepidoptera with descriptions of new species, larvae and pupae. Proceedings of the United States National Museum. 57: 53–96.

Heinrich, C. 1923. Revision of the North American moths of the subfamily Eucosminae of the family Olethreutidae. Bulletin of the United States National Museum. 123: 1–128.

Heinrich, C. 1926. Revision of the North American moths of the subfamilies Laspeyresiinae and Olethreutinae. Bulletin of the United States National Museum. 132. 1–216.

Heinrich, C. 1931. Notes on and descriptions of some American moths. Proceedings of the United States National Museum. 79: 1–16.

Horak, M. 1984. Assessment of taxonomically significant structures in Tortricinae (Lep., Tortricidae). Mitteilungen der Schweizerischen Entomologischen Gesellschaft. 57: 3–64.

Horak, M. 2006. Olethreutine moths of Australia (Lepidoptera: Tortriacidae). Monographs on Australian Lepidoptera 10. 522 pp.

Hübner, J. 1823. Zuträge zur Sammlung exotischer Schmettlinge [sic], bestehend in Betundigung einzeiner Fliegmuster neuer oder rarer Nichteuropäischer Gattungen. Zwentes Hundert. Augsburg [no publisher listed]. 32 pp., 33 pls. published [1819–1822].

Katoh, K., K. Misawa, K. Kuma and T. Miyata. 2002. MAFFT: a novel method for rapid multiple sequence alignment based on fast Fourier transform. Nucleic Acid Research. 30(14): 3059–3066.

Katoh, K. and H. Toh. 2008. Improved accuracy of multiple ncRNA alignment by incorporating structural information into a MAFFT-based framework. BMC Bioinformatics. 9: 212.

Kearfott, W. D. 1907. New North American Tortricidae. Transactions of the American Entomological Society. 33: 42–43.

Kluge, A. G. 1989. A concern for evidence and a phylogenetic hypothesis for relationships among *Epicrates* (Boidae, Serpentes). Systematic Zoology. 38: 7–25.

Kluge, A. G. and J. S. Farris. 1969. Quantitative phyletics and the evolution of Anurans. Systematic Zoology. 18: 1–32.

Lanfear, R., B. Calcott, S. Y. W. Ho and S. Guindon. 2012. PartitionFinder: combined selection of partitioning schemes and substitution models for phylogenetic analyses. Molecular Biology and Evolution. 29: 1695–1701.

Lederer, J. 1859. Classification der europäischen Tortricinen. Wiener Entomologische Monatsschrift. 3: 328–346.

Maddison, W. P. 1997. Gene trees in species trees. Systematic Biology. 46: 523–536.

Maddison, D. R. and W. P. Maddison. 2005. MacClade: analysis of phylogeny and character evolution, version 4.08. Sunderland, Massachusetts, Sinauer. http://macclade.org.

Miller, W. E. 1987. Guide to the olethreutine moths of midland North America (Tortricidae). USDA Forest Service Agriculture Handbook 660. 104 pp.

Nixon, K. C. 1999. The parsimony ratchet, a new method for rapid parsimony analysis. Cladistics. 15: 407–414.

Nixon, K.C. and J. M. Carpenter. 1996. On simultaneous analysis. Cladistics. 12: 221–242.

Obraztsov, N. S. 1946 (1945). Versuch einer systematischen Übersicht der europäischen Eucosmini-Gattungen (Lepidoptera, Tortricidae). Zeitschrift d. Wiener Entomologische Gesellschaft. 30: 20–46.

Obraztsov, N. S. 1952. *Thiodia* Hb. as not a North American genus (Lepidoptera: Tortricidae). Entomological News. 63: 146–149.

Obraztsov, N. S. 1967. Die Gattungen der palaearktischen Tortricidae. II. Die Unterfamilie Olethreutinae. 7. Teil. Tribus Eucosmini (Heinr. 1923) – Fortsetzung. Tidjschrift voor Entomologie. 110: 65–88.

Obraztsov, N. S. 1968. Die Gattungen der palaearktischen Tortricidae. II. Die Unterfamilie Olethreutinae. 8. Teil und Schluss. Tijdschrift voor Entomologie. 111: 1–48.

Pierce, F. N. and J. W. Metcalfe. 1922. The genitalia of the group Tortricidae of the Lepidoptera of the British Islands. Oundle, Liverpool, England. 101 pp.

Powell, J. A. 1968. Host associations and taxonomy of Nearctic conifer cone moths in the genus *Eucosma* (Lepidoptera: Tortricidae). Hilgardia. 39: 1–36.

Powell, J. A. 1983. Tortricidae, pp. 31–41. *In*: Hodges, R. W. (ed.), Check list of the Lepidoptera of America north of Mexico. E. W. Classey & Wedge Entomological Research Foundation, London.

Powell, J. A. and P. A. Opler. 2009. Moths of western North America. University of California Press, Berkeley. 369 pp.

Razowski, J. 1989. The genera of Tortricidae (Lepidoptera). Part II: Palaearctic Olethreutinae. Acta Zoologica Cracoviensia. 32: 107–328.

Razowski, J. 1999. Catalogue of the species of Tortricidae. Part V: Palaearctic *Eucosmina* and *Enarmoniina* (Insecta: Lepidoptera). SHILAP Revista de Lepidopterologia. 27: 437–506.

Razowski, J. 2003. Tortricidae of Europe, Volume 2, Olethreutinae. František Slamka. 301 pp.

Regier, J. C., J. W. Brown, C. Mitter, J. Baixeras, S. Cho, M. P. Cummings and A. Zwick. 2012. A molecular phylogeny for the leaf-roller moths (Lepidoptera: Tortricidae) and its implications for classification and life history evolution. PLoS ONE. 7(4): e35574. doi:10.1371/journal.pone.0035574.

Regier, J. C., C. P. Cook, C. Mitter and A. Hussey. 2008. A phylogenetic study of the “bombycoid complex” (Lepidoptera) using five proteincoding nuclear genes, with comments on the problem of macrolepidopteran phylogeny. Systematic Entomology. 33: 175–189.

Regier, J.C., A. Zwick, M. P. Cummings, A. Y. Kawahara, S. Cho, S. J. Weller, A. D. Roe, J. Baixeras-Almela, J. W. Brown, C. S. Parr, D. R. Davis, M. E. Epstein, W. Hallwachs, A. Hausmann, D. H. Janzen, I. J. Kitching, M. A. Solis, S.-H. Yen, A. Bazinet, C. Mitter. 2009. Toward reconstructing the evolution of advanced moths and butterflies (Lepidoptera: Ditrysia): an initial molecular study. BMC Evolutionary Biology 9: 280.

Siddall, M.E. 1998. Success of parsimony in the four-taxon case: long-branch repulsion by likelihood in the Farris Zone. Cladistics. 14: 209–220.

Simmons, M. P. 2004. Independence of alignment and tree search. Molecular Phylogenetics and Evolution. 31: 874–879.

Simmons, M. P. and J. V. Freudenstein. 2011. Spurious 99% bootstrap and jackknife support for unsupported clades. Molecular Phylogenetics and Evolution. 61: 177–191.

Simmons, M. P. and H. Ochoterena. 2000. Gaps as characters in sequence-based phylogenetic analyses. Systematic Biology. 49: 369–381.

Stöver. B. C. and K. F. Müller. 2010. TreeGraph 2: Combining and visualizing evidence from different phylogenetic analyses. BMC Bioinformatics. 11: 7.

Sukumaran, J. and M. T. Holder. 2010. DendroPy: A Python library for phylogenetic computing. Bioinformatics. 26: 1569–1571.

Walker, F. 1863. List of the specimens of lepidopterous insects in the collection of the British Museum. Part XXVIII. Tortricites & Tineites. British Museum (Natural History), London. 287–561 pp.

Walsingham, Lord T. de Grey. 1897. Revision of the West-Indian Microlepidoptera with descriptions of new species. Proceedings of the Entomological Society of London. 1897: 54–183.

Walsingham, Lord T. de Grey. 1914 (1909-1915). Insecta. Lepidoptera-Heterocera, Vol. IV. Tineina, Pterophorina, Orneodina, and Pyralidina and Hepialina (part), pp. 1–482. *In*: Godman, F. D. and O. Salvin (eds.), Biologia Centrali-Americana. R. H. Porter, London.

Wahlberg, N. and C. W. Wheat. 2008. Genomic outposts serve the phylogenomic pioneers: designing novel nuclear markers for genomic DNA extractions of Lepidoptera. Systematic Biology. 57: 231–242.

Wright, D. J. 2007. A new species of *Pelochrista* Lederer from eastern North America (Tortricidae). Journal of the Lepidopterists' Society. 61: 84–86.

Wright, D. J. 2011. Review of the *Eucosma pulveratana* (Walsingham) species group, with descriptions of eight new species (Tortricidae). Journal of the Lepidopterists' Society. 65: 101–118.

Yang, Z. 1993. Maximum likelihood estimation of phylogeny from DNA sequences when substitution rates differ over sites. Molecular Biology and Evolution. 10: 1396–1401.

Yang, Z., 2006. Computational Molecular Evolution. Oxford University Press, New York. 376 pp.

Zhang, A. H. and H. H. Li. 2005. Catalogue of Eucosmini from China (Lepidoptera: Tortricidae). SHILAP Revista de Lepidopterologia. 33: 265–298.

Zurawski, G. and M. T. Clegg. 1987. Evolution of higher-plant chloroplast DNA-encoded genes: implications for structure-function and phylogenetic studies. Annual Review of Plant Physiology. 38: 391–418.

Zwickl, D. J., 2006. Genetic algorithm approaches for the phylogenetic analysis of large biological sequence datasets under the maximum likelihood criterion. Ph.D. dissertation, University of Texas at Austin, Texas.

APPENDIX I

Morphological Character States

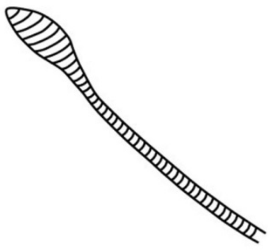
Morphological characters used in the adult and larval keys are outlined in the following appendix. Numbers below the character state illustrations correspond to character states in the keys; these are described below the illustrations. A short discussion of each character follows the state descriptions.

Adult morphological character states

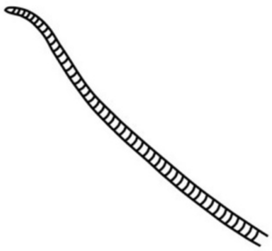
Family-level characters

The following characters are used to separate tortricid adults from those of other families, including members of the Gelechioidea, Geometroidea, Noctuoidea, Pyraloidea, Tineoidea, and Yponomeutoidea.

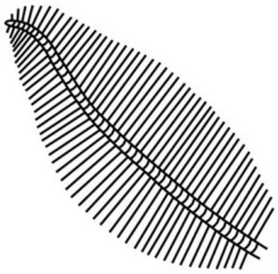
Antennae



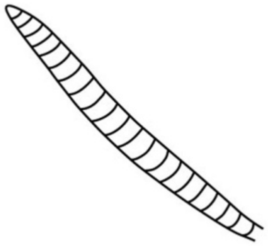
1



2



3

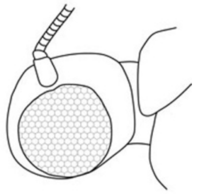


4

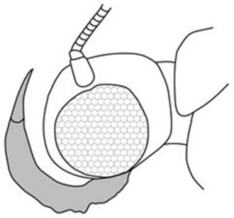
1: clubbed
2: filiform
3: pectinate
4: other

All of the tortricid species covered in the adult key have filiform antennae. Choosing any other character state will eliminate all tortricid species from the key.

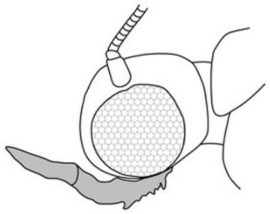
Labial palpi



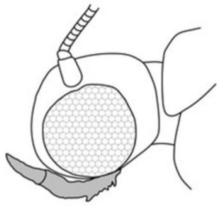
1



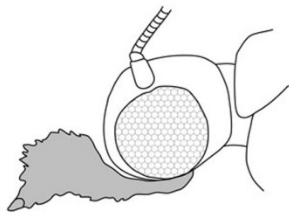
2



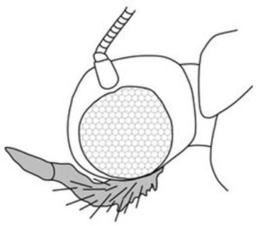
3



4



5

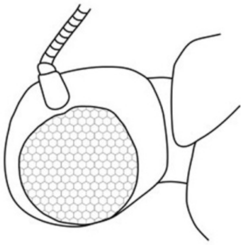


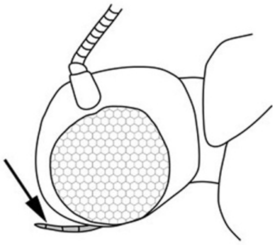
6

1: reduced or absent
2: strongly upcurved
3: moderate, upcurved
4: short, weakly upcurved
5: porrect (beaklike)
6: bristles on second segment

All of the tortricid species covered in the adult key have labial palpi that are projecting horizontally and beaklike (“porrect” – state #5). Some members of the Grapholitini have shorter, weakly upcurved palpi and are also scored for state #4. Many gelechioids can be separated from tortricids by their long, strongly upcurved palpi (state #2). Some tineoids have stiff bristles on the second segment (state #6). Other character states are shared widely across the included superfamilies.

Maxillary palpi

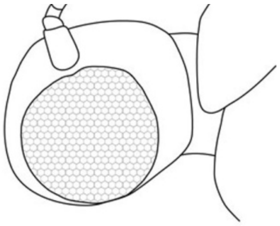

1

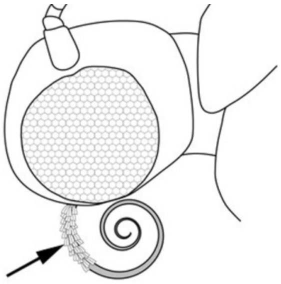

2

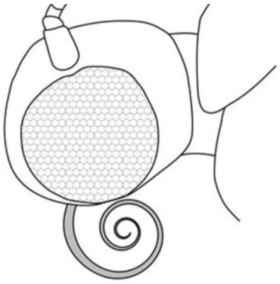
1: minute or absent
2: conspicuous

Maxillary palpi are absent (greatly reduced) in all of the tortricid species covered here. This character is useful in separating tortricoids from pyraloids, which have large, conspicuous maxillary palpi.

Proboscis


1

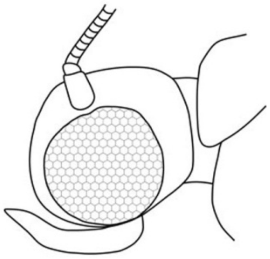

2


3

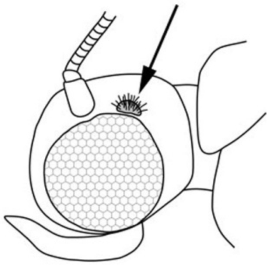
1: reduced or absent
2: scaled at base
3: unscaled

Tortricid species covered here have a proboscis which is unscaled. A scaled proboscis is present in members of the Gelechioidea and Pyraloidea. The proboscis may be greatly reduced in some members of the Noctuoidea and Tineoidea.

Chaetosema



1



2

1: absent
2: present

Chaetosema are present in all tortricid species covered here, although the structures may be difficult to detect, especially on specimens recovered from sticky traps. Other superfamilies included in the key with chaetosema present are the Geometroidea, Pyraloidea, and Yponomeutoidea.

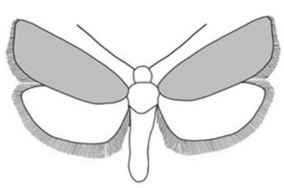
Forewing length

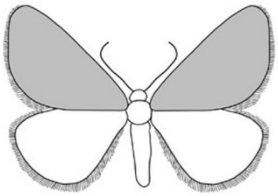
[not illustrated]

FWL: minimum (in mm)
FWL: maximum (in mm)

Forewing length (FWL) can be used to separate tortricids from some of the other superfamilies covered in the key. A measurement for the minimum and maximum FWL for each tortricid species is included in this section of the key and also in the “Wings” section. A general range is provided for each of the other superfamilies.

Wing shape



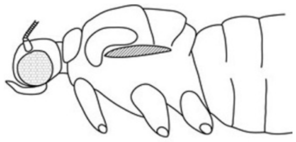


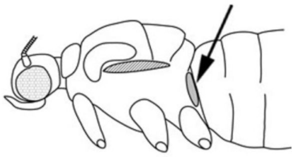
1 2

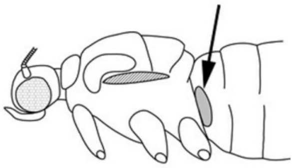
1: longer than wide
2: broadly triangular

General wing shape can be used to separate tortricids from macrolepidoptera in other superfamilies.

Tympana







1 2 3

1: absent
2: on thorax
3: on abdomen

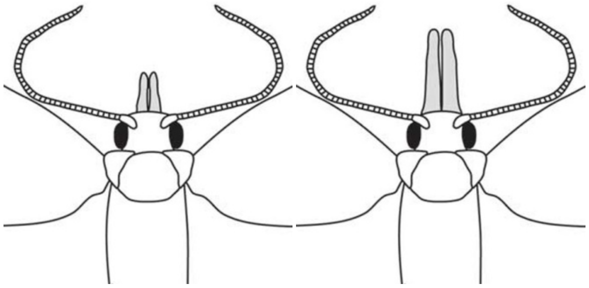
The presence of a tympanum can be used to separate tortricids from many other Lepidoptera. A tympanum is present on the thorax in most Noctuoidea, while it is present on the abdomen in many Geometroidea and Pyraloidea. This character may be difficult to detect in some specimens, especially those recovered from sticky traps.

Tortricid-specific characters

The following morphological characters are only available for scoring if one or more tortricid taxa remain in the key after scoring family-level characters. This section covers structures on the head, wings, and genitalia.

Head

Labial palpi

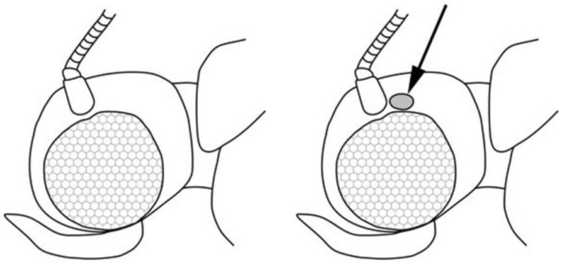


1 2

1: short/moderate
2: long

Labial palpi are elongate in many member of the Sparganothini. In taxa where it may be difficult to separate “moderate” from “long,” both character states are scored.

Ocelli

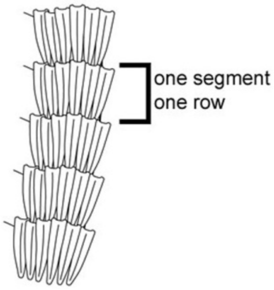


1 2

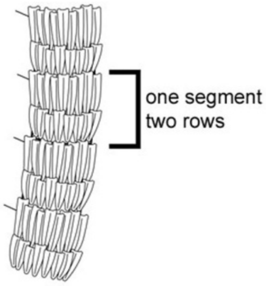
1: absent
2: present

Ocelli are present in all tortricid species covered in the key with the exception of Amorbia *cuneana* and *Amorbia emigratella*.

Antennae



1



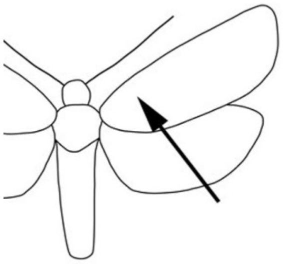
2

1: one row per segment
2: two rows per segment

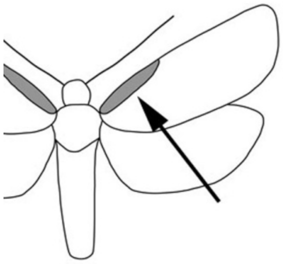
Tortricids in the subfamily Olethreutinae have one row of scales per antennal segment. Taxa in the subfamily Tortricinae have two rows of scales per antennal segment.

Wings

Forewing costal fold



1

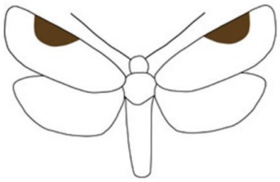


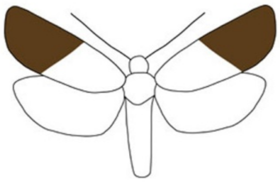
2

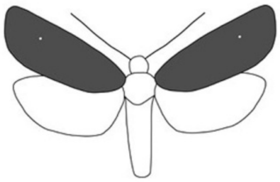
1: absent
2: present

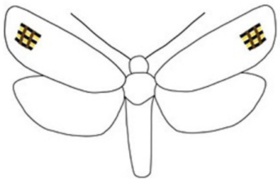
A forewing costal fold is present in the males of many tortricid species. In some genera or tribes, both character states are present. In taxa where the costal fold is very small and difficult to detect, both character states are scored.

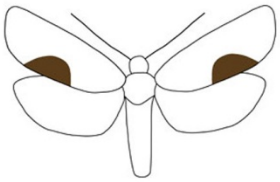
Forewing pattern

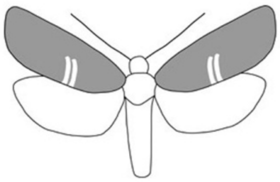

1

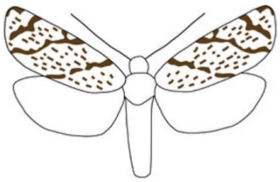

2

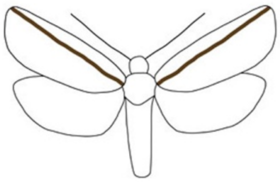

3

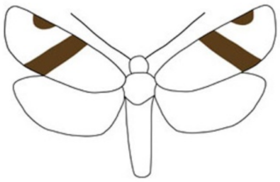

4

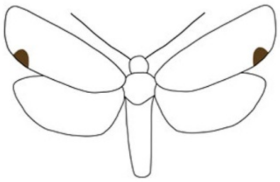

5

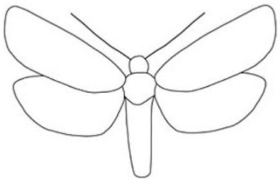

6

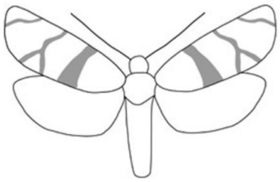

7

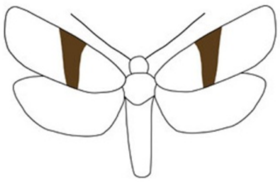

8

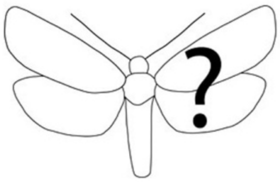

9


10


11


12


13



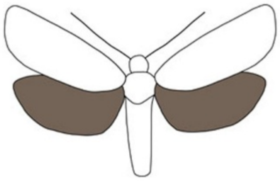
14

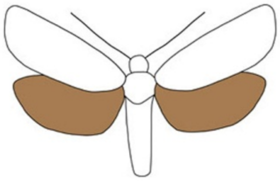
1: costal spot
2: dark apical half
3: dark brown or black
4: defined ocellus
5: dorsal spot
6: dorsal strigulae
7: reticulations
8: line to apex
9: median fascia and costal spot

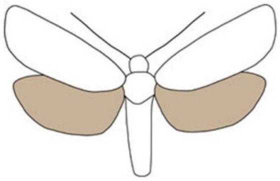
10: tornal spot
11: unmarked
12: gray or metallic bars
13: median fascia (only)
14: other (or unknown)

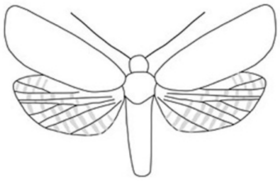
Forewing pattern, if visible, can narrow the identification choices to a genus or species group for many taxa included in the key. Major wing pattern elements are illustrated, and taxa are scored for all of the elements present. In some cases this may result in the user scoring multiple states for a single individual (“costal spot” + “median fascia and costal spot”).

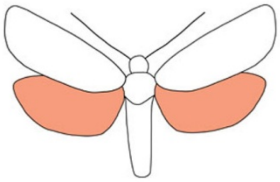
Hindwing color or markings

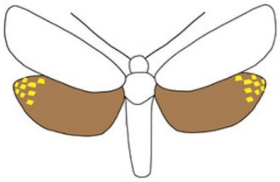

1

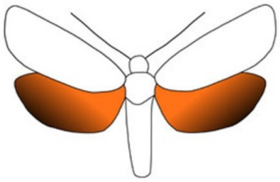

2

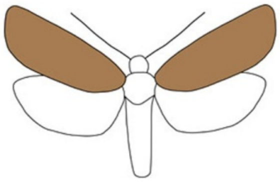

3

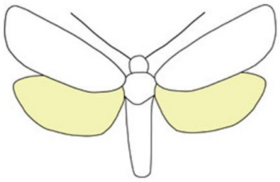

4


5


6


7


8


9


10

1: dark brown, gray, or black
2: brown or gray
3: light brown or gray
4: mottled
5: dull orange
6: brown with yellowish apex
7: orange and black
8: white
9: yellow
10: with modified scale pocket (*T. leucotreta* only)

Hindwing color or markings can be useful in separating out certain species or species groups. Some character states are species-specific (#7 = *C. pronubana*; #10 = *T. leucotreta*), while others are generalized (#1-3). In cases where it may be difficult to determine exact color (e.g., brown vs. dark brown), both states are scored.

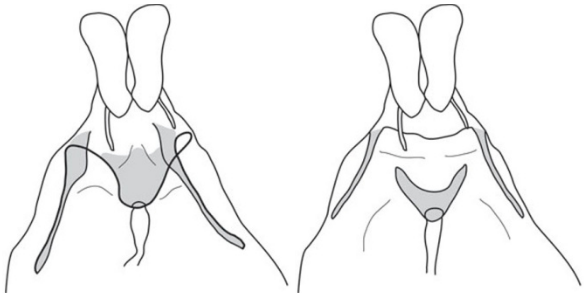
Male genitalia

The key contains male genitalia scored for each tribe, genus, and species. In order to add functionality to the key for a wide range of users, the entire genitalic capsule is illustrated and scored instead of individual structures (e.g., uncus or valvae). Tribes and genera are scored as generic “types” for those groups. Individual species are scored using photos of genitalic dissections for each species. Photos of male genitalia used in the key are illustrated in Figs. 30-43.

Female genitalia

Female genitalia are more generalized than male genitalia, and a species-level identification may be more difficult using only female genitalia (e.g., in the Archipini). As such, individual structures are listed for females which can be used in conjuction with other characters to provide an identification.

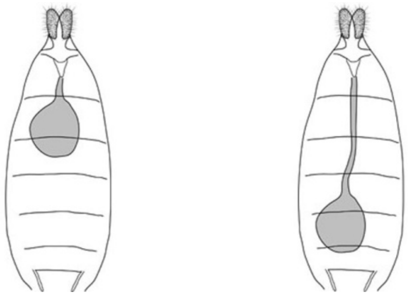
Sterigma connected to apophyses anteriores


1 2

1: yes (Tortricinae)
2: no (Olethreutinae)

This character, although difficult to interpret in some taxa, can be used to broadly separate members of the Tortricinae from those of the Olethreutinae. In the key, an exception is found in the genus *Cryptaspasma*, whose members retain the tortricine character state (#1).

Ductus bursae length

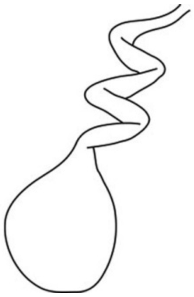

1 2

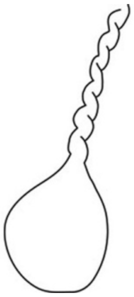
1: short
2: long

The length of the ductus bursae in relation to the abdomen can be used to differentiate certain species. In cases where it is difficult to determine if the structure is “short” or “long,” both character states are scored.

Ductus bursae structure


1


2


3

1: straight
2: twisted
3: spiraled (tightly)

A twisted or spiraled ductus bursae is present in several taxa included in the key. In cases where it may be difficult to determine if the ductus bursae is twisted, both states “straight” and “twisted” are scored.

Cestum in ductus bursae


1


2

1: absent
2: present

A cestum is present in the ductus bursae in a majority of the Archipini included in the key, as well as in a few members of other tribes.

Signa


1


2


3


4

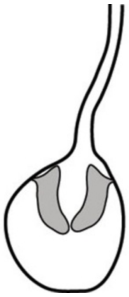

5


6


7


8


9

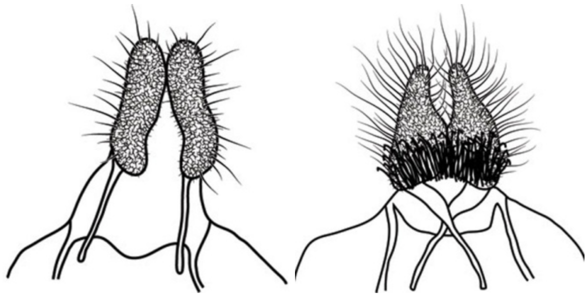

10


11

1: absent
2: one – bandlike
3: one – bladelike
4: one – daggar-shaped
5: one – scobinate patch
6: one – thornlike
7: one – variable ridge
8: two – bladelike
9: two – thornlike
10: two – thumblike
11: variable sclerotized (Cochylini)

The presence or absent of signa in the corpus bursae can be used to distinguish closely related species in several genera (e.g., *Clepsis peritana* vs. *Clepsis virescana*). The structure and number of signa can be used to separate out various genera and species groups.

Papillae anales



1: “normal”
2: “modified” (Cnephasiini)

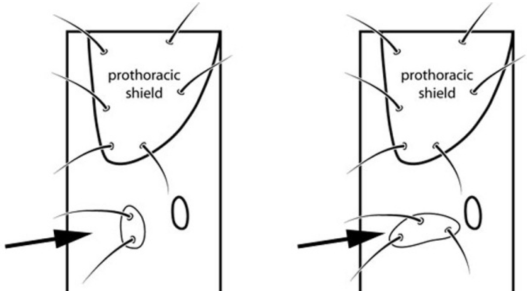
This character can be used to separate out members of the Cnephasiini (*Cnephasia* and *Decodes*) from other taxa in the key.

Larval morphological character states

Family-level characters

The following characters are used to separate tortricid larvae from those of other families, including members of the Geometroidea, Noctuoidea, Pyraloidea, other microlepidoptera, and other macrolepidoptera.

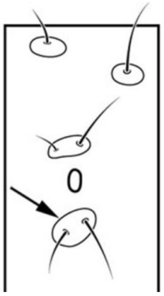
Prespiracular pinaculum on prothorax



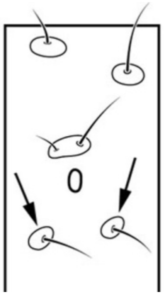
1: bisetose
2: trisetose

Tortricidae larvae have a trisetose prespiracular pinaculum (L-group). This character can be used to broadly separate the larvae of microlepidoptera from those of macrolepidoptera.

L1 and L2 setae on A2-7



1

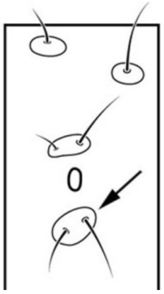


2

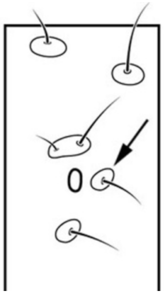
1: close together or on same pinaculum
2: widely separated

The L1 and L2 setae are on the same pinaculum in all of the tortricid species treated here. These two setae are widely separated and on separate pinacula in other families.

L1 position on abdomen



1

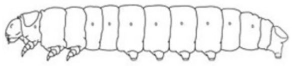


2

1: below spiracle
2: posterior to spiracle

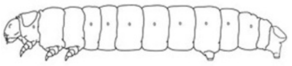
If the L1 and L2 setae are separate (see the previous character), the L1 seta may be displaced dorsally and located next to (usually posterior to) the spiracle. This character state is found in many Noctuoidea and in some other macrolepidoptera.

Segments with prolegs



1

2

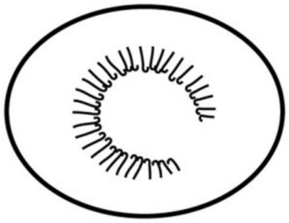


3

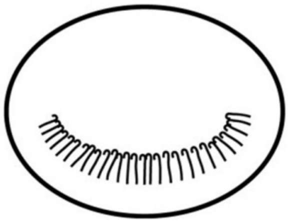
1: prolegs on A3-6 and A10
2: prolegs on A5-6 and A10
3: prolegs on A6 and A10 only (or greatly reduced on A5)

A majority of Lepidoptera larvae, including those of Tortricidae, have prolegs on abdominal segments A3-6 and A10. Some Noctuoidea have prolegs on A5-6 and A10. A majority of Geometroidea have prolegs on A6 and A10 only (although there is a greatly reduced proleg on A5 in some species).

Crochet arrangement



1

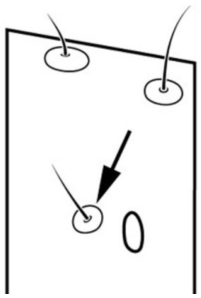


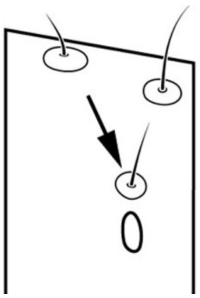
2

1: circle or ellipse
2: mesoseries (or series)

All of the tortricid larvae covered here have crochets on the prolegs arranged in a circle or ellipse. Many of the macrolepidopteran families have larvae with crochets arranged in a mesoseries.

SD1 position on A8


1


2

1: anterior to spiracle
2: dorsal to spiracle

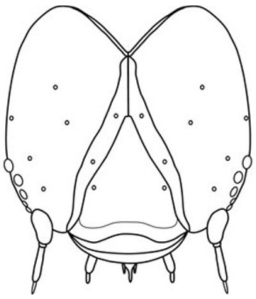
The SD1 position on all of the tortricid taxa covered here is anterior to the spiracle. In a limited number of other Tortricidae (some *Eucosma*), and in other families, it may be dorsal to the spiracle.

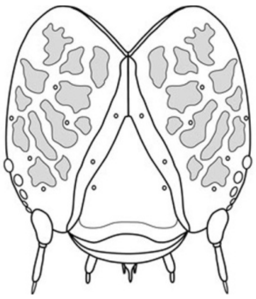
Tortricid-specific characters

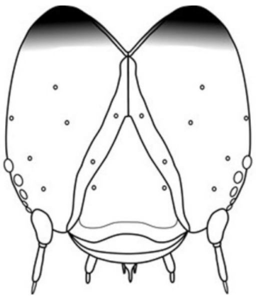
The following morphological characters are only available for scoring if one or more tortricid taxa remain in the key after scoring family-level characters. This section covers structures on the head, thorax, and abdomen.

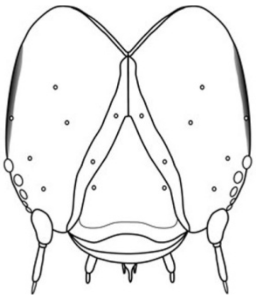
Head

Markings


1


2

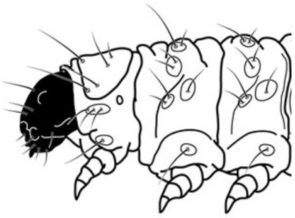

3

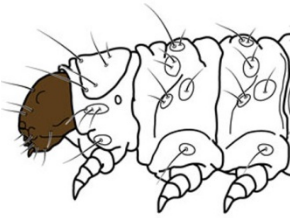

4

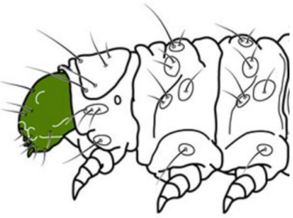
1: unmarked
2: mottled
3: posterior shading
4: lateral line or shading

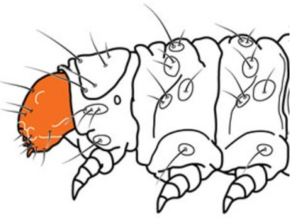
Head markings have limited utility in larval diagnostics because of their variability between instars. It is also possible for head markings to vary significantly within the same species. Dark posterior or lateral shading appears to vary less than overall mottling within the same species, although some markings are not present until the final instar. When head markings are questionable for a particular species, more than one state is scored.

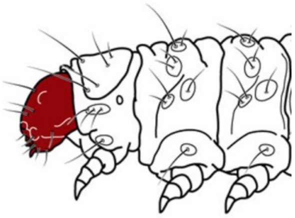
General color

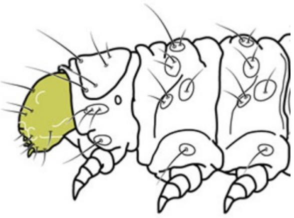

1


2


3


4

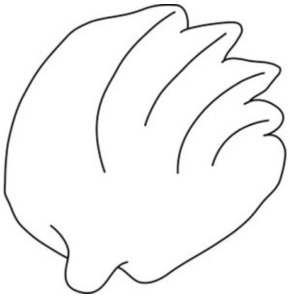

5

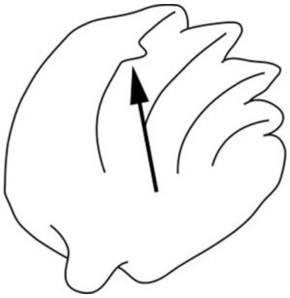

6

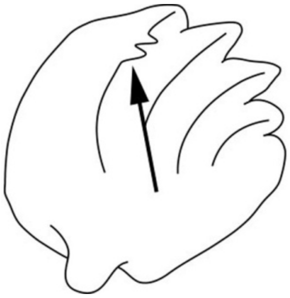
1: black
2: brown
3: green
4: orange
5: red
6: yellow

Head color has limited use for distinguishing the larvae of most species due to the variability of this character within species and between instars. In some species, such as *E. postvittana*, differentiating between a “light” (yellowish-brown) and “dark” (black) head can be helpful. Many species are scored with multiple states, allowing for combinations of colors or variability between instars.

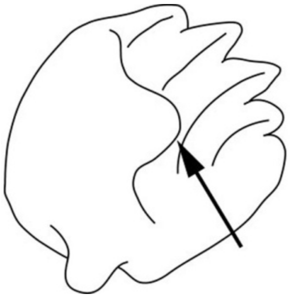
Mandibles


1


2


3


4

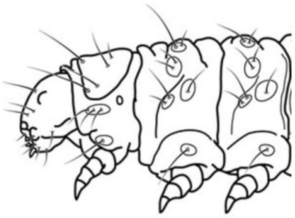

5

1: no inner teeth
2: one inner tooth
3: two inner teeth
4: multiple inner teeth
5: large retinaculum

Mandible morphology was examined for available specimens; other species were scored from descriptions in the literature, primarily from Dugdale et al. (2005). Mandible terminology follows Passoa (1985), and varies slightly from that used by Dugdale et al. (2005). Specifically, the “retinaculum” is treated as separate from the “inner teeth.” The number of inner teeth was observed to vary between instars of the same species, so the presence or absence of inner teeth or a retinaculum may be as useful as coding specific numbers of teeth. The shape of the “outer teeth” (*sensu* Passoa 1985) changes with different larval host plants and feeding time, and was not useful as a diagnostic character across the entire family. More specific mandible differences may be useful at a local level (within a genus or species group).

Thorax

Legs



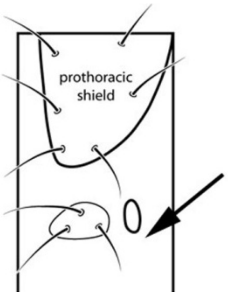
1

2

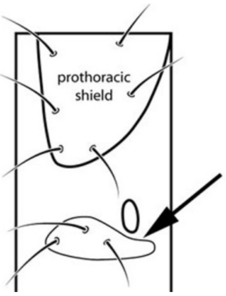
1: unmarked
2: marked

The presence or absence of “marked” (heavily sclerotized and/or pigmented) legs is useful in separating certain groups of species. In many Archipini, larvae can have marked or unmarked legs, and are scored for both states.

Prothoracic prespiracular pinaculum position



1

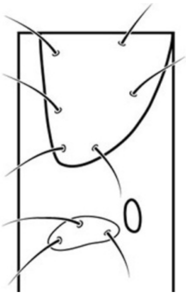


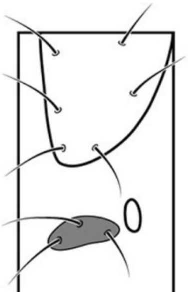
2

1: pinaculum does not extend beneath spiracle
2: pinaculum extends below and/or beyond spiracle

The prothoracic prespiracular pinaculum (L-group) extending below and/or beyond the spiracle is useful in distinguishing some species of Grapholitini (*Cryptophlebia*, *Thaumatotibia*, etc.) and Cochylini.

Prothoracic prespiracular pinaculum color

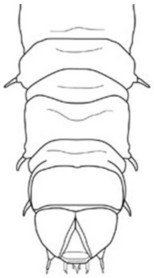

1

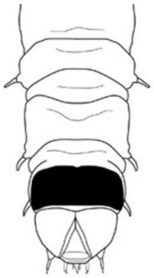

2

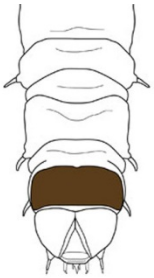
1: light (body color)
2: dark (brown or black)

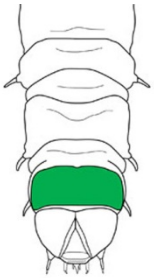
The dark coloration of the prothoracic prespiracular pinaculum (and possibly other pinacula – see “pinacula color”) is useful in distinguishing the larvae of some genera, such as *Gymnandrosoma* and *Pammene*.

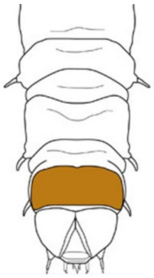
Prothoracic shield general color

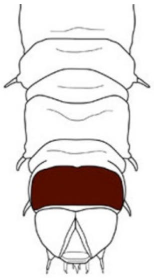

1

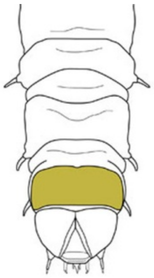

2


3


4


5

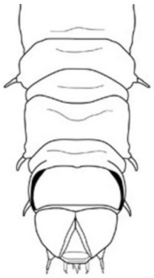

6


7

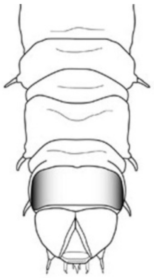
1: body color
2: black
3: brown
4: green
5: orange
6: red
7: yellow

Prothoracic shield color is not useful for distinguishing the larvae of most species due to the variability of this character within species and between instars. Many species are scored with multiple states, allowing for combinations of colors or variability between instars.

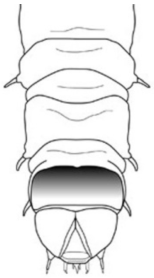
<u>Prothoracic shield markings</u>



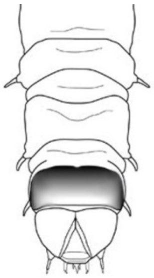
1



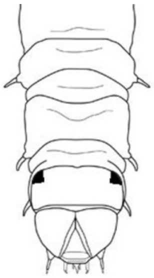
2



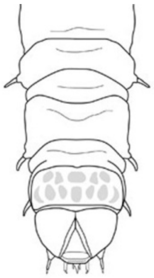
3



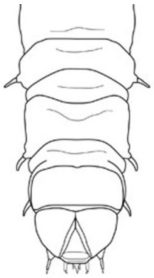
4



5



6



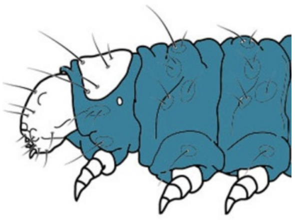
7

1: dark lateral line
2: lateral shading or patch
3: posterior shading or patch
4: posterolateral shading or patch
5: posterolateral patch
6: mottled
7: unmarked

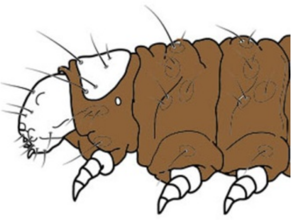
Prothoracic shield markings are also variable, both within species and between instars. Some species, such as *Amorbia* spp. and *C. pronubana*, have particular markings that help distinguish them from similar species.

Abdomen

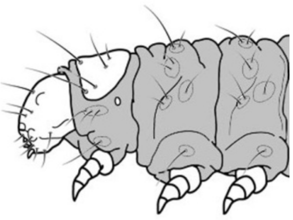
<u>Body color (abdomen and thorax)</u>



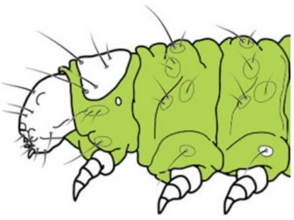
1



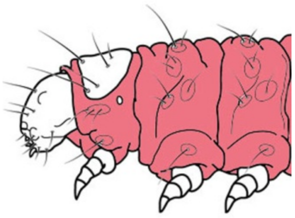
2



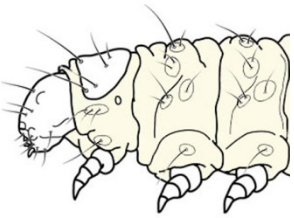
3



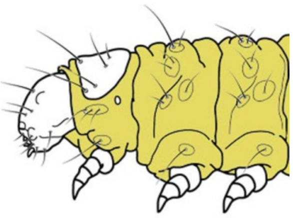
4



5



6

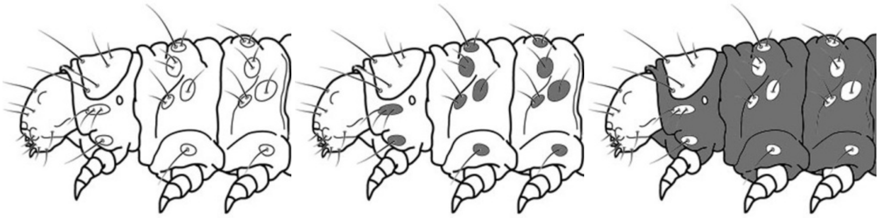


7

1: blue
2: brown
3: gray
4: green
5: pink, red, or orange
6: white
7: yellow

General body color varies with the host plant that is utilized, making this character too variable to be diagnostic except in a few cases. It is most useful in distinguishing the larvae of many Grapholitini, which are pink, red, orange, or white.

Pinacula color

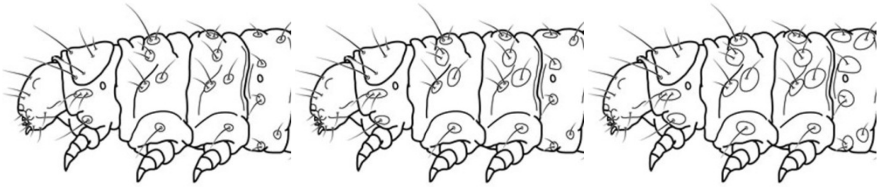


1 2 3

1: body color
2: dark
3: pale

Pinacula color (pigmentation or heavy sclerotization) is useful in distinguishing several species included here. Pinacula may be darker than body color, such as in *P. fasciana* and *T. viridana*, or lighter than body color, such as in some *Cydia* species.

Pinacula size

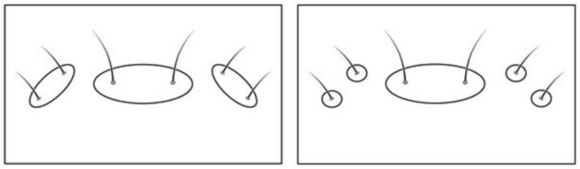


1 2 3

1: small
2: moderate
3: large

Pinacula size is useful for distinguishing some species of Grapholitini. As size is subjective, this character is scored as two states for most taxa, preventing errors in differentiating between small-moderate and moderate-large.

Dorsal and subdorsal setal arrangement: D1 and SD1 on A9

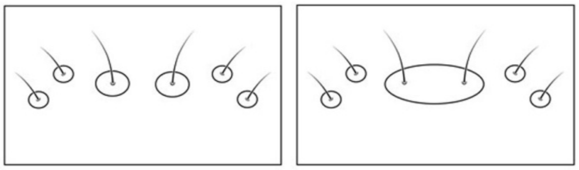


1 2

1: D1 and SD1 on same pinaculum
2: D1 and SD1 on separate pinacula

As a general rule, D1 and SD1 located on the same pinaculum on abdominal segment A9 is characteristic of larvae in the Olethreutinae, while D1 and SD1 located on separate pinacula on A9 is characteristic of larvae in the Tortricinae. However, many exceptions are found throughout the family.

Dorsal and subdorsal setal arrangement: D2s on A9

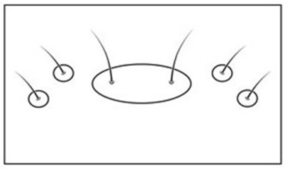


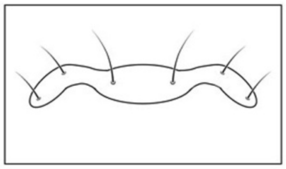
1 2

1: D2 setae on separate pinacula
2: D2 setae on single “saddle” pinacula

The D2 setae on abdominal segment A9 located on a single “saddle” pinacula is a general characteristic of most tortricid larvae. The pinacula of some species of internal-feeding Grapholitini are weakly sclerotized, resulting in the appearance of the D2 setae on separate pinacula; these species are scored for both character states in the key.

Dorsal and subdorsal setal arrangement: D1, D2, and SD1 on A9

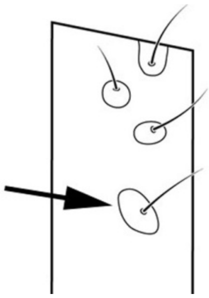

1

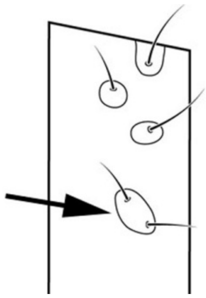

2

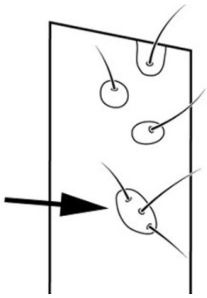
1: D1, D2, and SD1 setae not on same enlarged pinaculum
2: D1, D2, and SD1 all on same enlarged (ill-defined) pinaculum

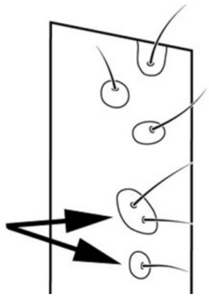
Two taxa included in the key, *Cryptaspasma* spp. and *T. batesi*, have the D1, D2, and SD1 setae all on the same enlarged pinaculum that consists of a fusion of the dorsal “saddle” pinaculum and the subdorsal pinacula.

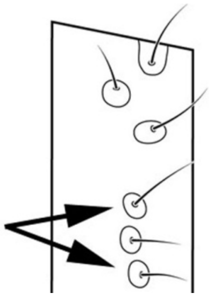
Lateral setal arrangement: L pinaculum on A9


1


2


3

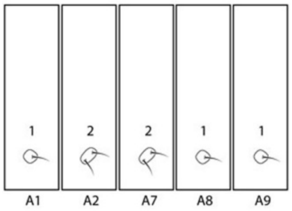

4


5

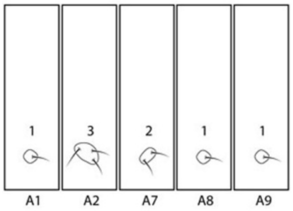
1: unisetose
2: bisetose
3: trisetose (all setae on same pinaculum)
4: trisetose (2 setae on one, 1 seta on other pinaculum)
5: trisetose (all setae on separate pinacula)

The number of setae in the L-group on abdominal segment A9 is useful in differentiating between many of the species included here. In many of the species with a trisetose L-group, the number of pinacula vary between individuals, and more than one state is scored in the key.

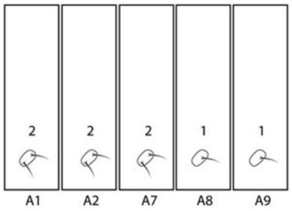
Subventral setal arrangement: SV group on A1,2,7,8,9



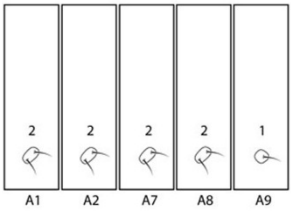
1



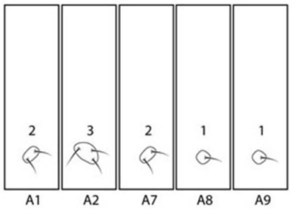
2



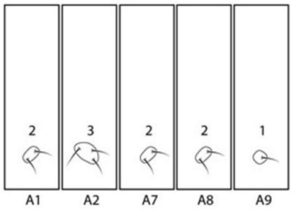
3



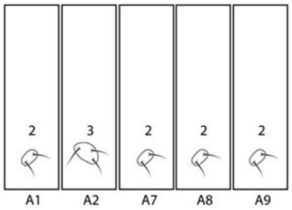
4



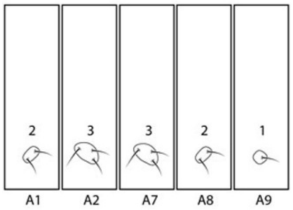
5



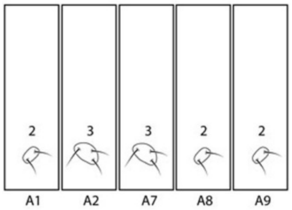
6



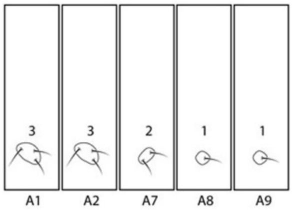
7



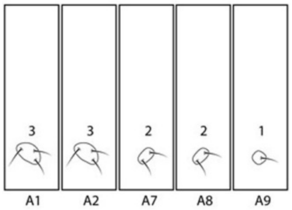
8



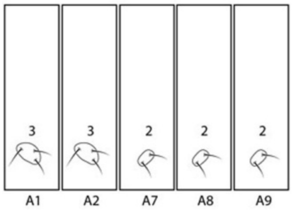
9



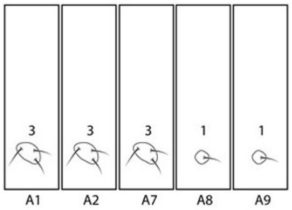
10



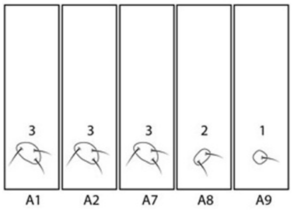
11



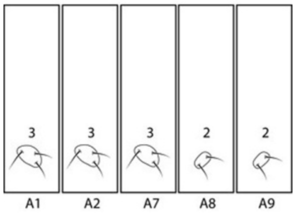
12



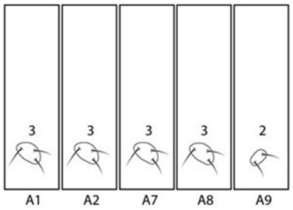
13



14



15

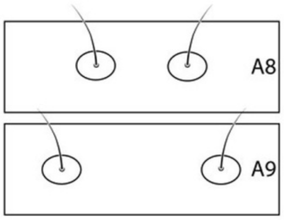


16

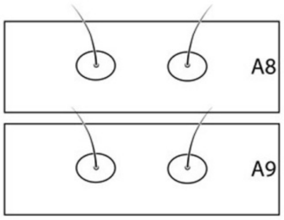
1: (1:2:2:1:1)
2: (1:3:2:1:1)
3: (2:2:2:1:1)
4: (2:2:2:2:1)
5: (2:3:2:1:1)
6: (2:3:2:2:1)
7: (2:3:2:2:2)
8: (2:3:3:2:2)
9: (2:3:3:2:1)
10: (3:3:2:1:1)
11: (3:3:2:2:1)
12: (3:3:2:2:2)
13: (3:3:3:1:1)
14: (3:3:3:2:1)
15: (3:3:3:2:2)
16: (3:3:3:3:2)

Subventral setal counts on abdominal segments A1,2,7,8,9 are useful in distinguishing several genera and species covered in the key. This character varys extensively in some taxa, such as *Cydia* spp., while it is consistent across other taxa, such as in many of the Archipini (with counts of 3:3:3:2:2).

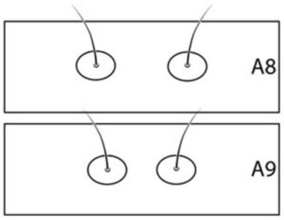
Ventral setal arrangement: V setae on A8-9



1



2

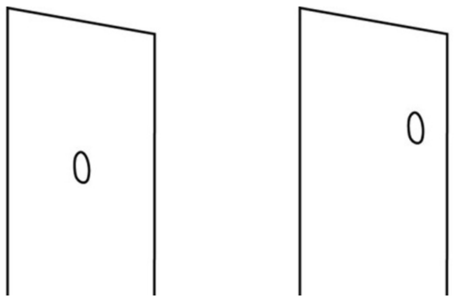


3

1: distance btwn Vs on A9 ca. 1.25-3X distance btwn Vs on A8
2: distance btwn Vs on A9 ca. equal to distance btwn Vs on A8
3: distance btwn Vs on A9 less than distance btwn Vs on A8

Spacing of the ventral setae on abdominal segments A8 vs. A9 is useful in distinguishing between several species included in the key. Spacing between the V setae on A7 was also examined for some species but was determined to not be as useful.

Spiracle position on A8

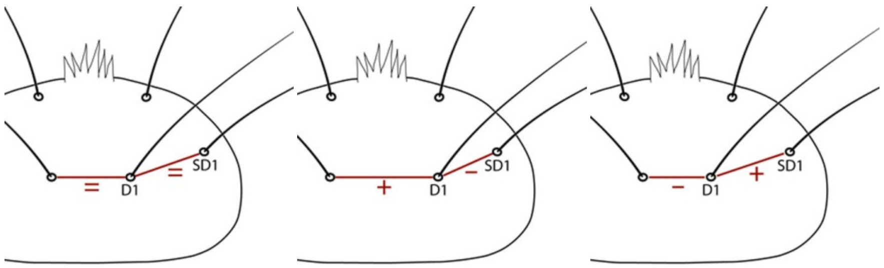


1 2

1: spiracle in anterior 2/3 of segment
2: spiracle in posterior 1/3 of segment and displaced dorsally

The spiracle on abdominal segment A8 is located in the posterior 1/3 of the segment and displaced dorsally in some Grapholitini, such as *Cryptophlebia*, *Ecdytolopha*, and *Gymnandrosoma*. This character varies within *T. leucotreta*, and is scored for both states in that taxon.

Anal shield: distance between D1 and SD1 setae

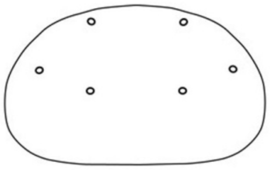


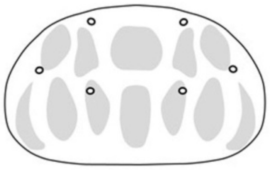
1 2 3

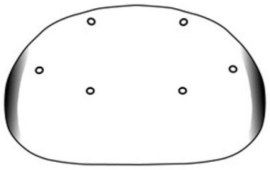
1: distance btwn D1-D1 equal to distance btwn D1-SD1
2: distance btwn D1-D1 greater than distance btwn D1-SD1
3: distance btwn D1-D1 less than distance btwn D1-SD1

Relative distance between the D1-D1 and D1-SD1 setae on the anal shield can be diagnostic for some species. In taxa where distance varied or was questionable, multiple character states are scored.

<u>Anal shield markings</u>


1

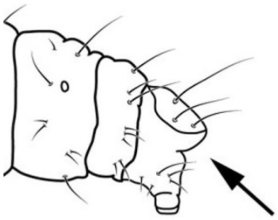

2

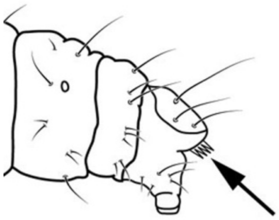

3

1: unmarked
2: mottled
3: lateral shading

Anal shield markings are variable and were not scored for most taxa included in the key. In instances where they were found to be useful, specific states (“mottled” and “lateral shading”) were included.

<u>Anal comb</u>


1

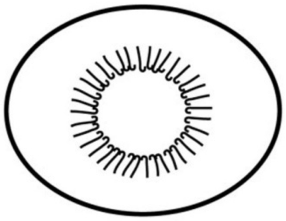

2

1: absent
2: present
3: number of teeth [not illustrated]

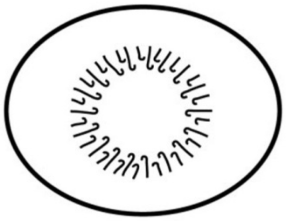
The presence or absence of an anal comb is useful in distinguishing between large groups of tortricid larvae. In general, internal feeders (many Olethreutinae) lack an anal comb, while it is present in external feeders (many Tortricinae). Several notable exceptions in the key are some Grapholitini, which have an anal comb, and *Cryptophlebia*, which were found to have both character states present in the same species.

The number of teeth in the anal comb is also a useful diagnostic character. The key includes a range for the number of teeth, allowing for variation and problems in counting very small teeth.

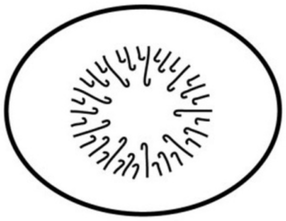
Abdominal prolegs: crochet length



1



2

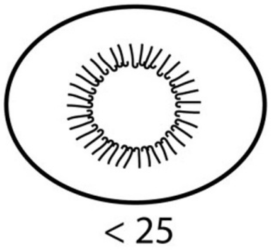


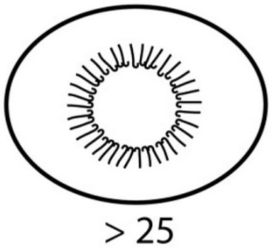
3

1: uniordinal
2: biordinal
3: triordinal

The length of crochets on the abdominal prolegs was scored for taxa when the information was available. In cases where it is difficult to determine crochet length (biordinal vs. weakly triordinal), multiple character states are scored.

Abdominal prolegs: number of crochets





1 2

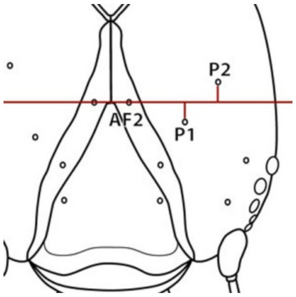
1: less than 25
2: more than 25
3: exact count [not illustrated]

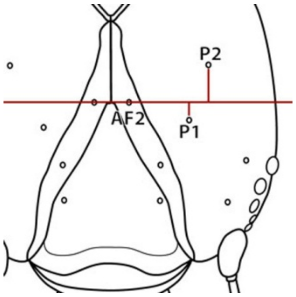
Crochet counts are included for most species in the key. These are scored as a range (>25 vs. <25), or as exact counts. In general, it is more useful to differentiate between low numbers and high numbers of crochets, and an exact count is not necessary. In species that may have exactly 25 crochets, both character states are coded.

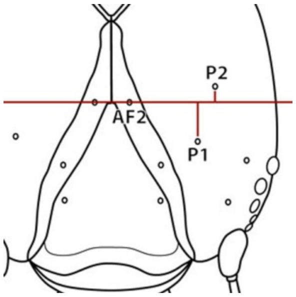
Characters examined but not used in the larval key

The following characters were examined for several taxa but were not included in the key due to their inability to separate taxa across the family. Some characters are useful at a local (species group) level, and others are too variable or difficult to measure. Reasons for not including these characters in the key are discussed below.

Head setae: AF2 versus P1 and P2 spacing





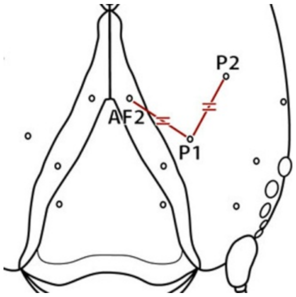


1 2 3

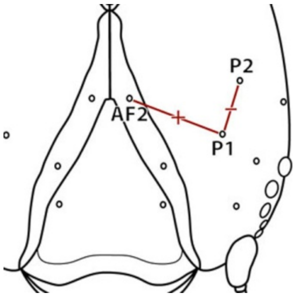
1: line btwn AF2s midway btwn P1 and P2
2: line btwn AF2s closer to P1 than P2
3: line btwn AF2s closer to P2 than P1

The position of AF2 setae on the head in relation to P1 and P2 was measured in an attempt to separate *E. postvittana* from similar species in California. Position of the P setae was variable, with several species scored for all three character states. This variability may be due to the character being difficult to quantify, as position and angle of the head greatly affects the perceived distance between setae.

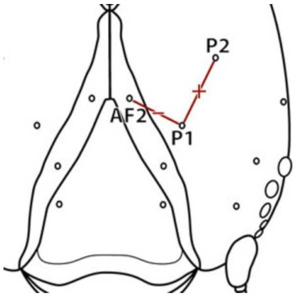
Head setae: P1, P2 and AF2 ratios



1



2

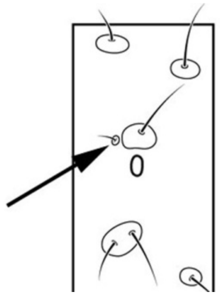


3

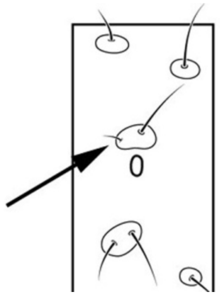
1: distance btwn AF2 & P1 equal to distance btwn P1 & P2
2: distance btwn AF2 & P1 greater than distance btwn P1 & P2
3: distance btwn AF2 & P1 less than distance btwn P1 & P2

The distance between head setae AF2 and P1 versus the distance between setae P1 and P2 was measured in several species. As with the proceeding character, position and angle of the head makes comparing measurements difficult. The setal distances were equal in a majority of the species examined (character state #1).

Subdorsal setal arrangement: SD1 and SD2



1



2

1: on separate pinacula
2: on joined pinaculum
3: SD2 highly reduced or absent [not illustrated]

The position and presence of the SD2 seta on abdominal segments A1-8 is useful in separating some species groups; however, this character was found to be too difficult to observe and quantify across the entire family. Reduction of the size of SD2 and intraspecific variation complicated the scoring of this character, and several taxa were scored for all three states.

Other characters not illustrated

The following characters were examined for some taxa but were determined to not be useful in multiple species diagnoses. Further examination of these characters is needed to determine if they are useful in separating tortricid larvae at higher levels.

- Stemmata (ocelli) size, spacing, and position on the head
- Shape of the adfrontal sutures on the head
- Antennal size/coloration
- Setal position on the prothoracic shield
- Integument spinule presence vs. absense (spiny vs. smooth)
- Spiracle size in relation to pinacula sizes
- Setal length (on head, shields, and abdomen)

APPENDIX II

List of Sampled Specimens

Collogenes albocingulata: TORTAG-TMG-533, Australia, 2009, T. M. Gilligan, det. T. M. Gilligan & M. Horak; ***Cryptaspasma brachyptycha***: ANIC DNA 002783, Australia, M. Horak, det. M. Horak; ***Cryptaspasma sordida***: TOR-DNA-382, Australia, 2009, T. M. Gilligan, det. T. M. Gilligan & M. Horak; ***Dactylioglypha* sp.**: TOR-DNA-582, Australia, 2009, T. M. Gilligan, det. T. M. Gilligan & M. Horak; ***Metrioglypha* sp.**: TOR-DNA-581, Australia, 2009, T. M. Gilligan, det. T. M. Gilligan & M. Horak; ***Fibuloides japonica***: SWC-07-2037, Korea, det. C. Soowon & J. Sohn; ***Sonia paraplesiana***: TOR-DNA-0254, AL: Baldwin Co., Weeks Bay, 21 Jun 2008, Gilligan, Wright, Gibson, det. T. M. Gilligan; ***Rhopobota dietziana***: TOR-DNA-0257, MS: Oktibbeha Co., 27 Jun 2008, T. M. Gilligan, det. T. M. Gilligan; ***Rhyacionia pinivorana***: TOR-DNA-0425, Spain, Castello, Parc Natural de Penyagolosa, 22 Jun 2009, Gilligan, Baixeras, det. T. M. Gilligan & J. Baixeras; ***Notocelia culminana***: TOR-DNA-0512, OH: Hancock Co., 31 Aug 2009, T. M. Gilligan, det. T. M. Gilligan; ***Thiodia citrana***: TORTAG-TMG-101, Spain: Parc Natural de Penyagolosa, 22 Jun 2009, T. M. Gilligan, J. Baixeras, det. T. M. Gilligan; ***Epiblema foenella***: KTP-06-0124-6, Korea, det. Park & Kyu Tek; ***Epiblema abruptana***, KTP-94-0521, USA, det. Park & Kyu Tek; ***Epiblema otiosana***: TOR-06-0032, OH: Wyandot Co., Killdeer WLA, 16 Aug 2006, T. M. Gilligan, det. T. M. Gilligan; ***Epiblema carolinana***: TOR-06-0079, KY: Gallatin Co., Markland Dam, 13 Aug 2006, L. D. Gibson, det. L. D. Gibson; ***Epiblema gibsoni***: TOR-06-0093, KY: Larue Co., Cecil Ridge, 4 mi. W of New Haven, 31 Jul 2006, L. D. Gibson, det. L. D. Gibson; ***Epiblema strenuana***: TOR-DNA-511, OH: Hancock Co., 16 Aug 2009, T. M. Gilligan, det. T. M. Gilligan; ***Eucosma bobana***: TOR-DNA-218, WA: Chelan Co., 10 mi. NE Leavenworth, 8 Jul 2010, T. M. Gilligan, det. T. M. Gilligan; ***Eucosma siskiyouana***, TOR-DNA-222, WA: Chelan Co., 10 mi. NE Leavenworth, 9 Jul 2010, T. M. Gilligan, det. T. M. Gilligan; ***Eucosma* nr. *bobana***: TOR-DNA-0632, Albany Co., WY, south of Happy Jack Rd, NE of Pole Mt, 12 Jun 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Eucosma* nr. *bobana***: TOR-DNA-0646, Albany Co, just north of Rd 726, 1 Jul 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Eucosma sonomana***: TOR-DNA-0633, Albany Co., WY, just SW of road 712H off Happy Jack Rd, 13 Jun 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Eucosma morrisoni***: DJW-06-0002 WY: Albany Co., 6 Jul 2006, D. J. Wright, det. D. J. Wright; ***Eucosma caniceps***: TOR-06-0007, UT: Sanpete Co., Ephraim Canyon, 20 Jul 2006, T. M. Gilligan, det. T. M. Gilligan & D. J. Wright; ***Eucosma caniceps***: TOR-DNA-0663, Albany Co. west of Woods Landing, Hyw 130, 30 Jul 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Eucosma maculatana***: DJW-06-0012, ID: Oneida Co., 15 Jul 2006, D. J. Wright, det. D. J. Wright; ***Eucosma sombreana***: TOR-06-0022, OH: Wyandot Co., Killdeer WLA, 16 Aug 2006, T. M. Gilligan, det. T. M. Gilligan; ***Eucosma derelicta***: DJW-06-0026, OH: Wyandot Co., Killdeer WLA, 16 Aug 2006, D. J. Wright, det. D. J. Wright; ***Eucosma circulana***: TOR-06-0098, KY: McCracken Co., Paducah, Littleville zip track, 27 Jun 2008, L. D. Gibson, det. L. D. Gibson; ***Eucosma metzneriana***: TORTAG-TMG-175, Romania: Cluj, 26 May 2009, T. M. Gilligan & J. Powell, det. T. M. Gilligan; ***Eucosma serpentana***: TOR-DNA-0203, MT: Custer Co., 2.5 mi. S. Miles City, 12 Jul 2010, D. J. Wright, det. D. J. Wright; ***Eucosma agricolana***: TOR-DNA-0205, WA: Chelan Co., FR 7520, N of Eagle Cr. Rd., 9 Jul 2010, D. J. Wright, det. D. J. Wright; ***Eucosma agricolana***: TOR-DNA-221, WA: Chelan Co., 10 mi. NE Leavenworth, 8 Jul 2010, T. M. Gilligan, det. T. M. Gilligan; ***Eucosma agricolana***: TOR-DNA-0635, Albany Co., WY, 2217 Sky View Lane, 22 Jun 2011, J. Nordin det. J. Nordin & D. J. Wright; ***Eucosma subflavana***: TOR-DNA-0206, WA: Chelan Co., FR 7520, N of Eagle Cr. Rd., 9 Jul 2010, D. J. Wright, det. D. J. Wright; ***Eucosma biquadrana***: TOR-DNA-0209, WA: Chelan Co., FR 7520, N of Eagle Cr. Rd., 9 Jul 2010, D. J. Wright, det. D. J. Wright; ***Eucosma denverana***: TOR-DNA-404, CO: Cheyenne Co., Wild Horse, post office, 11 Aug 2009, T. M. Gilligan, det. T. M. Gilligan; ***Eucosma ridingsana***: TOR-DNA-468, CO: Lincoln Co., Hugo, hospital, 11 Aug 2009, T. M. Gilligan, det. T. M. Gilligan; ***Eucosma ridingsana***: TOR-DNA-0676, Albany Co., WY, 2217 Sky View Lane, 18 Aug 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Eucosma pulveratana***: TOR-DNA-476, CO: Cheyenne Co., Wild Horse, post office, 11 Aug 2009, T. M. Gilligan, det. T. M. Gilligan & D. J. Wright; ***Eucosma similiana***: TOR-DNA-

0513, OH: Hancock Co., 31 Aug 2009, T. M. Gilligan, det. T. M. Gilligan; ***Eucosma giganteana***: TOR-DNA-0574, MS: Oktibbeha Co., 24 Jun 2008, T. M. Gilligan, det. T. M. Gilligan; ***Eucosma conterminana***: TOR-DNA-0579, France: Mormoiron, 17 Jun 2009, T. M. Gilligan, det. J. Baixeras & T. M. Gilligan; ***Eucosma snyderana***: TOR-DNA-0634, Albany Co., WY, 2217 Sky View Lane, 22 Jun 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Eucosma conspiciendana***: TOR-DNA-0649, Albany Co., WY, just west of Gelatt Lake, 3 Jul 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Eucosma smithiana***: TOR-DNA-0653, Albany Co., WY, 2217 Sky View Lane, 6 Jul 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Eucosma canariana***: TOR-DNA-0664, Albany Co. west of Woods Landing, Hyw 130, 30 Jul 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Eucosma ragonoti***: TOR-DNA-0665, Albany Co., WY, 2217 Sky View Lane, 30 Jul 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Eucosma crambitana***: TOR-DNA-0667, Albany Co., WY, 2217 Sky View Lane, 4 Aug 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Eucosma totana***: TOR-DNA-0668, Albany Co. 1.1 miles west of Woods Landing, 5 Aug 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Eucosma fernaldana***: TOR-DNA-0675, Albany Co., WY, 2217 Sky View Lane, 18 Aug 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Eucosma optimana***: TOR-DNA-0680, Albany Co, just north of Rd 726, 22 Aug 2011 J. Nordin, det. J. Nordin & D. J. Wright; ***Eucosma robinsonana***: JD0726, FLA: Marion Co.: Ocala National Forest, 21 Jun 2006, J. J. Dombroskie, et al., det. J. J. Dombroskie; ***Phaneta griseocapitana***: DJW-06-0003, WY: Albany Co., 6 Jul 2006, D. J. Wright, det. D. J. Wright; ***Phaneta griseocapitana***: TOR-DNA-0657, Albany Co., WY, 2217 Sky View Lane, 18 Jul 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Phaneta pallidarcis***: DJW-06-0005, ID: Oneida Co., 9 Jul 2006, D. J. Wright, det. D. J. Wright; ***Phaneta castrensis***: DJW-06-0008, ID: Oneida Co., 15 Jul 2006, D. J. Wright, det. D. J. Wright; ***Phaneta ochrocephala***: TOR-06-0028, OH: Wyandot Co., Killdeer WLA, 16 Aug 2006, T. M. Gilligan, det. T. M. Gilligan; ***Phaneta tarandana*** (group): JD2117, Canada: AB: Kootenay Plains, near Whirlpool Point, 30 May 2007, J. J. Dombroskie, det. J. J. Dombroskie; ***Phaneta tarandana***, TOR-DNA-0685, Albany Co., WY, 2217 Sky View Lane, 24 Aug 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Phaneta umbrastriana***: TOR-DNA-0258, MS: Oktibbeha Co., 27 Jun 2008, T. M. Gilligan, det. T. M. Gilligan; ***Phaneta indagatricana***: TOR-DNA-467, CO: Lincoln Co., Hugo, hospital, 11 Aug 2009, T. M. Gilligan, det. T. M. Gilligan & D. J. Wright; ***Phaneta modicellana***: TOR-DNA-611, CO: Weld Co., Pawnee Nat'l Grassland, 28 Jun 2010, T. M. Gilligan & D. J. Wright, det. T. M. Gilligan & D. J. Wright; ***Phaneta salmicolorana***: TOR-DNA-0659, Albany Co., WY, 2217 Sky View Lane, 20 Jul 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Phaneta bucephaloides***: TOR-DNA-0669, Albany Co., WY, 2217 Sky View Lane, 6 Aug 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Phaneta offectalis***: TOR-DNA-0691, Albany Co., WY, 2217 Sky View Lane, 4 Sep 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Phaneta tenuiana***: TOR-DNA-0692, Albany Co. 1.1 miles west of Woods Landing, 10 Sep 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Phaneta montanana***: JD0848, Canada: AB: Jasper N. P., Opal Mountain, 2263m, 5 Jul 2006, J. J. Dombroskie & A. Rose, det. J. J. Dombroskie; ***Eucosma ainsliei***: TOR-DNA-0202, MT: Custer Co., 2.5 mi. S. Miles City, 12 Jul 2010, D. J. Wright, det. D. J. Wright; ***Pelochrista mediostriata***: TOR-DNA-0207, MT: Custer Co., 2.5 mi. S. Miles City, 12 Jul 2010, D. J. Wright, det. D. J. Wright; ***Pelochrista daemonicana***: TOR-DNA-0365, NM: Colfax Co., 5 Aug 2008, T. M. Gilligan, det. T. M. Gilligan; ***Pelochrista corosana***: TOR-DNA-401, CO: Cheyenne Co., Wild Horse, post office, 11 Aug 2009, T. M. Gilligan, det. T. M. Gilligan & D. J. Wright; ***Pelochrista scintillana***: TOR-DNA-498, IA: Adair Co., Rest Area, I-80 MM 80, 15 Aug 2009, T. M. Gilligan, det. T. M. Gilligan; ***Pelochrista rorana***: TOR-DNA-0575, CO: Cheyenne Co., 15 Sep 2007, T. M. Gilligan & C. E. Harp, det. D. J. Wright; ***Pelochrista rorana***: TOR-DNA-472 CO: Cheyenne Co., Kit Carson, church 11 Aug 2009 T. M. Gilligan det. T. M. Gilligan, D. J. Wright; ***Pelocrista popana***: TOR-DNA-0652, Albany Co., WY, 2217 Sky View Lane, 6 Jul 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Pelochrista argenteana***: TOR-DNA-0201 MT: Custer Co., 2.5 mi. S. Miles City 12 Jul 2010 D. J. Wright det. D. J. Wright; ***Pelochrista argenteana***: TOR-DNA-612, CO: Larimer Co., 12 mi.

NNW Ft. Collins, Owl Canyon, 29 Jun 2010, T. M. Gilligan & D. J. Wright, det. T. M. Gilligan & D. J. Wright; ***Pelocrista argenteana***: TOR-DNA-0658, Albany Co., WY, 2217 Sky View Lane, 19 Jul 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Pelochrista fuscosparsa***: TOR-DNA-0204, MT: Custer Co., 2.5 mi. S. Miles City, 12 Jul 2010, D. J. Wright, det. D. J. Wright; ***Pelocrista fuscosparsa***: TOR-DNA-0681, Albany Co., WY, 2217 Sky View Lane, 3 Jul 2011, J. Nordin, det. J. Nordin & D. J. Wright; ***Pelochrista mollitana***: TOR-DNA-0580, France: Sisteron, 14 Jun 2009, T. M. Gilligan, det. T. M. Gilligan.

APPENDIX III

Morphological Characters and Character States

FOREWING

1. Male forewing costal fold presence;
   - 0) absent
   - 1) present

2. Venation R4 and R5 divergence;
   - 0) separate
   - 1) connate
   - 2) stalked
   - 3) fused

HINDWING

3. Venation M3-CuA1 divergence;
   - 0) connate
   - 1) stalked

MALE GENITALIA

4. Uncus presence;
   - 0) absent
   - 1) present

5. Uncus shape;
   - 0) fingerlike
   - 1) spatulate
   - 2) hump-shaped, often with distal margin medially indented
   - 3) distally bifid process
   - 4) with antennalike processes

6. Uncus setation;
   - 0) sparse
   - 1) moderate
   - 2) dense
   - 3) long apical tuft

7. Socii shape;
   - 0) short, stubby
   - 1) fingerlike
   - 2) lobelike

8. Valva clasper presence;
   - 0) absent
   - 1) present

9. Valva process other than costal process from above basal excavation presence;
   - 0) absent
   - 1) present

10. Valva saccular angle;
   0) acute
   1) ≈ 90 º
   2) obtuse

11. Valva overlap of basoventral margin of cucullus on ventral margin of neck;
   0) none to weak
   1) moderate to extensive

12. Valval setae on margin of cucullus/ventral margin of neck type;
   0) filiform/standard
   1) 1 or few stout setae near distal end of neck
   2) 1 stout seta at apex of anal angle, sometimes with lesser setae on distal margin of cucullus
   3) series of stout setae on distal margin of cucullus, sometimes with 2 or 3 clustered at ventral apex

13. Valval neck with medial ridge presence;
   0) absent
   1) present

14. Cornuti presence;
   0) absent
   1) present

FEMALE GENITALIA

15. Papillae anales size relative to length of sternum 7 + 8;
   0) small
   1) medium
   2) large

16. Papillae anales orientation;
   0) facing laterally
   1) facing ventrally
   2) posterior lobes facing ventrally, anterior lobes facing laterally (inc. ventral extensions)

17. Papillae anales hook-tipped setae presence;
   0) absent
   1) present

18. Length of apophyses anteriores vs. length of apophyses posteriores;
   0) longer
   1) approximately equal
   2) shorter

19. Tergum 8 shape;
    - 0) narrow (medial width distinctly smaller than distance between bases of apophyses anteriores)
    - 1) of moderate width (medial width ≈ distance between bases of apophyses anteriores)
    - 2) broad (medial width distinctly larger than distance between bases of apophyses anteriores)

20. Sterigma connected to apophyses anteriores;
    - 0) no
    - 1) yes

21. Sterigma shape;
    - 0) not differentiated
    - 1) tube or funnel
    - 2) platelike
    - 3) raised rim
    - 4) projecting ventral lip

22. Sterigma, anterior margin of ostium structure;
    - 0) separated from sternum 7 by lamella antevaginalis and/or band of membrane
    - 1) fused with sternum 7

23. Sterigma, posterior margin sternum 7 shape;
    - 0) with U-shaped emargination, approximate to/fused laterally with sterigma
    - 1) concave, laterally diverging from sterigma
    - 2) straight to medially convex
    - 3) undifferentiated, sterigma entirely fused with sternum 7

24. Signa number;
    - 0) one
    - 1) two

25. Signa size;
    - 0) equal-sized
    - 1) of different sizes

26. Signa shape;
    - 0) scobinate
    - 1) horn-shaped
    - 2) thorn- or blade-shaped

27. Juncture of ductus bursae and ductus seminalis location;
    - 0) approximately midway between ostium and corpus bursae
    - 1) closer to ostium
    - 2) closer to corpus bursae

APPENDIX IV

Supplementary Trees

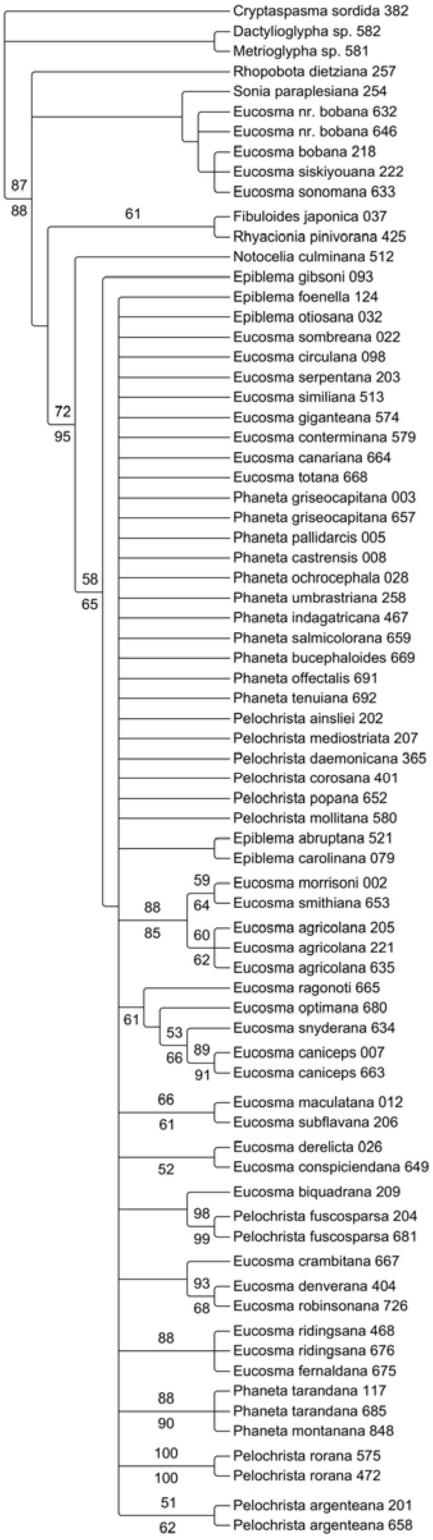


Figure 102. Parsimony strict consensus gene tree for 28S rDNA. Parsimony JK values are above each branch and likelihood BS values are below each branch.

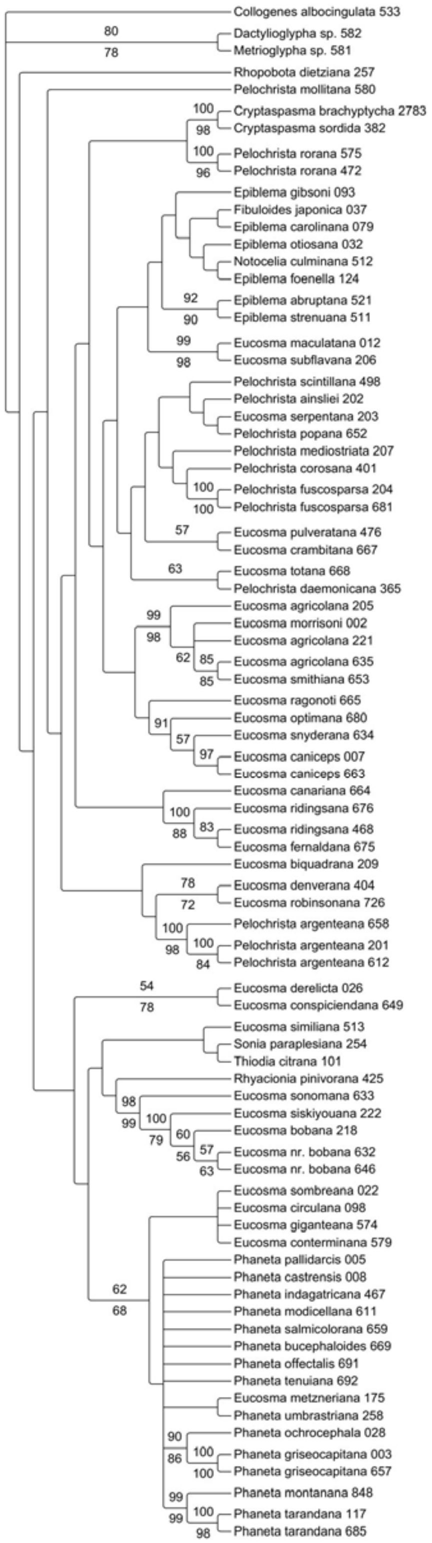


Figure 103. Parsimony strict consensus gene tree for COI. Parsimony JK values are above each branch and likelihood BS values are below each branch.

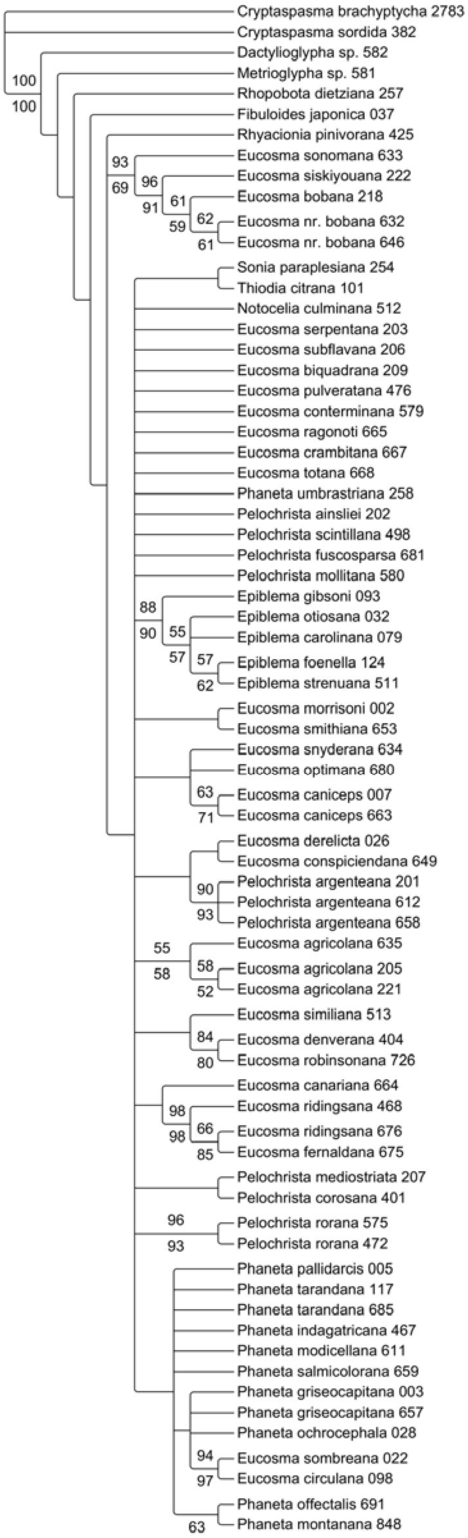


Figure 104. Parsimony strict consensus gene tree for EF-1a. Parsimony JK values are above each branch and likelihood BS values are below each branch.

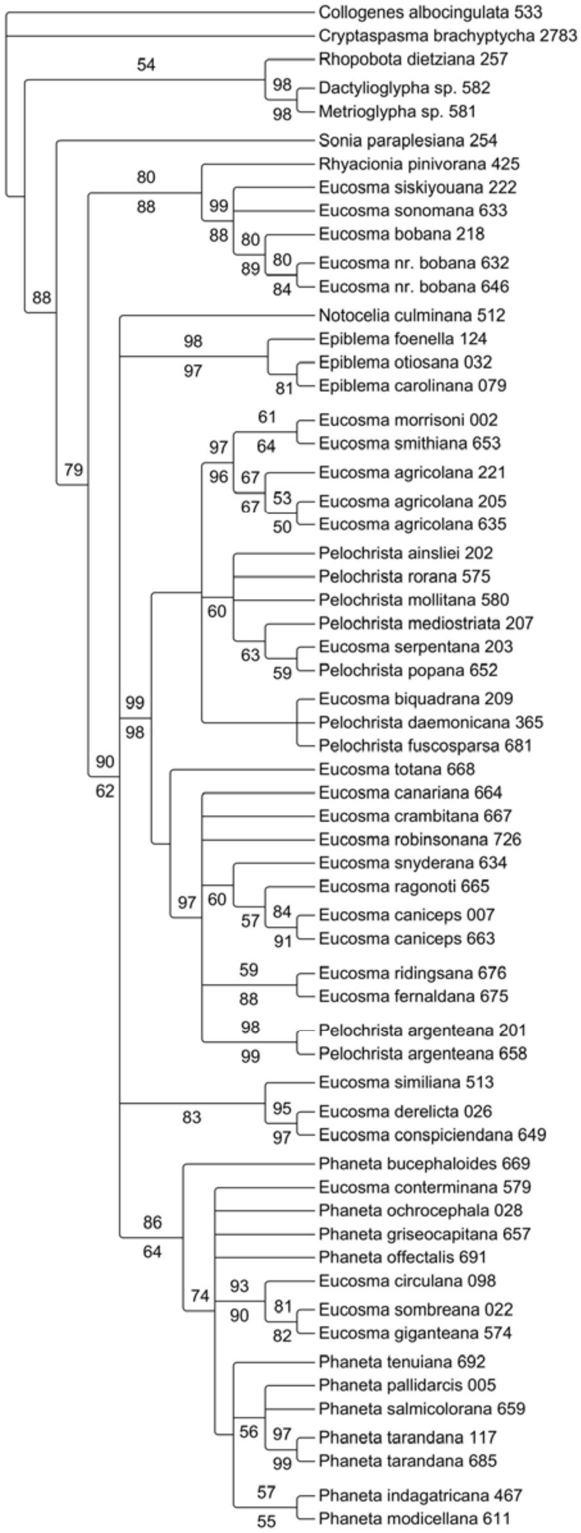


Figure 105. Parsimony strict consensus gene tree for CAD. Parsimony JK values are above each branch and likelihood BS values are below each branch.

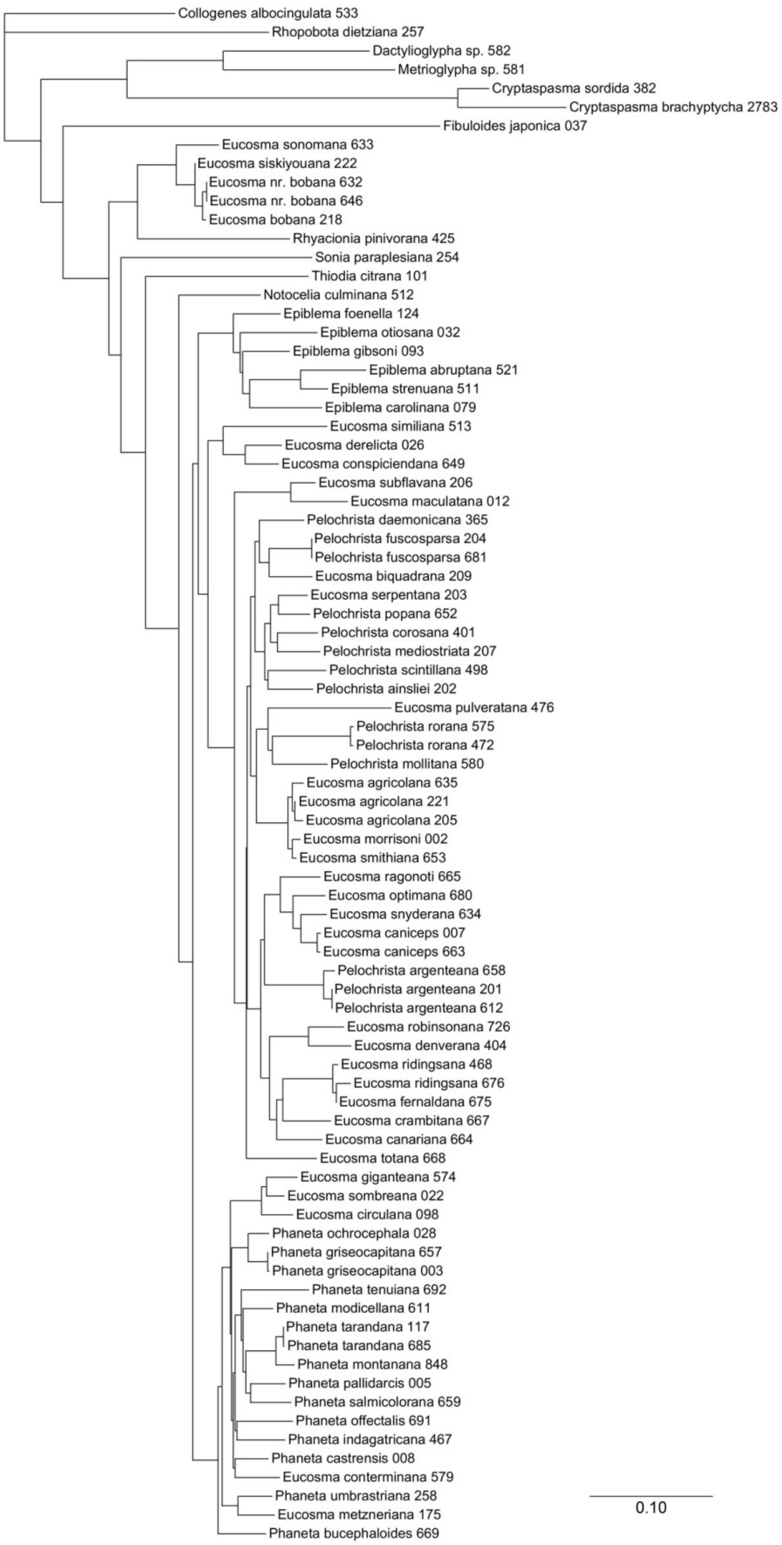


Figure 106. Combined molecular maximum likelihood tree showing relative branch lengths.

APPENDIX V

Revised World Catalogue of *Eucopina*, *Eucosma*, *Pelochrista*, and *Phaneta*

EUCOPINA

bobana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 26. USA: Colorado, Salida. Lectotype (USNM); female.

> ***antichroma*** Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 35. unnecessary replacement name for *bobana*; no type.

cocana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 26. USA: North Carolina, Tryon. Holotype (AMNH); male.

> ***rhodopaea*** Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 35. unnecessary replacement name for *cocana*; no type.

crymalana Powell 1968 (*Eucosma*); Hilgardia. 39: 27. USA: Arizona, Coconino Co., Grand Canyon, South Rim P.O. Holotype (CAS); male.

franclemonti Powell 1968 (*Eucosma*); Hilgardia. 39: 16. USA: Arizona, Coconino Co., Hochdoffer Hill. Holotype (CUIC); male.

gloriola Heinrich 1931 (*Eucosma*); Proc. ent. Soc. Wash. 33: 196. USA: Connecticut, Stamford, B.T.R. lab colony. Holotype (USNM); male.

monitorana Heinrich 1920 (*Eucosma*); Proc. U.S. natn. Mus. 57: 58. USA: Pennsylvania, Danville. Holotype (USNM); male.

monoensis Powell 1968 (*Eucosma*); Hilgardia. 39: 17. USA: California, Mono Co., Crooked Creek Lab., White Mountains, 3 airline mi N Inyo Co. line. Holotype (CAS); male.

ponderosa Powell 1968 (*Eucosma*); Hilgardia. 39: 13. USA: Oregon, Lake Co., Embody's Mill, near Silver Lake. Holotype (USNM); male.

rescissoriana Heinrich 1920 (*Eucosma*); Proc. U.S. natn. Mus. 57: 58. USA: Oregon, Sprague River. Holotype (USNM); male.

siskiyouana Kearfott 1907 (*Evetria*); Can. Ent. 39: 77. USA: California, Siskiyou Co. Lectotype (AMNH); male.

sonomana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 27. USA: California, Sonoma Co. Lectotype (AMNH); female.

tocullionana Heinrich 1920 (*Eucosma*); Proc. U.S. natn. Mus. 57: 59. USA: Connecticut, Lyme. Holotype (USNM); male.

EUCOSMA

abacana Erschoff 1877 (*Grapholitha*); Horae Soc. ent. Ross. 12: 342. Russia: Siberia, Irkutsk. Holotype (Unknown); female.

> *abascana* Kurentsov 1950 (*Semasia*); Soobshch. Dalnevost. Akad. Nauk SSSR: 30. misspelling of *abacana*; no type.

aemulana Schlager 1848 (*Grapholitha*); Ber. Lepid. Tauschver.: 38. Germany. Holotype (Unknown); unknown.

> latiorana Herrich-Schäffer 1851 (*Tortrix* (*Semasia*) *aemulata* var.); Syst. Bearbeitung Schmett. Eur. 4: 248. Germany. Syntype(s) (Unknown); unknown.

agnatana Christoph 1872 (*Grapholitha*); Horae Soc. ent. Ross. 9: 15. Russia. Syntype(s) (Unknown); unknown.

> *agnathana* Razowski 1971 (*Eucosma*); Acta zool. cracov. 17: 142. misspelling of *agnatana*; no type.

alatana McDunnough 1938 (*Thiodia*); Can. Ent. 70: 100. Canada: British Columbia, Kreuger Mt., Osoyoos. Holotype (CNC); male.

albarracina Hartig 1941 (*Eucosma*); Mitt. mnch. ent. Ges. 31: 156. Spain: Albarracin. Holotype (URIZ); male.

albertana McDunnough 1925 (*Thiodia*); Can. Ent. 57: 12. Canada: Alberta, Lethbridge. Holotype (CNC); male.

albicosta Falkovitsh 1964 (*Eucosma*); Trud. Zool. Inst. Leningrad 34: 279. Kazakhstan. Holotype (ZMAS); male.

albidulana Herrich-Schäffer 1851 (*Tortrix* (*Semasia*)); Syst. Bearbeitung Schmett. Eur. 4: 245. Germany: Ofen. Syntype(s) (Unknown); unknown.

> *albidulata* Drenovski 1925 (*Epiblema*); Z. Wiss. Insektenbiol 20: 559. misspelling of *albidulana*; no type.
>
> *albidulana* junior Herrich-Schäffer 1848 (unimonial); Syst. Bearbeitung Schmett. Eur. 4: pl. 44, fig. 310. nomen nudum; no type.

albuneana Zeller 1847 (*Paedisca*); Isis von Oken (Leipzig) 1847 (10): 725. ? (Syracuse). Syntype(s) (Unknown); unknown.

> *albuncana* Lederer 1859 (*Tortrix*) Wien. ent. Monatschr. 3: 332. misspelling of *albuneana*; no type.

altana McDunnough 1927 (*Thiodia*); Can. Ent. 59: 272. Canada: British Columbia, Alta Lake. Holotype (CNC); male.

ambodaidaleia Miller 1983 (*Phaneta*); Ann. ent. Soc. Am. 76: 101. USA: South Carolina, Berkeley Co., Wedge Plantation. Holotype (USNM); female.

amellana Preissecker 1930 (*Semasia*); Verh. zool.-bot. Ges. Wien 80: 20. Austria. Holotype (Unknown); unknown.

amphorana Walsingham 1879 (*Semasia*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 63. USA: Oregon, John Day River, Camp Watson. Syntypes (4) (MCZ); unknown.

annetteana Kearfott 1907 (*Thiodia*); Trans. Am. ent. Soc. 33: 42. USA: Ohio, Cincinnati. Lectotype (AMNH); male.

apacheana Walsingham 1884 (*Semasia*); Trans. ent. Soc. Lond. 1884: 143. USA: Arizona. Holotype (BMNH); male.

apocrypha Falkovitsh 1964 (*Eucosma* (*Phaneta*)); Trud. Zool. Inst. Leningrad 34: 280. Kazakhstan. Holotype (ZMAS); male.

> *tagarica* Falkovitsh 1964 (*Eucosma* (*Phaneta*) *apocrypha* ssp.); Trud. Zool. Inst. Leningrad 34: 282. Russia: Siberia. Holotype (ZMAS); male.

apocryphoides Budashkin 2009 (*Eucosma*); Proc. Zoo. Mus. Kiev Taras Shevchenko Nat. U. (Esse) 5: 29. Ukraine Eastern Crimea, Karadagh Nature Reserve. Holotype (ZMKU); male.

apriliana Grote 1877 (*Exentera*); Can. Ent. 9: 227. USA: New York, Albany. Syntypes (USNM); male, female.

arenana Wright 2010 (*Phaneta*); J. Lepid. Soc. 64: 120. USA: Utah, Emery Co., Goblin Valley Rd. Holotype (USNM); male.

argenticostana Walsingham 1879 (*Semasia*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 61. USA: northern Oregon. Holotype (BMNH); unknown.

argentifera Razowski 1972 (*Eucosma*); Acta zool. cracov. 17: 138. Mongolia: Barhany, Bala Suka. Holotype (TMB); female.

argutipunctana Blanchard & Knudson 1983 (*Phaneta*); J. Lepid. Soc. 37: 143. USA: Texas, Hemphill Co., Canadian. Holotype (USNM); male.

artemisiana Walsingham 1879 (*Semasia*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 56. USA: California, Mount Shasta. Holotype (BMNH); male.

aspidiscana Hübner [1814-1817] (*Tortrix*); Samml. Eur. Schmett. 7: pl. 41, fig. 256. Europe. Syntypes(s) (Unknown); unknown.

> *obscurana* Fabricius 1798 (?*Pyralis*); Suppl. Entomologiae Systematicae: 476. France. Syntype(s) (Unknown); unknown.
>
> *rubescana* Constant 1895 (*Grapholitha aspidiscana* var.); Annls Soc. ent. Fr. 1895 (Bulletin): LI. France. Syntype(s) (Unknown); unknown.

dahlbomiana Zetterstedt 1839 (*Phoxopteris*); Insecta Lapponica Descripta: 987. Sweden: Lapponia. Syntype(s) (UZIL); unknown.

aspidana Frölich 1828 (Tortrix); Enum. Tortr. Wrtemberg: 96. misspelling of *aspidiscana*; no type.

zachana Hübner [1814-1817] (Tortrix); Samml. Eur. Schmett. 7: pl. 38, fig. 243. Syntype(s) (Unknown); unknown.

aurantiradix Kuznetzov 1962 (*Eucosma* (*Catoptria*)); Bull. Soc. ent. Mulhouse 1962: 55. Russia: Far East, Amur Region, Klimout-sy, 40 km W Sovobodnyy. Holotype (ZMAS); unknown.

autumnana McDunnough 1942 (*Thiodia*); Can. Ent. 74: 66. Canada: Quebec, Lac-Ste-Marie. Holotype (CNC); male.

awemeana Kearfott 1907 (*Thiodia*); Trans. Am. ent. Soc. 33: 41. Canada: Manitoba, Aweme. Lectotype (AMNH); male.

balatonana Osthelder 1937 (*Epiblema expallidana* ssp.); Dt. ent. Z. Iris 51: 109. Hungary. Holotype (Unknown); unknown.

danicana Schantz 1962 (*Eucosma*); Notulae Ent. 42: 5. Denmark. Holotype (Unknown); unknown.

afflicta Falkovitsh 1964 (*Eucosma*); Trud. Zool. Inst. Leningrad 34: 277. Kazakhstan: Vaganovo. Holotype (ZMAS); male.

baloghi Wright 2010 (*Phaneta*); J. Lepid. Soc. 64: 135. USA: New Mexico, Otero Co., vic. Holloman Lakes. Holotype (USNM); male.

benjamini Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 66. USA: Utah, Vineyard. Holotype (USNM); male.

bilineana Kearfott 1907 (*Eucosma*); Can. Ent. 39: 54. USA: Illinois. Lectotype (AMNH); female.

bipunctella Walker 1863 (*Affa*); List Specimens lepid. Insects Colln. Br. Mus 27: 202. North America. Holotype (BMNH); male.

worthingtoniana Fernald 1878 (*Paedisca*); Can. Ent. 10: 83. USA: Northern Illinois. Lectotype (USNM); male.

brachysticta Meyrick, in Caradja & Meyrick 1935 (*Eucosma*); Mat. Microlepid. Fauna Chin. Prov.: 55. China: Kiangsu Pro-vince, Lungtan, near Nanking. Lectotype (MGAB); male.

browni Wright 2010 (*Phaneta*); J. Lepid. Soc. 64: 119. USA: Colorado, Chaffee Co., 4 mi N Buena Vista, Co. Rd. 375. Holotype (USNM); male.

bucephaloides Walsingham 1891 (*Semasia*); Insect Life 3: 465. USA: California, Siskiyou Co., Little Shasta. Holotype (BMNH); female.

caliacrana Caradja 1931 (*Semasia luciana* ssp.); Memle Sect. Stiint. Acad. roum. (3) 7 (8): 37. Romania: Balcic, Val. Ag-Bunar. Holotype (MGAB); female.

galactic Obraztsov 1968 (*Eucosma* (*Phaneta*)); Tijdschr. Ent. 111: 247. replacement name for *luciana*; no type.

nessebarana Soffner 1962 (*Semasia*); Dt. ent. Z. (N.F.) 9: 140. Bulgaria. Holotype (Unknown); unknown.

luciana Kennel 1919 (*Semasia*); Mitt. mnch. ent. Ges. 8: 81. Russia: Siberia. (preoccupied) Holotype (MNHU); unknown.

camdenana McDunnough 1925 (*Thiodia*); Can. Ent. 57: 17. USA: Alaska, Camden Bay, W Kanganevik. Holotype (CNC); male.

campoliliana [Denis & Schiffermüller] 1775 (*Tortrix*); Syst. Verz. Schmett. Wienergegend :318. Austria. Syntype(s) (Unknown); unknown.

nigromaculata Wood 1839 (*Anticlea*); Index Ent. (3):144 fig. 962. misspelling of *nigromaculana*; no type.

nigrimaculana Barrett 1874 (*Calosetia*); Ent. mon. Mag. 11: 134. misspelling of *nigromaculana*; no type.

capitinivana Bruand 1850 (*Grapholitha*); Mm. Soc. Emul. Doubs. (1) 3: 91. France. Syntype(s) (Unknown); unknown.

freyeriana Fischer von Roslerstamm 1839 (*Grapholitha*); Abbild. Berich. Ergnz Schmett.-Kunde 1: 132. Germany. Syntype(s) (Unknown); unknown.

ussuriana Caradja 1916 (*Steganoptycha nigromaculana* var.); Dt. ent. Z. Iris 30: 60. Russia Far East, Primorsky Krai, Kazakevich. Lectotype (MGAB); male.

albana Haworth 1811 (*Tortrix*); Lepid. Br. (3): 436. United Kingdom: Great Britain. Syntype(s) (BMNH); unknown.

nigromaculana Haworth 1811 (*Tortrix*); Lepid. Br. (3): 436. United Kingdom: Great Britain. Syntype(s) (BMNH); unknown.

cana Haworth 1811 (*Tortrix*); Lepid. Br. (3): 456. United Kingdom: Great Britain. Syntype(s) (BMNH); unknown.

carduana Guenée 1845 (*Catoptria*); Annls Soc. ent. Fr. (2) 3: 188. France. Syntype(s) (MNHN); unknown.

subvittana Staudinger 1892 (*Grapholitha cana* var.); Dt. ent. Z. Iris 5: 299. Armenia: Kasicoparan. Syntype(s) (MNHU); unknown.

marmorana Fabricius 1798 (?*Pyralis*); Suppl. Entomologiae Systematicae: 477. Germany: Saxonia. Syntype(s) (Unknown); unknown.

> *monetulana* Hübner [1814-1817] (*Tortrix*); Samml. Eur. Schmett. 7: pl. 41, fig. 257. Syntype(s) (Unknown); unknown.

canusana Wright, in Wright, Brown & Gibson 1997 (*Phaneta*); J. Lepid. Soc. 51: 122. USA: Ohio, Adams Co., Lynx Prairie Preserve. Holotype (USNM); male.

castrensis McDunnough 1929 (*Thiodia*); Can. Ent. 61: 271. USA: Wyoming, Yellowstone National Park, Camp Roosevelt. Holotype (CNC); male.

catharaspis Meyrick 1922 (*Ancylis*); Exotic Microlepid. 2: 514. China: Zikawy. Holotype (MNHN); female.

> *cataraspis* Razowski 1960 (*Eucosma*); Polskie Pismo Ent. 30: 385. misspelling of *catharaspis*; no type.

catoptrana Rebel 1903 (*Semasia*); Verh. zool.-bot. Ges. Wien 53: 91. Austria: Schneeberg. Holotype (NHMV); unknown.

> *heringiana* Jackh 1953 (*Catoptria* (*Semasia*)); Z. Lepid. 3: 43. Germany: Wangerooge, Watt, Wiese. Holotype (USNM); male (lost).

certana Kuznetzov 1967 (*Eucosma* (*Phaneta*)); Trud. Zool. Inst. Leningrad 41: 68. Russia: Far East. Holotype (ZMAS); male.

cetratana Kennel 1901 (*Semasia*); Dt. ent. Z. Iris 13 (1900): 264. Europe. Lectotype (MNHU); male.

chrysyphis Razowski 1972 (*Eucosma*); Acta zool. cracov. 17: 144. Mongolia: Bulgan Province, 25 km E Somon Lun. Holotype (TMB); male.

cibolana Wright 2010 (*Phaneta*); J. Lepid. Soc. 64: 125. USA: New Mexico, Cibola Co., Cibola Nat. For., Lobo Canyon Picnic Area. Holotype (USNM); male.

circulana Hübner 1823 (*Tortrix*); Zutr. Samml. exot. Schmett. 2: 28. USA: Pennsylvania. Syntype(s) (Unknown); unknown.

citricolorana McDunnough 1942 (*Thiodia*); Can. Ent. 74: 66. Canada: Saskatchewan, Saskatoon. Holotype (CNC); male.

clarescens Kuznetzov 1964 (*Eucosma* (*Phaneta*)); Trud. Zool. Inst. Leningrad 34: 264. Kazakhstan. Holotype (ZMAS); male.

clarkei Blanchard & Knudson 1983 (*Phaneta*); Proc. ent. Soc. Wash. 85: 847. USA: Texas, Hemphill Co., Canadian National Grassland, Lake Marvin. Holotype (USNM); male.

clavana Fernald 1882 (*Semasia*); Trans. Am. ent. Soc. 10: 72. USA: Massachusetts, Truro. Lectotype (USNM); male.

clementeana Wright 2010 (*Phaneta*); J. Lepid. Soc. 64: 135. USA: California, Los Angeles Co., San Clemente Island, Wilson Cove. Holotype (EME); male.

coagulana Kennel 1901 (*Epiblema*); Dt. ent. Z. Iris 13(1900): 278. Armenia: Kasikoparan. Lectotype (MNHU); female.

columbiana Walsingham 1879 (*Semasia*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 57. USA: Oregon, near Columbia River. Holotype (BMNH); male.

complicana McDunnough 1925 (*Thiodia*); Can. Ent. 57: 16. Canada: British Columbia, Osoyoos. Holotype (CNC); male.

conformana Mann 1872 (*Grapholitha*); Verh. zool.-bot. Ges. Wien 22: 36. Croatia: Dalmatia, Ragusa. Holotype (NHMV); unknown.

> *significantana* Kennel 1901 (*Epiblema*); Dt. ent. Z. Iris (1900) 13: 282. Macedonia. Syntype(s) (MNHU); unknown.
>
> *umbratana* Staudinger 1880 (*Grapholitha*); Horae Soc. ent. Ross. 15(1879): 253. Turkey: Amasia. Lectotype (MNHU); male.

confunda Kuznetzov 1966 (*Eucosma* (*Eucosma*)); Trud. Zool. Inst. Leningrad 37: 192. Russia: Far East, Primorsky Krai, vic Vladivostok. Holotype (ZMAS); male.

conterminana Guenée 1845 (*Catoptria*); Annls Soc. ent. Fr. (2) 3: 189. France. Holotype (MNHN); unknown.

convergana McDunnough 1925 (*Thiodia*); Can. Ent. 57: 15. Canada: Manitoba, Aweme. Holotype (CNC); male.

corculana Zeller 1874 (*Semasia*); Verh. zool.-bot. Ges. Wien 24: 433. Canada: British Columbia, Vancouver Island. Holotype (BMNH); female.

crassana McDunnough 1938 (*Thiodia*); Can. Ent. 70: 99. Canada: British Columbia, Kreuger Mt., Osoyoos. Holotype (CNC); male.

cretaceana Kennel 1899 (*Grapholitha*); Dt. ent. Z. Iris 11: 42. Spain: Albarracin. Holotype (MNCNM); male.

cruentana Blanchard & Knudson 1982 (*Phaneta*); J. Lepid. Soc. 35 (1981): 169. USA: Texas, Anderson Co., Engeling Wildlife Management Area, near Tennessee Colony. Holotype (USNM); male.

culmana Muller-Rutz 1932 (*Epiblema*); Mitt. schweiz. ent. Ges. 15: 198. Switzerland: Graubundner Alps. Holotype (NHMB); unknown.

cumulana Guenée 1845 (*Catoptria*); Annls Soc. ent. Fr. (2) 3: 189. France. Syntype(s) (MNHN); unknown.

> *obscurana* Caradja 1916 (*Epiblema cumulana* var.); Dt. ent. Z. Iris 30: 64. Spain: Cuenca. (preoccupied) Lectotype (MGAB); male.
>
> *caradjai* Obraztsov 1968 (*Eucosma cumulana* ssp.); Tijdschr. Ent. 111: 10. replacement name for *obscurana*; no type.

hornigiana Lederer 1855 (*Grapholitha*); Verh. zool.-bot. Ges. Wien: 77. Hungary: Ofen. Syntype(s) (MNHU); unknown.

decempunctana Walsingham 1879 (*Semasia*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 58. USA: Oregon, The Dalles. Holotype (BMNH); unknown.

denigratana Kennel 1901 (*Epiblema*); Dt. ent. Z. Iris (1900) 13: 281. Russia: Far East, Primorsky K, Sutschan. Holotype (MNHU); male.

diakonoffi Gibeaux 1984 (*Eucosma*); Ent. Gall. 1(3): 155. France: Hautes-Alpes. Holotype (CGAC); male.

discernata Kuznetzov 1966 (*Eucosma* (*Eucosma*)); Trud. Zool. Inst. Leningrad 37: 194. Russia: Far East, Primorsky Krai, Yakovlevka. Holotype (ZMAS); female.

dorsiatomana Kearfott 1905 (*Thiodia*); Can. Ent. 37: 209. Canada Saskatchewan, Regina. Lectotype (AMNH); male.

elongana Walsingham 1879 (*Semasia*?); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 56. USA: northern Oregon. Holotype (BMNH); unknown.

ephedrana Christoph 1877 (*Grapholitha*); Horae Soc. ent. Ross 12: 291. Iran: Schahrud. Syntype(s) (ZMAS); unknown.

essexana Kearfott 1907 (*Thiodia*); Trans. Am. ent. Soc. 33: 39. USA: New Jersey, Caldwell. Lectotype (AMNH); male.

explicatana Kennel 1900 (*Steganoptycha*); Dt. ent. Z. Iris 13: 141. Russia: Ural [Guberli]. Holotype (MNHU); female.

fasciculatana McDunnough 1938 (*Thiodia*); Can. Ent. 70: 96. Canada: British Columbia, Shingle Creek, Penticton. Holotype (CNC); male.

ferruginana Fernald 1882 (*Semasia*); Trans. Am. ent. Soc. 10: 72. USA: Massachusetts, Goodell. Lectotype (USNM); female.

fertoriana Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 264. Canada: British Columbia, Goldstream. Holotype (USNM); male.

fervidana Zeller 1847 (*Paedisca*); Isis von Oken (Leipzig) 1847 (10): 730. ? (Syracuse). Holotype (BMNH); unknown.

festivana Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 265. Canada: Manitoba, Aweme. Holotype (AMNH); male.

flavispecula Kuznetzov 1964 (*Eucosma*); Trud. Zool. Inst. Leningrad 34: 260. Russia: Far East, Primorsky Krai. Holotype (ZMAS); female.

floridana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 21. USA: Florida, Hastings. Lectotype (AMNH); male.

formosana Clemens 1860 (*Ioplocama*); Proc. Acad. Nat. Sci. Philad. 12: 360. USA: Illinois. Lectotype (ANSP); female.

> ***sagittana*** Walker 1863 (*Grapholitha*); List Specimens lepid. Insects Colln. Br. Mus 28: 368. Canada: Nova Scotia. Holotype (BMNH); unknown.

> ***stercoreana*** Zeller 1875 (*Grapholitha*); Verh. zool.-bot. Ges. Wien 25: 290. USA: Massachusetts or Maine. Holotype (BMNH); unknown.

fraudulentana Kennel 1901 (*Semasia*); Dt. ent. Z. Iris (1900) 13: 265. Kazakhstan: Caucasus. Holotype (MNHU); unknown.

fuscida Kuznetzov 1966 (*Eucosma*); Trud. Zool. Inst. Leningrad 37: 191. Russia: Far East, Amur Region, Klimoutsy, 40 km W Svobodnyy. Holotype (ZMAS); male.

gemellana Heinrich 1923 (*Eucosma circulana* ssp.); Bull. U.S. natn. Mus. 123: 96. USA: Florida, St. Petersburg. Holotype (USNM); male.

getonia Razowski 1972 (*Eucosma*); Acta zool. cracov. 17: 140. Mongolia: Mongolia Central [Tov] Province, 25 km E Somon Lun. Holotype (TMB); male.

giganteana Riley 1881 (*Paedisca*); Trans. St. Louis Acad. Sci 4: 318. USA: Kansas and Iowa. Syntype(s) (USNM); unknown (lost).

> *minorata* Heinrich 1924 (*Eucosma giganteana* ssp.); J. Wash. Acad. Sci. 14: 388. USA: Texas, Liberty Co., Liberty. Holotype (USNM); male.

glebana Snellen 1883 (*Grapholitha*); Tijdschr. Ent. 26: 206. Russia: Far East. Lectotype (RMNH); male.

> *sinensis* Walsingham 1900 (*Thiodia*); Ann. Mag. nat. Hist. (7) 6: 406. China: Chekiang, Ningpo. Holotype (BMNH); male.

glomerana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 49. USA: Texas. Holotype (BMNH); unknown.

> *sandana* Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 22. USA: Colorado, Chimney Gulch. Lectotype (AMNH); male.

> *griphodes* Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 34. unnecessary replacement name for *sandana*; no type.

goblinana Wright 2010 (*Phaneta*); J. Lepid. Soc. 64: 124. USA: Utah, Emery Co., Rt. 24 & Goblin Valley Rd. Holotype (USNM); male.

gonzalezalvarezi Agenjo 1970 (*Eucosma*); Eos 45 (1969): 9. Spain: Madrid Province, Monte de Regajal, Aranguez. Holotype (MNCNM); male.

gorodkovi Kuznetzov 1979 (*Eucosma*); Trud. Zool. Inst. Leningrad 81: 85. Russia: Far East. Holotype (ZMAS); female.

gradensis Galvagni 1909 (*Semasia*); Mitt. Naturwiss. Ver. Wien. Univ 7: 248. Croatia: Grado. Holotype (Unknown); unknown.

granulatana Kearfott 1908 (*Cydia*); J. New York ent. Soc. 16: 173. USA: Colorado, Denver. Lectotype (AMNH); male.

grindeliana Busck 1906 (*Cydia*); Can. Ent. 38: 211. USA: Texas, Clarendon. Syntypes (USNM); male, female.

griseocapitana Walsingham 1879 (*Semasia*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 61. USA: California, Mount Shasta. Syntypes (BMNH), (AMNH); unknown.

gypsatana Kennel 1921 (*Epiblema*); Palaear. Tortr.: 557. Turkey: Anatolia, Konia. Holotype (MGAB); female.

haberhaueri Kennel 1901 (*Epiblema*); Dt. ent. Z. Iris (1900) 13: 280. Uzbekistan: Namangan. Lectotype (MNHU); male.

halophilana Budashkin 2009 (*Eucosma*); Proc. Zoo. Mus. Kiev Taras Shevchenko Nat. U. (Esse) 5: 32. Ukraine Eastern Crimea, Karadagh Nature Reserve. Holotype (ZMKU); male.

haydenae Wright 2006 (*Eucosma*); J. Lepid. Soc. 60: 162. USA: Iowa, Howard Co., Hayden Prairie. Holotype (USNM); male.

hebescana Kuznetzov 1986 (*Eucosma*); Vestnik Zool. 1986 (4): 23. Uzbekistan: Samarkand. Holotype (MNHU); male.

> *hebesana* Razowski 1999 (*Eucosma*); SHILAP Revta. Lepid. 27: 474. misspelling of *hebescana*; no type.

hodgesi Wright & Gilligan 2010 (*Phaneta*); J. Lepid. Soc. 64: 107. USA: Colorado, Alamosa Co., Zapata ranch. Holotype (USNM); male.

hohenwartiana [Denis & Schiffermüller] 1775 (*Tortrix*); Syst. Verz. Schmett. Wienergegend:129. Austria. Syntype(s) (Unknown); unknown (lost).

> *fulvana* Stephens 1834 (*Carpocapsa* (*Epiblema*)); Illust. Br. Ent. (Haustellata) 4: 123. United Kingdom: Great Britain, London. Lectotype (BMNH); female.
>
> *fulvana* Stephens 1829 (*Carpocapsa* (*Epiblema*)); Syst. Cat. Br. Insects (2): 180. United Kingdom: Great Britain. nomen nudum; no type.
>
> *cervana* Scopoli 1763 (?*Phalaena*); Ent. Carn.: 234. Slovenia: Carniola [Slovenia]. Syntype(s) (Unknown); unknown.
>
> *strigana* Curtis 1831 (*Carpocapsa*); Br. Ent. 6: folio 352. United Kingdom: Great Britain. Syntype(s) (DEMV); unknown.
>
> *hohenwarthiana* Treitschke 1829 (*Grapholitha*); Schmett. Eur. 7: 232. misspelling of *hohenwartiana*; no type.

decipiana Laharpe 1858 (*Semasia*); Nouv. Mm. Soc. Helv. Sci. Nat. 16: 93. Switzerland. Syntype(s) (MZL); unknown.

scopoliana Haworth 1811 (*Tortrix*); Lepid. Br. (3): 456. United Kingdom: Great Britain. (preoccupied) Syntype(s) (BMNH); unknown.

pupillana Hübner [1796-1799] (*Tortrix*); Samml. Eur. Schmett. 7: pl. 4, fig. 20. (preoccupied) Syntype(s) (Unknown); unknown.

jaceana Herrich-Schäffer 1851 (*Tortrix* (*Semasia*)); Syst. Bearbeitung Schmett. Eur. 4: 248. Germany. Syntype(s) (Unknown); unknown.

ignotana Caradja 1916 (*Semasia*); Dt. ent. Z. Iris 30: 64. Russia: Siberia, Kasakewitsch [Kazakevich]. Lectotype (MGAB); male.

implicata Heinrich 1931 (*Thiodia*); Proc. U.S. natn. Mus. 79: 7. USA: Washington, Rochester. Holotype (USNM); male.

incinerana Constant 1888 (*Grapholitha*); Annls Soc. ent. Fr. (6) 8: 167. France: Alpes Maritimes. Holotype (MNHN); female.

indagatricana Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 56. USA: Utah, Utah Co., Provo. Holotype (USNM); male.

verecundana Blanchard 1980 (*Phaneta*); J. Lepid. Soc. 33 (1979): 210. USA: Texas, Hemphill Co., Canadian. Holotype (USNM); male.

indeterminana McDunnough 1925 (*Thiodia*); Can. Ent. 57: 13. Canada: Quebec, Aylmer. Holotype (CNC); male.

individiosana Kennel 1901 (*Epiblema*); Dt. ent. Z. Iris (1900) 13: 278. Algeria. Holotype (MNHU); unknown.

infimbriana Dyar 1904 (*Thiodia*); Proc. U.S. natn. Mus. 27: 927. Canada: British Columbia, Kaslo. Holotype (USNM); female.

candidula Heinrich 1924 (*Thiodia infimbriana* ssp.); J. Wash. Acad. Sci. 14: 387. Canada: Manitoba, Aweme. Holotype (CNC); male.

influana Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 49. USA: California, Siskiyou Co., Shasta Retreat. Holotype (USNM); male.

insignata Heinrich 1924 (*Thiodia*); J. Wash. Acad. Sci. 14: 386. USA: Colorado, San Juan Co., Silverton. Holotype (USNM); male.

intermediana Kennel 1900 (*Semasia*); Dt. ent. Z. Iris 13: 146. Russia: Far East, Amur. Holotype (MNHU); male.

kiscana Kearfott 1905 (*Thiodia*); Trans. Am. ent. Soc. 33: 92. USA: New Jersey, Greenwood Lake. Lectotype (AMNH); male.

> ***speculigera*** Meyrick 1912 (*Thiodia*); Ent. mon. Mag. 48: 35. unnecessary replacement name for *kiscana*; no type.

kokana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 29. USA: Ohio, Cincinnati. Lectotype (AMNH); female.

> ***chortaea*** Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 35. unnecessary replacement name for *kokana*; no type.

> ***sororiana*** Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 263. Canada: Manitoba, Aweme. Holotype (CNC); male.

krygeri Rebel 1937 (*Semasia*); Z. st. Ent. Verz. 22: 42. Denmark: Bramsnus. Holotype (Unknown); unknown.

labiata Wright 2010 (*Phaneta*); J. Lepid. Soc. 64: 121. USA: Texas, Cottle Co., Paducah. Holotype (USNM); male.

lacteana Treitschke 1835 (*Grapholitha*); Schmett. Eur. 10: 113. Hungary. Lectotype (TMB); male.

> *maritime* Humphreys & Westwood 1845 (*Carpocapsa*); Brit. Moths Transf. 2: 138. United Kingdom: Great Britain. Syntype(s) (BMNH); unknown.

> *caudidulana* Obraztsov 1968 (*Eucosma*); Tijdschr. Ent 111: 15. misspelling of *candidulana*; no type.

> *candidulana* Nolcken 1870 (*Grapholitha*); Arb. Naturf.-Ver. Riga 3: 413. Europe. Syntype(s) (Unknown); unknown.

landana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 18. Canada: Manitoba, Rounthwaite. Lectotype (AMNH); female.

> *isospora* Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 34. unnecessary replacement name for *landana*; no type.

lapidana Walsingham 1879 (*Semasia*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 58. USA: Oregon, Crooked River near Klamath Lake. Holotype (BMNH); unknown.

> ***lepidana*** Heinrich 1929 (Thiodia); Proc. U.S. natn. Mus. 75: 2. misspelling of *lapidana*; no type.

latens Heinrich 1929 (*Thiodia*); Proc. U.S. natn. Mus. 75: 2. USA: California, Tulare Co., Monachee Meadows. Holotype (USNM); male.

lignana Snellen 1883 (*Grapholitha* (*Semasia*)); Tijdschr. Ent. 26: 235. Russia: Siberia, Irkutsk Syntype(s) (RMNH); unknown.

> *malitiosana* Kennel 1901 (*Semasia*); Dt. ent. Z. Iris (1900) 13: 266. Mongolia: Zavhan Province, Uliassutai. Lectotype (MNHU); male.

elongata Filipjev 1924 (*Semasia*); Jahbr. Martijanova Staatsmus. Minussinsk 2: 24. Russia: Siberia. Syntype(s) (ZMAS); male.

linitipunctana Blanchard & Knudson 1983 (*Phaneta*); J. Lepid. Soc. 37: 140. USA: Texas, Nueces Co., North Padre Island. Holotype (USNM); male.

lyrana Snellen 1883 (*Grapholitha* (*Semasia*)); Tijdschr. Ent. 26: 208. Russia: Siberia. Syntype(s) (RMNH); unknown (lost).

magnana Kuznetzov 1978 (*Eucosma*); Opred. Nasekomykh Evop. SSSR 4: 531. Kyrgyzstan: Alai Mountains. Holotype (ZMAS); unknown.

marmontana Kearfott 1907 (*Proteopteryx*); Can. Ent. 39: 155. Canada: Manitoba, Rounthwaite. Lectotype (AMNH); male.

mayelisana Blanchard 1979 (*Phaneta*); J. Lepid. Soc. 33: 209. USA: Texas, Cottle Co., Paducah. Holotype (USNM); male.

messingiana Fischer von Roslerstamm 1837 (*Grapholitha*); Abbild. Berich. Ergnz Schmett.-Kunde 1: 89. Germany: Neustrelitz. Syntype(s) (Unknown); unknown.

metana Kennel 1919 (*Semasia*); Mitt. mnch. ent. Ges. 8 (1917-1918): 84. China: Shanghai. Holotype (MNHU); male.

metzneriana Treitschke 1830 (*Conchylis*); Schmett. Eur. 8: 277. Hungary. Lectotype (TMB); female.

joannisiola Dufrane 1955 (*Eucosma metzneriana* ssp.); Bull. Inst. R. Sci. Nat. Belg. 31 (33): 3. China. Holotype (IRSN); unknown.

osthelderi Dufrane 1955 (*Eucosma metzneriana* form); Bull. Inst. R. Sci. Nat. Belg. 31(33): 3. Belgium. Holotype (IRSN); unknown.

anserana Heinemann 1863 (*Grapholitha* (*Semasia*)); Schmett. Deut. Schweiz 2: 169. Hungary. Syntype(s) (Unknown); unknown.

intacta Walsingham 1900 (*Thiodia*); Ann. Mag. nat. Hist. (7) 6: 405. Japan. Holotype (BMNH); male.

migratana Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 53. USA: California, Inyo Co., Olanche. Holotype (USNM); male.

minimana Walsingham 1879 (*Semasia*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 60. USA: California, Siskiyou Co. Holotype (BMNH); unknown.

mirificana Peyerimhoff 1877 (*Grapholitha*); Annls Soc. ent. Fr. (5) 6 (1876): 590. France. Syntype(s) (Unknown); unknown.

misturana Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 54. Canada: Saskatchewan, Oxbow. Holotype (USNM); male.

modernana McDunnough 1925 (*Thiodia*); Can. Ent. 57: 13. Canada: Quebec, Aylmer. Holotype (CNC); male.

modicellana Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 265. Canada: Manitoba, Aweme. Holotype (CNC); male.

monogrammana Zeller 1875 (*Paedisca*); Verh. zool.-bot. Ges. Wien 25: 313. USA: Texas, Dallas. Holotype (BMNH); unknown.

monstratana Rebel 1906 (*Epiblema*); Dt. ent. Z. Iris 19: 235. Switzerland. Syntype(s) (Unknown); unknown.

montanana Walsingham 1884 (*Semasia*); Trans. ent. Soc. Lond. 1884: 143. USA: Montana. Holotype (BMNH); unknown.

> ***triangulana*** Kearfott 1905 (*Thiodia*); Can. Ent. 37: 46. Canada: Saskatchewan, Regina. Lectotype (AMNH); male.

mormonensis Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 44. USA: Utah, Salt Lake City. Holotype (USNM); male.

muguraxana Kostyuk 1975 (*Eucosma*); Vestnik Zool. 1975 (2): 63. Russia: Tuva, Uyukskiy. Holotype (IZUK); male.

muliebris Meyrick 1922 (*Eucosma*); Exotic Microlepid. 2: 515. Korea: Mai-Hou-Wong. Holotype (MNHN); male.

musetta Blanchard & Knudson 1983 (*Phaneta*); Proc. ent. Soc. Wash. 85: 845. USA: New Mexico, Socorro Co., Gran Quivara National Monument. Holotype (USNM); male.

nepotinana Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 263. USA: Utah, Eureka. Holotype (USNM); male.

nitorana Kuznetzov 1962 (*Eucosma*); Bull. Soc. ent. Mulhouse 1962: 57. Russia: Far East, Primorsky Krai, Yakovleka. Holotype (ZMAS); unknown.

niveicaput Walsingham 1900 (*Thiodia*); Ann. Mag. nat. Hist. (7) 6: 406. Japan. Syntypes (BMNH); male, female.

obumbratana [Lienig] & Zeller 1846 (*Grapholitha*); Isis von Oken (Leipzig) 1846 (3): 240. Latvia: [Latvia]. Syntype(s) (Unknown); unknown.

> *westwoodiana* Doubleday 1850 (*Grapholitha*); Synon. List Br. Lepid.: 26. United Kingdom: Great Britain. Syntype(s) (BMNH); unknown.
>
> *rufana* Stephens 1834 (*Peronea*); Illust. Br. Ent. (Haustellata) 4: 160. Europe. Syntype(s) (BMNH); unknown.
>
> *laharpana* Laharpe 1858 (*Semasia*); Nouv. Mm. Soc. Helv. Sci. Nat. 16: 97. Switzerland. [attributed to Rothenbach] Syntype(s) (MZL); unknown.

> *ibiceana* Herrich-Schäffer 1951 (*Tortrix* (*Semasia*)); Syst. Bearbeitung Schmett. Eur.: 249. Austria: Grossglockner. Syntypes (Unknown); unknown.

ochricostana Razowski 1972 (*Eucosma*); Acta zool. cracov. 17: 126. China: Manchuria, Kirin Province, Hsiaoling. Holotype (ISEZ); male.

ochrocephala Walsingham 1895 (*Semasia*); Trans. ent. Soc. Lond. 1895: 513. USA: Colorado, Loveland. Lectotype (BMNH); unknown.

> ***imbridana*** Fernald 1905 (*Cydia*); Can. Ent. 37: 400. USA: Kansas, Onaga. Lectotype (USNM); male.

ochroterminana Kearfott 1907 (*Thiodia*); Can. Ent. 39: 57. USA: New Jersey, Montclair. Lectotype (AMNH); male.

octopunctana Walsingham 1895 (*Semasia*); Trans. ent. Soc. Lond. 1895: 512. USA: Colorado, Larimer Co. Holotype (BMNH); unknown.

oculatana Kennel 1900 (*Semasia*); Dt. ent. Z. Iris 13: 143. Russia: Siberia, Irkutsk. Syntypes (MNHU); male, female.

offectalis Hulst 1886 (*Crambus*); Trans. Am. ent. Soc. 13: 166. USA: Colorado. Holotype (USNM); female (lost).

> ***obliterana*** Walsingham 1895 (*Semasia*); Trans. ent. Soc. Lond. 1895: 513. USA: Arizona. Holotype (BMNH); unknown.

olivaceana Riley 1881 (*Grapholitha*); Trans. St. Louis Acad. Sci 4: 320. USA: Illinois. Holotype (USNM); male.

ommatoptera Falkovitsh 1965 (*Eucosma*); Ent. Obozr. 44: 434. Russia: Primorsky Krai, 25 km SW Ussuriysk. Holotype (ZMAS); male.

> *kurilensis* Kuznetzov 1968 (*Eucosma ommatoptera* ssp.); Ent. Obozr. 47: 579. Russia Kuril Islands, Kunashir, near Sernovodsk. Holotype (ZMAS); male.

> *opulentana* Christoph 1882 (*Grapholitha*); Bull. Soc. Imp. Nat. Moscou 56(4) (1881): 413. Russia(?): Far East. Syntype(s) (Unknown); unknown.

oregonensis Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 47. USA: Oregon, Klamath Co., Crater Lake. Holotype (USNM); male.

ornatula Heinrich 1924 (*Thiodia*); J. Wash. Acad. Sci. 14: 385. USA: Pennsylvania, Allegheny Co., Oak Station. Holotype (USNM); male.

ottoniana Kennel 1919 (*Palpocrinia*); Mitt. mnch. ent. Ges. 8: 66. China: Lob-Noor. Holotype (MNHU); unknown (lost).

paetulana Kennel 1900 (*Semasia*); Dt. ent. Z. Iris 13: 145. Russia: Siberia, South Urals. Holotype (MNHU); unknown.

mirana Caradja 1916 (*Semasia*); Dt. ent. Z. Iris 30: 62. Russia: Siberia, Khabarovsky Krai, Radd. Holotype (MGAB); male.

pallidarcis Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 60. USA: California, San Diego. Holotype (AMNH); male.

pallidicostana Walsingham 1879 (*Semasia*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus (1) 4: 62. USA: California. Syntypes (BMNH), (AMNH); unknown.

paragemellana Gilligan & Wright 2012 (*Eucosma*); Zootaxa [to be published – Chapter 8]. USA: Alabama, Baldwin Co., Weeks Bay Estuarine Reserve. Holotype (USNM); male.

parmatana Clemens 1860 (*Ephippiphora*); Proc. Acad. Nat. Sci. Philad. 12: 352. USA: Pennsylvania. Lectotype (ANSP); male.

kennebecana Kearfott 1907 (*Epinotia*); Can. Ent. 39: 157. USA: Maine, Kennebunkport. Lectotype (AMNH); male.

crispana Clemens 1865 (*Steganoptycha*); Proc. ent. Soc. Philad. 5: 137. USA: Virginia? Lectotype (ANSP); female.

sinestrigana McDunnough 1938 (*Thiodia*); Can. Ent. 70: 97. Canada: Nova Scotia, Queens Co., White Point Beach. Holotype (CNC); male.

alterana Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 46. USA: Maryland, Montgomery Co., Plummers Island. Holotype (USNM); male.

perfuscana Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 40. USA: Pennsylvania, Allegheny Co., Oak Station. Holotype (AMNH); male.

pamirana Kuznetzov 1972 (*Eucosma* (*Phaneta*)); Nasekhomye Mongolii 1: 718. Tadzhikistan: Pamir, vic. Khorog, Dasht. Holotype (ZMAS); unknown.

parvana Walsingham 1879 (*Semasia*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 60. USA: northern Oregon. Holotype (BMNH); unknown.

parvulana Wilkinson 1859 (*Catoptria*); Brit. Tortrices: 91. United Kingdom: Great Britain (Isle of Wight). Lectotype (HMUG); female.

scutana Constant 1893 (*Grapholitha*); Annls Soc. ent. Fr. 62: 391. France. Syntype(s) (Unknown); unknown.

parvula Wright 2010 (*Phaneta*); J. Lepid. Soc. 64: 128. USA: Idaho, Oneida Co., Curlew National Grassland, T14S, R32E, Sec 30. Holotype (USNM); male.

pastigiata Heinrich 1929 (*Thiodia*); Proc. U.S. natn. Mus. 75: 3. USA: California, Tulare Co., Monachee Meadows. Holotype (USNM); male.

perangustana Walsingham 1879 (*Semasia*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 58. USA: California, Siskiyou Mountains. Holotype (BMNH); unknown.

pergratana Rebel 1914 (*Steganoptycha*); Dt. ent. Z. Iris 28: 274. China: Tian-Shan, Kuldsha. Lectotype (NHMV); male.

persiae Razowski 1963 (*Eucosma*); Acta zool. cracov. 8: 256. Iran: SW Persia, Shiraz. Holotype (LNK); male.

pupillana Clerck 1759 (*Phalaena*); Icones Insectorum Rariorum 1: pl. 11, fig. 9. [probably] Sweden. Syntype(s) (Unknown); unknown.

> *saerdabana* Obraztsov 1968 (*Eucosma* (*Phaneta*) *pupillana* ssp.); Tijdschr. Ent. 111: 17. Iran: Elbur Mountains, Srdab-Tal. Holotype (ZSM); male.
>
> *absynthiana* Hübner 1822 (*Olethreutes*); Syst. alphab. Verz. 58. unjustified emendation of *absinthiana*; no type.
>
> *absinthiana* Hübner [1796-1799] (*Tortrix*); Samml. Eur. Schmett. 7: pl. 6, fig. 34. Europe. Syntype(s) (Unknown); unknown.

pylonitis Meyrick 1932 (*Eucosma*); Exotic Microlepid. 4: 307. India: Kashmir, Gulmarg. Lectotype (BMNH); male.

radiatana Walsingham 1879 (*Semasia*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 55. USA: Eastern. Syntype(s) (BMNH); unknown.

raracana Kearfott 1907 (*Thiodia*); Trans. Am. ent. Soc. 33: 44. USA: Arkansas, Washington Co. Lectotype (AMNH); male.

> ***fastidiosa*** Meyrick 1912 (*Thiodia*); Ent. mon. Mag. 48: 34. unnecessary replacement name for *raracana*; no type.

refusana Walker 1863 (*Grapholita*); List Specimens lepid. Insects Colln. Br. Mus 28: 382. Canada: Hudson Bay, Albany River, St. Martin's Falls. Holotype (BMNH); male.

recentana Zerny 1933 (*Epiblema*); Dt. ent. Z. Iris 47: 16. Lebanon: Becharr. Holotype (Unknown); unknown.

> *iberica* Kuznetzov 1973 (*Eucosma recentana* ssp.); Trud. Zool. Inst. Leningrad 52: 317. Georgia: Manglisi. Holotype (ZMAS); male.

rigidana Snellen 1883 (*Grapholitha* (*Semasia*)); Tijdschr. Ent. 26: 203. Russia: Primorsky Krai, Askold Island. Lectotype (RMNH); male.

> *subrigidana* Caradja 1916 (*Epiblema*); Dt. ent. Z. Iris 30: 66. Russia: Primorsky Krai, Kazakevich. Holotype (MGAB); male.

rupestrana McDunnough 1925 (*Thiodia*); Can. Ent. 57: 17. Canada: Alberta, Nordegg. Holotype (CNC); male.

rusticana Kearfott 1905 (*Eucosma*); Proc. U.S. natn. Mus. 28: 358. USA: Texas, Kerrville. Lectotype (AMNH); male.

salmicolorana Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 62. USA: Utah, Tooele Co., Stockton. Holotype (USNM); male.

sardoensis Rebel 1935 (*Eucosma*); Dt. ent. Z. Iris 50: 95. Sardinia. Holotype (Unknown); unknown.

saussureana Benander 1928 (*Phaneta* (*Semasia*)); Ent. Tidskr. 49: 142. Sweden: Jmtland. Holotype (NHRS); male.

scalana Walsingham 1879 (*Semasia*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 57. USA: California/Oregon. Holotype (BMNH); male.

scorzonerana Benander 1942 (*Epiblema*); Opusc. Ent. 7: 44. Sweden: Oland. Holotype (Unknown); unknown.

scotiana McDunnough 1958 (*Thiodia*); Am. Mus. Novit. 1917: 7. Canada: Nova Scotia, St. Paul Island, Cabot Strait. Holotype (CNC); male.

scutiformis Meyrick, in Caradja 1931 (*Eucosma*); Bull. Sect. scient. Acad. roum 14: 64. China: Kwanhsien. Lectotype (BMNH); male.

segregata Heinrich 1924 (*Thiodia*); J. Wash. Acad. Sci. 14: 388. USA: California, Tulare Co., Monachee Meadows. Holotype (USNM); male.

setonana McDunnough 1927 (*Thiodia*); Can. Ent. 59: 273. Canada: British Columbia, Seton Lake. Holotype (CNC); male.

simplex McDunnough 1925 (*Eucosma*); Can. Ent. 57: 21. Canada: Alberta, Calgary. Holotype (CNC); male.

sombreana Kearfott 1905 (*Eucosma*); Proc. U.S. natn. Mus. 28: 357. USA: North Carolina, Tryon. Lectotype (AMNH); male.

> *phaeodes* Meyrick 1920 (*Eucosma*); Exotic Microlepid. 2: 344. unnecessary replacement name for *sombreana*; no type.

southamptonensis Heinrich 1935 (*Thiodia*); Mem. Carnegie Mus. 12 (2): 29. Canada: Nunavut Territory, Southampton Island. Holotype (CMNH); male.

sparsana Rebel 1935 (*Semasia*); Bull. Soc. Lpid. Genve 7: 175. Turkey: Ankara. Holotype (Unknown); unknown.

spectana McDunnough 1938 (*Thiodia*); Can. Ent. 70: 100. Canada: Alberta, Edmonton. Holotype (CNC); male.

spiculana Zeller 1875 (*Grapholitha*); Verh. zool.-bot. Ges. Wien 25: 289. USA: Texas, Dallas. Holotype (MCZ); female.

stramineana Walsingham 1879 (*Semasia*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 60. USA: Colorado, Denver. Holotype (BMNH); male.

striatana Clemens 1860 (*Anchylopera*); Proc. Acad. Nat. Sci. Philad. 12: 349. USA: Maryland, Baltimore or Pennsylvania, Easton. Lectotype (ANSP); female.

> ***trivittana*** Zeller 1875 (*Grapholitha*); Verh. zool.-bot. Ges. Wien 25: 287. USA: Texas. Holotype (BMNH); male.
>
> ***albicepsana*** Walker 1863 (*Paedisca*); List Specimens lepid. Insects Colln. Br. Mus 28: 379. North America. Holotype (BMNH); unknown.
>
> ***occidentalis*** Heinrich 1923 (*Thiodia striatana* ssp.); Bull. U.S. natn. Mus. 123: 58. USA: California, Siskiyou Co., Shasta Retreat. Holotype (USNM); male.

striatiradix Kuznetzov 1964 (*Eucosma* (*Phaneta*)); Trud. Zool. Inst. Leningrad 34: 262. Russia: Far East, Primorsky Krai. Holotype (ZMAS); male.

sublapidana Walsingham 1879 (*Semasia*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 59. USA: Oregon, Klamath Lakes. Holotype (BMNH); unknown.

sublucidana Kennel 1901 (*Epiblema*); Dt. ent. Z. Iris (1900) 13: 282. Spain: Cuenca. Holotype (MNHU); male.

subminimana Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 61. USA: California, San Diego. Holotype (USNM); male.

suomiana Hoffmann 1893 (*Grapholitha*); Stettin. ent. Ztg. 54: 142. Finland. Syntype(s) (Unknown); unknown.

sybillana Kennel 1919 (*Semasia*); Mitt. mnch. ent. Ges. 8: 82. Central Asia: Sayan Mountains. Holotype (MNHU); male.

taosana Wright 2005 (*Eucosma*); J. Lepid. Soc. 59: 130. USA: New Mexico, Taos Co., S side U.S. 64, 10 mi SE Tres Piedras. Holotype (USNM); male.

tarandana Möschler 1874 (*Grapholitha*); Stettin. ent. Ztg. 35: 165. Canada: Labrador. Holotype (Unknown); unknown (lost).

> ***taradana*** Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 296. misspelling of *tarandana*; no type.

tenuiana Walsingham 1879 (*Semasia*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 143. USA: California, Siskiyou Co. Holotype (BMNH); unknown.

tetraplana Möschler 1866 (*Grapholitha*); Berl. Ent. Z. 10: 148. Russia: Volgograd, Sarepta. Syntypes (Unknown); 3 males, 2 females.

tomonana Kearfott 1907 (*Eucosma*); Can. Ent. 39: 78. USA: New Jersey, Essex County Park. Lectotype (AMNH); male.

> ***limigena*** Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 35. unnecessary replacement name for *tomonana*; no type.

transversa Walsingham 1895 (*Semasia*); Trans. ent. Soc. Lond. 1895: 514. USA: Colorado, Loveland. Holotype (BMNH); unknown.

tripoliana Barrett 1880 (*Catoptria* (*Grapholitha*)); Ent. mon. Mag. 17: 84. United Kingdom: Great Britain, Essex. Holotype (BMNH); unknown.

tundrana Kennel 1900 (*Semasia*); Dt. ent. Z. Iris 13: 148. Russia: Ural and Far East. Syntypes (MNHU); male, female.

> *cordulana* Rebel 1917 (*Semasia*); Dt. ent. Z. Iris 30: 192. Europe. Holotype (Unknown); unknown.

ukranica Budashkin 2009 (*Eucosma*); Proc. Zoo. Mus. Kiev Taras Shevchenko Nat. U. (Esse) 5: 33. Ukraine Eastern Crimea, Karadagh Nature Reserve. Holotype (ZMKU); male.

umbrastriana Kearfott 1907 (*Thiodia*); Trans. Am. ent. Soc. 33: 40. USA: Ohio, Cincinnati. Lectotype (AMNH); male.

> ***roseoterminana*** Kearfott 1907 (*Thiodia*); Trans. Am. ent. Soc. 33: 40. USA: Ohio, Cincinnati. Lectotype (AMNH); male.
>
> ***subcandida*** Heinrich 1929 (*Thiodia formosana* ssp.); Proc. U.S. natn. Mus. 75: 1. Canada: Alberta, Bilby. Holotype (USNM); male.

umbraticana Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 70. USA: Colorado, Jefferson Co., foothills above Golden. Holotype (USNM); male.

urbana Kennel 1901 (*Semasia*); Dt. ent. Z. Iris 13(1900): 271. Turkey: Amasia. Lectotype (MNHU); male.

verna Miller 1971 (*Phaneta*); J. Lepid. Soc. 25: 286. Canada: Manitoba, Aweme. Holotype (AMNH); male.

vernalana McDunnough 1942 (*Thiodia*); Can. Ent. 74: 67. Canada: Ontario, Ottawa District, S March. Holotype (CNC); male.

verniochreana Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 67. USA: New Jersey, Mount Holly. Holotype (AMNH); male.

victoriana Kennel 1919 (*Epiblema*); Mitt. mnch. ent. Ges. 8 (1917-1918): 86. Russia: Siberia. Holotype (MNHU); unknown.

viridis Wright & Gilligan 2010 (*Phaneta*); J. Lepid. Soc. 64: 103. USA: Arizona, Coconino Co., Hart Prairie, 10 mi NNW Flagstaff. Holotype (USNM); male.

vogelana Wright 2010 (*Phaneta*); J. Lepid. Soc. 64: 137. USA: Colorado, Otero Co., Vogel Canyon Picnic Area, 15 mi S La Junta. Holotype (USNM); male.

wimmerana Treitschke 1835 (*Grapholitha*); Schmett. Eur. 10: 111. Europe. Lectotype (TMB); male.

> *maritimana* Walker 1863 (*Grapholita*); List Specimens lepid. Insects Colln. Br. Mus 28: 263. nomen nudum; listed as synonym of *wimmerana*, attributed to Dale; no type.

> *incana* [Lienig] & Zeller 1846 (*Grapholitha*); Isis von Oken (Leipzig) 1846 (3): 239. Latvia: Livland/Kuland. Syntype(s) (Unknown); unknown.

> *wimmeriana* Lederer 1859 (*Grapholitha*); Wien. ent. Monatschr. 3: 337. misspelling of *wimmerana*; no type.

> *gracilis* Filipjev 1924 (*Semasia*); Jahbr. Martijanova Staatsmus. Minussinsk 2 (3): 61. Russia: Siberia. Holotype (ZMAS); unknown.

yasudai Nasu 1982 (*Eucosma*); Tinea 11: 113. Japan: Honshu, Iwate Prefecture, Kuzakai. Holotype (OPU); male.

youngi McDunnough 1925 (*Thiodia*); Can. Ent. 57: 16. Canada: Alberta, Waterton Lakes. Holotype (CNC); male.

PELOCHRISTA

adamantana Guenée 1845 (*Argyroptera*); Annls Soc. ent. Fr. (2) 3: 303. Lapponia? ("North America"). Holotype (OBERC); unknown.

agassizii Robinson 1869 (*Conchylis*); Trans. Am. ent. Soc. 2: 24. USA: Texas, Waco County. Holotype (Unknown); unknown (lost).

agrestana Treitschke 1830 (*Sciaphila*); Schmett. Eur. 8: 180. Croatia: Dalmatia. Lectotype (TMB); male.

> *fultana* Zeller 1847 (*Paedisca*); Isis von Oken (Leipzig) 1847 (10): 722. Europe. Syntype(s) (Unknown); unknown.

agricolana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 42. USA: Oregon, Douglas Co., Umpqua River. Lectotype (BMNH); male.

> ***pergandeana*** Fernald 1905 (*Eucosma*); Can. Ent. 37: 399. USA: Virginia. Lectotype (USNM); male.

> ***flavana*** Fernald 1905 (*Eucosma pergandeana* ssp.); Can. Ent. 37: 399. USA: Texas Lectotype (USNM); male.

> ***argentialbana*** Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 44. USA: Texas, Bosque Co. Lectotype (BMNH); female.

> ***ochreana*** Clemens 1865 (*Steganoptycha*); Proc. ent. Soc. Philad. 5: 520. USA: Virginia (unused synonym) Lectotype (ANSP); female.

ainsliei Wright 2008 (*Pelochrista*); J. Lepid. Soc. 62: 222. USA: New Mexico, Dona Ana Co., Mesilla. Holotype (USNM); male.

albiguttana Zeller 1875 (*Paedisca*); Verh. zool.-bot. Ges. Wien 25: 313. USA: Texas, Dallas. Holotype (MCZ); unknown.

alexinschiana Peiu & Nemes 1968 (*Pseudeucosma*); Rev. Roum. Biol. Sr. Zool. 13: 339. Romania: Balta Brailei. Holotype (TMB); male.

alphabetica Walsingham 1914 (*Eucosma*); Biol. Centr.-Am. Lepid. Heterocera 4: 236. Mexico: Puebla, Popocatepetl Park. Holotype (USNM); male.

angelana Wright 2012 (*Eucosma*); J. Lepid. Soc. 66: 31. USA: California, Los Angeles Co., Mint Canyon. Holotype (EME); male.

apheliana Kennel 1901 (*Epiblema*); Dt. ent. Z. Iris 13 (1900): 277. Kazakhstan: Kirizia/Kazakhstan. Holotype (MNHU); unknown.

> *corneliana* Kennel 1919 (*Epiblema*); Mitt. mnch. ent. Ges. 8 (1918): 88. Central Asia: Dsharkent. Holotype (MNHU); unknown.

arabescana Eversmann 1844 (*Cochylis*); Fauna Lepid. Volgo-Ural.: 528. Russia: Ural Plateau. Syntype(s) (ZMAS); unknown.

argenteana Walsingham 1895 (*Paedisca*); Trans. ent. Soc. Lond. 1895: 504. USA: Colorado, Loveland. Holotype (BMNH); unknown.

> *idahoana* Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 90. USA: Idaho, Blackfoot. Holotype (AMNH); male.

atascosana Blanchard 1980 (*Eucosma*); J. Lepid. Soc. 33 (1979): 212. USA: Texas, Cameron Co., Laguna Atascosa. Holotype (USNM); male.

atomosana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 42. USA: California, San Francisco. Syntypes (BMNH), (USNM); males.

aureliana Popescu-Gorj 1984 (*Pelochrista*); Trav. Mus. Hist. Nat. Gr. Antipa 25: 233. Romania: Danube Delta, Fort Letea. Holotype (MGAB); male.

aurilineana Ferris 2005 (*Eucosma*); Zootaxa 806: 2. USA: Wyoming, Albany Co., 1.6 km SE Laramie. Holotype (USNM); male.

austrina Miller 1985 (*Eucosma*); Ann. ent. Soc. Am. 78: 243. USA: Texas, Hemphill Co., Canadian. Holotype (USNM); male.

avalona McDunnough 1938 (*Eucosma*); Can. Ent. 70: 58. USA: California, Santa Catalina Island, Avalon. Holotype (CNC); male.

barbara Miller 1974 (*Eucosma*); Ann. ent. Soc. Am. 67: 604. USA: Minnesota, Ramsey Co., North Oaks. Holotype (AMNH); female.

biplagata Walsingham 1895 (*Paedisca*); Trans. ent. Soc. Lond. 1895: 507. USA: Colorado, Loveland. Holotype (BMNH); unknown.

biquadrana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 45. USA: California, Shasta Co., Pitt River. Syntypes (USNM), (BMNH); males.

> ***palousana*** Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 34. USA: Washington, Pullman. Lectotype (AMNH); male.
>
> ***tahoensis*** Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 112. USA: California, Lake Tahoe, Deer Park Springs. Holotype (USNM); male.

blanchardi Wright 2012 (*Eucosma*); J. Lepid. Soc. 66: 27. USA: Arizona, Pima Co., Madera Canyon, Santa Rita Mountains, 4400 ft. Holotype (USNM); male.

bleuseana Oberthur 1888 (*Grapholitha*); Etudes Ent. Comp. 12: 42. Morocco. Syntype(s) (Unknown); unknown.

> *nubilana* Caradja 1916 (*Epiblema bleuseana* var.); Dt. ent. Z. Iris 30: 64. Algeria: Lambze. Lectotype (MGAB); male.
>
> *beluseana* Vives Moreno 1991 (*Pelochrista*); Cat. Sist. Sinon. Lepid. Penin. Iberica Baleares: 177. misspelling of *bleuseana*; no type.

bolanderana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 42. USA: California, Mount Shasta. Syntypes (BMNH), (MCZ), (AMNH); 12 males.

buddhana Kennel 1919 (*Epiblema*); Mitt. mnch. ent. Ges. 8 (1917-1918): 92. China: West China. Holotype (MNHU); unknown.

caecimaculana Hübner [1796-1799] (*Tortrix*); Samml. Eur. Schmett. 7: pl. 5, fig. 27. Europe. Syntype(s) (Unknown); unknown.

> *coecimaculana* Frölich 1828 (*Tortrix*); Enum. Tortr. Wrtemberg : 97. misspelling of *caecimaculana*; no type.
>
> *kollariana* Herrich-Schäffer 1851 (*Tortrix* (*Semasia*)); Syst. Bearbeitung Schmett. Eur. 4: 246. Germany [attributed to Mann]. Syntype(s) (Unknown); unknown.
>
> *kollariana* Herrich-Schäffer 1848 (uninominal); Syst. Bearbeitung Schmett. Eur. 4: pl. 42, fig. 295. nomen nudum; no type.

caementana Christoph 1872 (*Grapholitha*); Horae Soc. ent. Ross. 9: 14. Russia: Volgograd Region, Sarepta. [attributed to Zeller] Syntype(s) (Unknown); unknown.

canana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 50. USA: California. Holotype (USNM); male.

canariana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 18. USA: Utah, Stockton. Lectotype (AMNH); male.

candida Wright 2012 (*Eucosma*); J. Lepid. Soc. 66: 31. USA: California, Los Angeles Co., Hungry Valley, 4 air miles S Gorman. Holotype (EME); male.

caniceps Walsingham 1884 (*Paedisca*); Trans. ent. Soc. Lond. 1884: 137. USA: Montana. Holotype (BMNH); unknown.

cannatana Trematerra 2000 (*Epiblema*); Boll. Zool. agr. Bachic. (2) 32 (2): 86. Italy: Sicily. Holotype (TREMC); unknown.

cataclystiana Walker 1863 (*Paedisca*); List Specimens lepid. Insects Colln. Br. Mus 28: 378. North America. Holotype (BMNH); female.

cinereolineana Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 52. USA: Utah, Juab Co., Eureka. Holotype (USNM); male.

coconana Wright 2011 (*Eucosma*); J. Lepid. Soc. 65: 110. USA: Arizona, Coconino Co., Fort Valley, 7.5 mi. NW Flagstaff, 7350 ft. Holotype (USNM); male.

collilonga Blanchard & Knudson 1984 (*Pelochrista*); Proc. ent. Soc. Wash. 86: 446. USA: Texas, Brown Co., Lake Brownwood State Park. Holotype (USNM); male.

comatulana Zeller 1876 (*Paedisca*); Verh. zool.-bot. Ges. Wien 25: 316. USA: Texas, Bosque Co. Holotype (BMNH); female.

> ***mandana*** Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 17. USA: Washington, District of Columbia. Lectotype (AMNH); female.
>
> ***amanda*** Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 34. unnecessary replacement name for *mandana*; no type.

confidana Chrétien 1915 (*Epiblema*); Annls Soc. ent. Fr. 84: 302. Tunisia: Thala. Holotype (MNHN); male.

consobrinana Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 128. USA: South Dakota, Elk Point. Holotype (USNM); male.

consociana Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 101. USA: Utah, Juab Co., Eureka. Holotype (USNM); male.

conspiciendana Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 135. USA: Utah, Stockton. Holotype (AMNH); male.

corosana Walsingham 1884 (*Paedisca*); Trans. ent. Soc. Lond. 1884: 139. USA: Montana. Holotype (BMNH); unknown.

costastriata Wright 2011 (*Eucosma*); J. Lepid. Soc. 65: 114. USA: Nebraska, Cherry Co., Valentine NWR, Hackberry Lake. Holotype (USNM); male.

costastrigulana Kearfott 1907 (*Eucosma*); J. New York ent. Soc. 16: 171. USA: California, San Diego. Lectotype (AMNH); male.

crambitana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 43. USA: California, Mount Shasta. Syntypes (BMNH); male.

curlewensis Wright 2007 (*Eucosma*); J. Lepid. Soc. 61: 47. USA: Idaho, Oneida Co., Curlew National Grassland, 4 mi ENE of Holbrook. Holotype (USNM); male.

daemonicana Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 11. USA: New Mexico, Manzano National Forest, Hell Canyon. Holotype (USNM); male.

dagestana Obraztsov 1949 (*Pseudeucosma*); Mitt. mnch. ent. Ges. 35-39: 219. Russia: Daghestan. Holotype (ZSM); male.

> *pinxana* Kuznetzov 1976 (*Pelochrista*); Trud. Inst. Zool. Leningrad 64: 29. Azerbaijan. Holotype (ZMAS); male.

danilevskyi Kostyuk 1975 (*Pelochrista*); Vestnik Zool. 1975 (2): 60. Russia: Tuva, Tsagan-Shibetu. Holotype (IZUK); male.

decolorana Freyer 1842 (*Paedisca*); Neuere Beitr. Schmett.-Kunde 4: 48. Europe. Syntype(s) (Unknown); unknown.

denverana Kearfott 1907 (*Eucosma*); Can. Ent. 39: 77. USA: Colorado, Denver. Lectotype (AMNH); male.

derelicta Heinrich 1929 (*Eucosma*); Proc. U.S. natn. Mus. 75: 13. USA: North Carolina, Tryon. Holotype (USNM); male.

diabolana Blanchard 1980 (*Eucosma*); J. Lepid. Soc. 33 (1979): 214. USA: Texas, Culberson, Co., Sierra Diablo Wildlife Management Area. Holotype (USNM); male.

dilatana Walsingham 1895 (*Paedisca*); Trans. ent. Soc. Lond. 1895: 510. USA: Arizona. Holotype (BMNH); unknown.

dira Razowski 1972 (*Pelochrista*); Acta zool. cracov. 17: 136. Mongolia: Suchebaator Province, Molzog. Holotype (TMB); male.

disquei Kennel 1901 (*Epiblema*); Dt. ent. Z. Iris 13 (1900): 274. Mongolia: Zavhan Province, Uliassutai. Holotype (MNHU); unknown.

> *congeminata* Obraztsov 1967 (*Pelochrista* (*Pseudeucosma*)); Tijdschr. Ent. 110: 80. Mongolia: Zavhan Province, Uliassutai. Holotype (Unknown); unknown.

dodana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 27. USA: southwest Colorado. Lectotype (AMNH); male.

> ***spilophora*** Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 35. unnecessary replacement name for *dodana*; no type.
>
> ***fofana*** Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 28. USA: Colorado, Berthoud Pass. Holotype (AMNH); male.
>
> ***annulata*** Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 35. unnecessary replacement name for *fofana*; no type.

dorsisignatana Clemens 1860 (*Poecilochroma*); Proc. Acad. Nat. Sci. Philad. 12: 353. USA: Pennsylvania. Lectotype (ANSP); male.

> ***distigmana*** Walker 1863 (*Carpocapsa*); List Specimens lepid. Insects Colln. Br. Mus 28: 394. North America. Holotype (BMNH); female.
>
> ***engelana*** Kearfott 1908 (*Eucosma*); J. New York ent. Soc. 16: 169. USA: Pennsylvania, Pittsburgh. Lectotype (AMNH); male.
>
> ***confluana*** Kearfott 1905 (*Eucosma dorsisignatana* var.); Proc. U.S. natn. Mus. 28: 355. USA: New Jersey, Essex Co. Syntypes (USNM); unknown.
>
> ***diffusana*** Kearfott 1905 (*Eucosma dorsisignatana* var.); Proc. U.S. natn. Mus. 28: 355. USA: Louisiana, Vernon Parish. Lectotype (AMNH); male.
>
> ***clavana*** Zeller 1876 (*Paedisca*); Verh. zool.-bot. Ges. Wien 25: 303. USA: Massachusetts, Cambridge. Lectotype (BMNH); male.

duercki Osthelder 1941 (*Epiblema*); Mitt. mnch. ent. Ges. 31: 369. Greece: Crete, Ida Mt. Holotype (Unknown); unknown.

edrisiana Chrétien, in Oberthur 1922 (*Epiblema*); Etudes Lepid. Comp 19 (2): 88. Morocco: Fez. Holotype (MNHN); male.

elegantana Kennel 1901 (*Epiblema*); Dt. ent. Z. Iris (1900) 13: 276. Algeria: Teniet el Haad. Holotype (MNHU); unknown.

emaciatana Walsingham 1884 (*Paedisca*); Trans. ent. Soc. Lond. 1884: 137. USA: Arizona. Lectotype (BMNH); male.

> *perpropinqua* Heinrich 1929 (*Eucosma*); Proc. U.S. natn. Mus. 75: 8. USA: Arizona, Pima Co., Indian Oasis, Sells Post Office. Holotype (USNM); male.

eversmanni Kennel 1901 (*Epiblema*); Dt. ent. Z. Iris (1900) 13: 275. Mongolia: Zavhan Province, Uliassutai. Holotype (MNHU); unknown.

> *aristidana* Rebel 1910 (*Semasia*); Dt. ent. Z. Iris 24: 7. Tadzhikistan: Altai Mountains. Lectotype (MGAB); male.

excerptionana Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 117. USA: Nevada, Washoe Co., Verdi. Holotype (USNM); male.

exclusoriana Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 110. USA: Texas, La Salle Co., Cotulla. Holotype (USNM); male.

excusabilis Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 123. USA: California, Lake Tahoe. Holotype (USNM); male.

expolitana Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 132. USA: Utah, Utah Co., Provo. Holotype (USNM); male.

fandana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 19. USA: Colorado, Denver. Holotype (AMNH); male(?).

> ***gandana*** Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 20. USA: Colorado, Denver. Holotype (AMNH); female.
>
> ***argyraula*** Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 34. unnecessary replacement name for *fandana*; no type.
>
> ***chloroleuca*** Meyrick 1912 (*Eucosma*) Ent. mon. Mag. 48: 34. unnecessary replacement name for *gandana*; no type.

fernaldana Grote 1880 (*Paedisca*); N. Am. Ent. 1880: 98. USA: Colorado. Syntype (USNM); male.

> ***magnidicana*** Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 83. USA: Southwest Colorado. Holotype (AMNH); male.

figurana Razowski 1972 (*Pelochrista*); Acta zool. cracov. 17: 137. Mongolia: Mongolia Central [Tov] Province. Holotype (TMB); male.

fiskeana Kearfott 1905 (*Eucosma*); Proc. U.S. natn. Mus. 28: 358. USA: North Carolina, Tryon. Lectotype (AMNH); male.

floridensis Wright 2011 (*Eucosma*); J. Lepid. Soc. 65: 111. USA: Florida, Highlands Co., Archbold Biol[ogical] Sta[tion], Lake Placid. Holotype (USNM); male.

fratruelis Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 98. USA: North Carolina, Southern Pines. Holotype (USNM); male.

fraudabilis Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 98. USA: North Carolina, Southern Pines. Holotype (USNM); male.

fritillana Blanchard & Knudson 1982 (*Eucosma*); J. Lepid. Soc. 35 (1981): 170. USA: Texas, Anderson Co., Engeling Wildlife Management Area, near Tennessee Colony. Holotype (USNM); male.

frustata Razowski 2006 (*Pelochrista*); Acta Zool. Cracov. 49B: 128. India: Jammu and Kashmir (Indien J & K, Kaschmir Ladakh, 15 km E Drass). Holotype (ISEZ); male.

fulvostrigana Constant 1888 (*Grapholitha*); Annls Soc. ent. Fr. (6) 8: 168. France: Corsica. Syntype(s) (Unknown); unknown.

fuscosparsa Walsingham 1895 (*Paedisca*); Trans. ent. Soc. Lond. 1895: 507. USA: Colorado, Loveland. Syntype(s) (BMNH); unknown.

fuscostriata Wright 2008 (*Pelochrista*); J. Lepid. Soc. 62: 228. USA: California, San Mateo Co., Edgewood Park. Holotype (EME); male.

fusculana Zeller 1847 (*Paedisca*); Isis von Oken (Leipzig) 1847 (10): 729. Italy: Sicily. Syntype (Unknown); unknown.

obstinatana Kennel 1901 (*Epiblema*); Dt. ent. Z. Iris (1900) 13: 286. Morocco: Mauretania. Syntype(s) (MNHU); unknown.

crustulana Lederer 1859 (*Tortrix*); Wien. ent. Monatschr. 3: 332. Croatia: Dalmatia. [attributed to Mann] Syntype(s) (MNHU); unknown.

galenapunctana Kearfott 1905 (*Eucosma*); J. New York ent. Soc. 16: 169. USA: Colorado. Lectotype (AMNH); female.

gelattana Wright 2007 (*Pelochrista*); J. Lepid. Soc. 61: 121. USA: Wyoming, Albany Co., W side of Gelatt Lake. Holotype (USNM); male.

gilletteana Dyar 1903 (*Eucosma*); Proc. ent. Soc. Wash. 5: 180. USA: Arizona, Williams; Colorado. Syntypes (USNM); female.

gilligani Wright 2008 (*Pelochrista*); J. Lepid. Soc. 62: 227. USA: Utah, Sanpete Co., Ephraim Canyon Road. Holotype (USNM); male.

graciliana Kearfott 1905 (*Eucosma*); Proc. U.S. natn. Mus. 23: 352. USA: North Carolina, Tryon. Lectotype (AMNH); female.

graduatana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 54. USA: Texas. Holotype (BMNH); unknown.

grandiflavana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 47. USA: California, Lake Co. Syntypes (BMNH), (USNM); female(s).

griselda Blanchard & Knudson 1982 (*Eucosma*); J. Lepid. Soc. 35 (1981): 173. USA: Texas, Brewster Co., Big Bend National Park, Chisos Basin. Holotype (USNM); male.

griseolana Zeller 1847 (*Paedisca*); Isis von Oken (Leipzig) 1847 (10): 726. Italy: Sicily, Messina. Syntype (Unknown); unknown.

guentheri Tengstrom 1869 (*Grapholitha*); Acta Soc. Fauna Flora Fenn. Frh. 10: 361. Russia: Karelia, Petrosawodsk. Syntypes (ZMH); unknown.

guttulana Blanchard 1980 (*Eucosma*); J. Lepid. Soc. 22 (1979): 214. USA: Texas, Kennedy Co., Padre Island National Seashore. Holotype (USNM); male.

handana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 20. USA: Utah, Stockton. Lectotype (AMNH); male.

caramitis Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 102. misspelling of *ceramitis*; no type.

ceramitis Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 34. unnecessary replacement name for *handana*; no type.

hasseanthi Clarke 1952 (*Eucosma*); Bull. S. Calif. Acad. Sci 52: 60. USA: California, Orange Co. Holotype (USNM); male.

hazelana Klots 1936 (*Eucosma*); Am. Mus. Novit. 867: 1. USA: Colorado, Colorado Springs, Fountain Valley School. Holotype (AMNH); male.

heathiana Kearfott 1907 (*Eucosma*); Can. Ent. 39: 56. Canada: Manitoba, Cartwright. Lectotype (AMNH); male.

hepatariana Herrich-Schäffer 1851 (*Tortrix* (*Paedisca*)); Syst. Bearbeitung Schmett. Eur. 4: 238. Hungary. Syntype(s) (Unknown); unknown.

> *obnuptana* Herrich-Schäffer 1851 (*Tortrix* (*Paedisca*)); Syst. Bearbeitung Schmett. Eur. 4: 238. Germany. Syntype(s) (Unknown); unknown.

hohana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 28. Canada: Alberta, Mount Piran. Lectotype (AMNH); male.

> ***syrtodes*** Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 35. unnecessary replacement name for hohana; no type.

huebneriana [Lienig] & Zeller 1846 (*Paedisca*); Isis von Oken (Leipzig) 1846 (3): 237. Latvia: Livlandia (East Balticum). Syntype(s) (Unknown); unknown.

hyponomeutana Walsingham 1895 (*Paedisca*); Trans. ent. Soc. Lond. 1895: 502. USA Colorado, Loveland. Holotype (BMNH); unknown.

idotatana Kennel 1901 (*Epiblema*); Dt. ent. Z. Iris (1900) 13: 277. Tian Shan: Thian-Shan (Kulda). Syntype(s) (MNHU); unknown.

> *idodatana* Kennel 1921 (*Epiblema*); Palaear. Tortr.: 556. misspelling of *idotatana*; no type.

immaculana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 35. USA: Washington, Pullman. Lectotype (AMNH); female.

infidana Hübner [1823-1824] (*Tortrix*); Samml. Eur. Schmett 7: pl. 47, figs. 296-298. Europe. Syntype(s) (Unknown); unknown.

> *bulgarica* Soffner 1967 (*Epiblema infidana* ssp.); Mitt. mnch. ent. Ges. 57: 112. Bulgaria: Nesebar. Holotype (Unknown); unknown.

inquadrana Walsingham 1884 (*Aphelia*?); Trans. ent. Soc. Lond. 1884: 134. USA: Arizona. Syntype(s) (BMNH); unknown.

invicta Walsingham 1895 (*Paedisca*); Trans. ent. Soc. Lond. 1895: 509. USA: Colorado. Holotype (BMNH); unknown.

invisitana Kuznetzov 1986 (*Pelochrista*); Vestnik Zool. 1986 (4): 24. Turkey: (Mardin) Asia Minor. Holotype (MNHU); male.

irroratana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 48. USA: California, Mendocino Co. Syntypes (USNM), (BMNH); male(s).

> ***perdricana*** Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 49. USA: California. Holotype (BMNH); unknown.

jodocana Kennel 1919 (*Epiblema*); Mitt. mnch. ent. Ges. 8 (1917-1918): 87. Russia: Tuva. Holotype (MNHU); unknown.

johnstoni Wright 2012 (*Eucosma*); J. Lepid. Soc. 66: 34. USA: Arizona, Mojave County, 3 mi. SE Kingman. Holotype (EME); male.

kandana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 20. USA: Utah, Stockton. Lectotype (AMNH); male.

> ***argillacea*** Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 34. unnecessary replacement name for kandana; no type.

kingi Wright 2008 (*Pelochrista*); J. Lepid. Soc. 62: 225. Canada: Saskatchewan, Saskatoon. Holotype (CNC); male.

kuznetzovi Kostyuk 1975 (*Pelochrista*); Vestnik Zool. 1975 (6): 39. Russia: Siberia, Dauriya. Holotype (IZUK); male.

labyrinthicana Christoph 1872 (*Grapholitha*); Horae Soc. ent. Ross. 9: 17. Russia: Volgograd R, Sarepta. Syntype(s) (Unknown); unknown.

lafontainei Wright 2012 (*Eucosma*); J. Lepid. Soc. 66: 30. USA: Wyoming, Albany Co., W side Gelatt Lake. Holotype (USNM); male.

langstoni Powell 1963 (*Eucosma*); Proc. Biol. Soc. Wash. 76: 235. USA: California, San Luis Obispo Co., Pozo. Holotype (CAS); male.

larana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 43. USA: California, Siskiyou Co., Sheep Rock. Lectotype (BMNH); male.

latericiana Rebel 1919 (*Epiblema*); Verh. zool.-bot. Ges. Wien 69: 126. Europe: Croatia(?). Syntype(s) (Unknown); unknown.

> *croatica* Rebel 1893 (*Paedisca*); Stettin. ent. Ztg. 54: 41. nomen nudum; no type.

lathami Forbes 1937 (*Eucosma*); J. New York ent. Soc. 45: 131. USA: New York, Long Island, Orient. Holotype (USNM); male.

laticurva Heinrich 1929 (*Eucosma*); Proc. U.S. natn. Mus. 75: 4. USA: California, Sierra Nevada. Holotype (USNM); male.

latipalpana Razowski 1967 (*Pseudeucosma*); Beitr. Naturk. Forsch. SdwDtl. 26: 95. Afghanistan: Panjao. Holotype (LNK); male.

lineolana Kuznetzov 1964 (*Pelochrista*); Trud. Zool. Inst. Leningrad 34: 259. Kazakhstan. Holotype (ZMAS); male.

lolana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 29. USA: Colorado. Lectotype (AMNH); male.

> ***leucomalla*** Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 35. unnecessary replacement name for *lolana*; no type.

lugubrana Treitschke 1830 (*Penthina*); Schmett. Eur. 8: 26. Hungary: Ofen. Lectotype (TMB); male.

> ***tariki*** Hartig 1949 (*Pygolopha*); Boll. Assoc. Rom. Ent. 4: 2. Italy: Garda Lake. Holotype (Unknown); unknown.
>
> ***tinacriana*** Lederer 1859 (*Pygolopha*); Wien. ent. Monatschr. 3: 280. Italy: Sicily, Messina. Lectotype (MNHU); male.
>
> ***trinacriana*** Rebel 1901 (*Pygolopha*); Cat. Lepid. Pal. Faun. 2: 115. misspelling of tinacriana; no type.

luridana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 44. USA: northern California. Holotype (BMNH); unknown.

maculatana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 48. USA: California, Lake Co. Syntypes (USNM), (BMNH), (AMNH); 2 females, others unknown.

maculosa Wright 2012 (*Eucosma*); J. Lepid. Soc. 66: 30. USA: Wyoming, Albany Co., W side Gelatt Lake, 7250 ft. Holotype (USNM); male.

mancipiana Mann 1855 (*Paedisca*); Verh. zool.-bot. Ges. Wien 5: 556. Italy: Corsica. Syntype(s) (Unknown); unknown.

matutina Grote 1873 (*Penthina*); Bull. Buffalo Soc. Nat. Sci 1: 92. USA: Texas. Neotype (BMNH); female.

> ***grotiana*** Kearfott 1908 (*Eucosma*); J. New York ent. Soc. 16: 179. USA: Iowa. Lectotype (AMNH); female.

mediostriata Walsingham 1895 (*Paedisca*); Trans. ent. Soc. Lond. 1895: 508. USA: Colorado, Loveland. Syntype(s) (BMNH); unknown.

> ***sepulcrana*** Meyrick 1927 (*Eucosma*); Exotic Microlepid. 3: 334. USA: Utah, Dividend. Lectotype (BMNH); male.
>
> ***sepulchrana*** Clarke 1958 (*Eucosma*); Cat. Type Spec. Microlepid. Br. Mus (Nat. Hist.) Descr. Edward Meyrick 3: 420. misspelling of *sepulcrana*; no type.

medullana Staudinger 1880 (*Grapholitha*); Horae Soc. ent. Ross. 15 (1979): 254. Turkey: Smyrna. Lectotype (MNHU); female.

mescalerana Wright 2012 (*Eucosma*); J. Lepid. Soc. 66: 28. USA: New Mexico, Chaves County, Mescalero Dunes east of Roswell. Holotype (USNM); male.

metariana Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 133. USA: California, Siskiyou Co., Shasta Retreat. Holotype (USNM); male.

metria Falkovitsh 1964 (*Pelochrista*); Trud. Zool. Inst. Leningrad 34: 274. Kazakhstan. Holotype (ZMAS); male.

milleri Wright 2007 (*Pelochrista*); J. Lepid. Soc. 61: 84. USA: Ohio, Adams Co., 1 mi SE of Lynx. Holotype (USNM); male.

mirosignata Heinrich 1929 (*Eucosma*); Proc. U.S. natn. Mus. 75: 11. USA: Arizona. Pima Co., Baboquivari Mountains. Holotype (USNM); male.

mobilensis Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 125. USA: Alabama, Mobile Bay, near Daphne. Holotype (USNM); male.

modicana Zeller 1847 (*Paedisca*); Isis von Oken (Leipzig) 1847 (10): 727. Italy: Sicily, Syracuse. Syntype (Unknown); unknown.

> *definitana* Kennel 1901 (*Epiblema*); Dt. ent. Z. Iris (1900) 13: 285. Turkey: Asia Minor (Amasia). Lectotype (MNHU); male.
>
> *hinnebergiana* Fuchs 1902 (*Semasia*); Stettin. ent. Ztg. 63: 321. Germany: Bornich. Syntype(s) (Unknown); unknown.
>
> *modestana* Herrich-Schäffer 1851 (*Tortrix* (*Semasia*)); Syst. Bearbeitung Schmett. Eur. 4: 249. Austria. [attributed to Frölich] Syntype(s) (Unknown); unknown.
>
> *modestana* Herrich-Schäffer 1848 (uninominal); Syst. Bearbeitung Schmett. Eur. 4: pl. 42, Fig. 301. nomen nudum; no type.

mojaveana Wright 2011 (*Eucosma*); J. Lepid. Soc. 65: 117. USA: California, San Bernardino Co, Afton Road, 23 mi SW Baker. Holotype (USNM); female.

mollitana Zeller 1847 (*Paedisca*); Isis von Oken (Leipzig) 1847 (10): 724. Italy: Sicily, Syracuse. Syntype (Unknown); unknown.

> *commodestana* Rössler 1877 (*Grapholitha*); Stettin. ent. Ztg. 38: 375. Italy: Sicily. Syntype(s) (Unknown); unknown.
>
> *modestana* Seebold 1879 (*Grapholitha*); Ann. Soc. Espa. Hist. Nat. 8: 122. Spain. Syntype(s) (MNCNM); unknown.
>
> *trisignana* Nolcken 1870 (*Grapholitha*); Arb. Naturf.-Ver. Riga 3: 427. Rotsikull. Syntype(s) (Unknown); unknown.

momana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 30. USA: Arizona, Yuma Co. Holotype (AMNH); male.

metaschista Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 35. unnecessary replacement name for *momana*; no type.

morrisoni Walsingham 1884 (*Paedisca*); Trans. ent. Soc. Lond. 1884: 138. USA: Montana. Syntype(s) (BMNH); unknown.

muhabbet Koçak, in Koçak & Kemal 2006 (*Pelochrista*); Misc. Papers, Centre Entomol. Studies Ankara 98: 4. replacement name for chanana Kennel, 1901; no type.

chanana Kennel 1901 (*Epiblema*); Dt. ent. Z. Iris 13 (1900): 273. Russia: Caucasus. Holotype (MNHU); unknown.

nandana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 17. Canada: Manitoba, Rounthwaite. Lectotype (AMNH); female.

chersaea Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 34. unnecessary replacement name for *nandana*; no type.

navajoensis Wright 2011 (*Eucosma*); J. Lepid. Soc. 65: 111. USA: Utah, San Juan Co., Comb Ridge west of Bluff. Holotype (USNM); male.

nordini Wright 2005 (*Eucosma*); J. Lepid. Soc. 59: 129. USA: Wyoming, Albany Co., Medicine Bow NF, 11.5 mi SE Laramie, Jct. Forest Roads 707 and 705. Holotype (USNM); male.

notialis Miller 1985 (*Eucosma*); Ann. ent. Soc. Am. 78: 244. USA: Texas, Kerr Co., Kerrville. Holotype (USNM); male.

notocelioides Oku 1972 (*Pelochrista*); Konty 40: 264. Japan: Honshu, Iwate Prefecture, Morioka. Holotype (EIHU); male.

nuntia Heinrich 1929 (*Eucosma*); Proc. U.S. natn. Mus. 75: 10. USA: Utah, Juab Co., Callao. Holotype (USNM); male.

obscura Kuznetzov 1978 (*Pelochrista*); Opred. Nasekomykh Evop. SSSR 4: 545. Kazakhstan. Holotype (ZMAS); unknown.

optimana Dyar 1903 (*Eucosma*); Proc. ent. Soc. Wash. 5: 180. USA: Colorado, Garfield Co., Glenwood Springs. Syntype (USNM); male.

oraria Wright 2011 (*Eucosma*); J. Lepid. Soc. 65: 180. Nova Scotia: Kings County, Grand Pre. Holotype (USNM); male.

ornamentana Rebel 1916 (*Semasia*); Dt. ent. Z. Iris 30: 192. Russia: Tuva. Holotype (Unknown); unknown.

ornata Kuznetzov 1967 (*Pelochrista*); Trud. Zool. Inst. Leningrad 41: 67. Russia: Amur Region. Holotype (ZMAS); male.

palabundana Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 267. Canada: Manitoba, Aweme. Holotype (CNC); male.

pallidipalpana Kearfott 1905 (*Eucosma*); Proc. U.S. natn. Mus. 28: 353. USA: District of Columbia. Lectotype (AMNH); male.

palpana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus (1) 4: 54. USA: California, Mount Shasta Syntypes (BMNH), (AMNH); 2 males.

parapulveratana Wright 2011 (*Eucosma*); J. Lepid. Soc. 65: 110. USA: Kansas, Morton Co., Cimarron R. & Hwy 51. Holotype (USNM); male.

passerana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 49. USA: California, Mendocino Co. Syntypes (USNM), (BMNH); female.

pediasios Miller 1985 (*Eucosma*); Ann. ent. Soc. Am. 78: 241. USA: Texas, Terrel Co., Sanderson. Holotype (USNM); male.

pfisteri Obraztsov 1952 (*Eucosma* (*Eucosma*)); Z. Wien. ent. Ges. 37: 122. Germany: Tauberbischofsheim, Baden. Holotype (PFISC); male.

piperata Wright 2005 (*Eucosma*); J. Lepid. Soc. 59: 129. USA: Utah, Vineyard. Holotype (USNM); male.

pollinaria Diakonoff 1971 (*Pelochrista*); Verff. Zool. Staatsamml. Mnchen 15: 181. India/Pakistan: NW Karakorum, Gilgit, Banidas. Holotype (ZSM); male.

popana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 31. USA: Utah, Stockton. Lectotype (AMNH); male.

> *carcharias* Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 35. unnecessary replacement name for *popana*; no type.

powelli Wright 2005 (*Pelochrista*); J. Lepid. Soc. 59: 132. USA: Idaho, Oneida Co., Curlew NG, 4 mi ENE Holbrook, jct Forest Roads 056 and 057. Holotype (USNM); male.

primulana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus (1) 4: 45. USA: California, Mendocino Co. Syntypes (BMNH), (MCZ); unknown.

pulveratana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 45. USA: California, San Francisco. Syntypes (BMNH), (USNM); male.

quinquemaculana Robinson 1869 (*Conchylis*); Trans. Am. ent. Soc. 2: 284. USA: Pennsylvania. Lectotype (AMNH); male.

ragonoti Walsingham 1895 (*Paedisca*); Trans. ent. Soc. Lond. 1895: 503. USA: Colorado, Loveland. Holotype (BMNH); unknown.

> ***barnesiana*** Dyar 1903 (*Eucosma ragonoti* ssp.); Proc. ent. Soc. Wash. 5: 180. USA: Colorado, Glenwood Springs. Holotype (USNM); male.

reversana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 22. USA: Texas, San Antonio. Lectotype (AMNH); male.

ridingsana Robinson 1869 (*Conchylis*); Trans. Am. ent. Soc. 2: 285. USA: Colorado. Holotype (Unknown); unknown (lost).

> ***argentifurcatana*** Grote 1876 (*Conchylis*); Can. Ent. 8: 206. Canada: Ontario, Port Stanley. Syntype (USNM); male.
>
> ***hipeana*** Grote 1876 (*Conchylis*); Can. Ent. 8: 207. Canada: Ontario, Port Stanley. Holotype (USNM); male.

rindgei Miller 1985 (*Eucosma*); Ann. ent. Soc. Am. 78: 243. USA: Colorado, Denver. Holotype (AMNH); male.

robinsonana Grote 1872 (*Conchylis*); Can. Ent. 4: 101. USA: Alabama. Holotype (ANSP); male.

> ***tryonana*** Kearfott 1905 (*Eucosma robinsonana* ssp.); Proc. U.S. natn. Mus. 38: 350. USA: North Carolina, Tryon. Lectotype (AMNH); male.
>
> ***quintana*** Zeller 1875 (*Paedisca*); Verh. zool.-bot. Ges. Wien 25: 304. USA: Texas, Dallas. Syntypes (2) (MCZ); unknown.

rorana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 31. USA: Utah, Stockton. Lectotype (AMNH); male.

> *sceletopa* Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 35. unnecessary replacement name for *rorana*; no type.

rosaocellana Knudson 1986 (*Eucosma*); J. Lepid. Soc. 40: 322. USA: Texas, Hemphill Co., Gene Howe WMA. Holotype (USNM); male.

rufocostana Wright 2012 (*Eucosma*); J. Lepid. Soc. 66: 35. USA: Idaho, Oneida Co., Curlew National Grassland, 5 mi. SSE of Holbrook, 4800 ft. Holotype (USNM); male.

rushana Obraztsov 1943 (*Epiblema*); Mitt. mnch. ent. Ges. 33: 100. Russia: Pamirs, Vamardara. Holotype (ZMKU); male.

russeola Heinrich 1929 (*Eucosma*); Proc. U.S. natn. Mus. 75: 6. USA: California, Los Angeles Co. Holotype (USNM); male.

salaciana Blanchard & Knudson 1982 (*Eucosma*); J. Lepid. Soc. 35 (1981): 176. USA: Texas, Nueces Co., North Padre Island. Holotype (USNM); male.

sandiego Kearfott 1908 (*Eucosma*); J. New York ent. Soc. 16: 172. USA: California, San Diego. Syntypes (AMNH); female.

> ***sandiegana*** Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 34. unnecessary replacement name for *sandiego*; no type.

scintillana Clemens 1865 (*Callimosema*); Proc. ent. Soc. Philad. 5: 142. USA: Pennsylvania. Holotype (ANSP); male.

randana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 21. USA: Colorado, Denver. Lectotype (AMNH); male.

paraglypta Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 34. unnecessary replacement name for *randana*; no type.

dodecana Zeller 1875 (*Paedisca*); Verh. zool.-bot. Ges. Wien 25: 311. USA: Texas, Dallas. Syntypes (5) (MCZ); unknown.

seamansi Wright 2011 (*Eucosma*); J. Lepid. Soc. 65: 105. Canada: Alberta, Lethbridge. Holotype (CNC); male.

sepiana Wright 2011 (*Eucosma*); J. Lepid. Soc. 65: 109. USA: Utah, [Juab Co.], Eureka. Holotype (USNM); male.

serapicana Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 266. USA: Montana, Cascade Co., Great Falls. Holotype (USNM); male.

serpentana Walsingham 1895 (*Paedisca*); Trans. ent. Soc. Lond. 1895: 504. USA: Colorado, Loveland. Holotype (BMNH); unknown.

shastana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus. 4: 46. USA: California, Mount Shasta. Syntypes (USNM), (BMNH); males.

similiana Clemens 1860 (*Poecilochroma*?); Proc. Acad. Nat. Sci. Philad. 12: 353. USA: Pennsylvania. Lectotype (ANSP); female.

similana Heinrich 1923 (*Eucosma dorsisignatana* ssp.); Bull. U.S. natn. Mus. 123: 121. misspelling of *similiana*; no type.

smithiana Walsingham 1895 (Paedisca); Trans. ent. Soc. Lond. 1895: 506. USA: Colorado. Lectotype (BMNH); male.

britana McDunnough 1927 (*Eucosma argentialbana* var.); Can. Ent. 59: 273. Canada: British Columbia, Seton Lake. Holotype (CNC); male.

snyderana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 89. USA: Idaho, Blackfoot. Holotype (AMNH); male.

sperryana McDunnough 1942 (*Eucosma*); Can. Ent. 74: 69. USA: Wyoming, Opal. Holotype (CNC); male.

sordicomana Staudinger 1859 (*Grapholitha*); Stettin. ent. Ztg. 20: 232. Spain: Chiclana. Lectotype (MNHU); female.

grammana Constant 1884 (*Grapholitha*); Annls Soc. ent. Fr. (6) 4: 213. France: Alpes Maritimes. Holotype (MNHN); unknown.

spaldingana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 19. USA: Utah, Stockton. Lectotype (AMNH); male.

suadana Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 130. USA: Utah, Vineyard. Holotype (USNM); male.

> ***aeana*** McDunnough 1942 (*Eucosma*); Can. Ent. 74: 68. USA: Utah, Fillmore. Holotype (CNC); male.

subflavana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus. 4: 48. USA: Oregon, Rouge River. Syntypes (BMNH); male.

subinvicta Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 33. USA: Arizona, Williams. Holotype (AMNH); male.

subtiliana Jackh 1960 (*Pseudeucosma*); Boll. Zool. agr. Bachic. (2) 3: 127. Italy: Rocciamelone, Piemonte. Holotype (USNM); unknown (lost).

succineana Kennel 1901 (*Epiblema*); Dt. ent. Z. Iris (1900) 13: 272. Uzbekistan. Syntype(s) (MNHU); unknown.

> *subditiva* Heinrich 1929 (*Eucosma tahoensis* ssp.); Proc. U.S. natn. Mus. 75: 9. USA: California, Lake Tahoe, Deer Park Springs. Holotype (USNM); male.

teleopa Razowski 2006 (*Pelochrista*); Acta Zool. Cracov. 49B: 129. India: Jammu and Kashmir (Indien J & K, Kaschmir, vic. Sonamarg). Holotype (ISEZ); male.

> *telopea* Razowski 2006 (*Pelochrista*); Acta Zool. Cracov. 49B: 129. misspelling of *teleopa*; no type.

tholera Falkovitsh 1964 (*Pelochrista*); Trud. Zool. Inst. Leningrad 34: 272. Kazakhstan. Holotype (ZMAS); male.

tibetana Caradja 1939 (*Epiblema*); Dt. ent. Z. Iris 53: 25. Tibet: Batang, Yangtze Valley. Lectotype (MGAB); male.

tolerans Meyrick 1930 (*Eucosma*); Exotic Microlepid. 3: 599. Tibet: Gyangtse. Holotype (BMNH); male.

tornimaculana Zerny 1935 (*Epiblema*); Mm. Soc. Sci. Nat. Maroc 42: 133. Morocco. Holotype (Unknown); unknown.

totana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 32. USA: Utah, Stockton. Lectotype (AMNH); male.

> ***spodias*** Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 35. unnecessary replacement name for totana; no type.

turiana Zerny 1927 (*Epiblema*); Eos 3: 470. Spain: Albarracin. Holotype (Unknown); male.

umbraculana Eversmann 1844 (*Teras*); Fauna Lepid. Volgo-Ural.: 519. Russia: Ural Plateau. Syntype(s) (ZMAS); unknown.

inignana Kennel 1901 (*Epiblema*); Dt. ent. Z. Iris 13 (1900): 283. China: Blagoweschtschensk. Holotype (MNHU); male.

iniquana Kennel 1921 (*Epiblema*); Palaear. Tortr.: 571. misspelling of inignana; no type.

subterminana Erschoff 1877 (*Grapholitha*); Horae Soc. ent. Ross 12: 342. Russia: Siberia, Irkutsk. Holotype (Unknown); male.

vagana McDunnough 1925 (*Eucosma*); Can. Ent. 57: 19. Canada: Manitoba, Aweme. Holotype (CNC); male.

wandana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 24. USA: Ohio, Cincinnati. Holotype (AMNH); male.

ustulatana Blanchard & Knudson 1983 (*Eucosma*); Proc. ent. Soc. Wash. 85: 849. USA: Texas, Washington Co., Brenham. Holotype (USNM); male.

uta Clarke 1953 (*Eucosma*); J. Wash. Acad. Sci. 43: 226. USA: Illinois, Putnam Co. Holotype (USNM); male.

eumaea Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 34. unnecessary replacement name for *wandana*; no type.

watertonana McDunnough 1925 (*Eucosma*); Can. Ent. 57: 115. Canada: Alberta, Waterton Lakes. Holotype (CNC); male.

williamsi Powell 1963 (*Eucosma*); Proc. Biol. Soc. Wash. 76: 237. USA: California, Alameda Co., Oakland Hills. Holotype (CAS); male.

womonana Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 88. USA: Ohio, Cincinnati. Lectotype (AMNH); male.

semnitis Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 35. unnecessary replacement name for *womonana*; no type.

zomonana Kearfott 1907 (*Eucosma*); Can. Ent. 39: 80. USA: Pennsylvania, New Brighton. Lectotype (AMNH); male.

explosa Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 36. unnecessary replacement name for *zomonana*; no type.

PHANETA

bimaculata Kuznetzov 1966 (*Astenodes*); Trud. Zool. Inst. Leningrad 37: 197. Russia: Far East, Primorsky Krai. Holotype (ZMAS); male.

pauperana Duponchel, in Godart 1842 (*Conchylis*); Hist. nat. Lpid. Papillons Fr. (Suppl.) 4: 170. France. Syntype(s) (MNHN); unknown.

SONIA

fulminana Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 50. USA: Texas. Holotype (BMNH); unknown. [wing venation and genitalia is consistent with *Sonia*]

ZEIRAPHERA

liturana Walsingham 1879 (*Steganoptycha*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus. 4: 71. USA: Oregon. Syntype(s) (BMNH); unknown. [genitalia is consistent with *Zeiraphera*]

EUCOSMINI UNPLACED

Eucosma? *abstemia* Meyrick 1932 (*Eucosma*); Exotic Microlepid. 4: 224. replacement name for *bactrana*; no type.

> *Eucosma*? *bactrana* Heinrich 1923 (*Eucosma*); Bull. U.S. natn. Mus. 123: 117. USA: Colorado, Silverton. (preoccupied) Holotype (USNM); male. [female unknown]

Eucosma? *albicostella* Turati & Kruger 1936 (*Semasia*); Mem. Soc. ent. Ital. 15: 75. Libya: Cyrenaica [Libya]. Syntypes(3) (Unknown); unknown. [unplaced according to Razowski 1999]

Eucosma? *aspidana* Walsingham 1884 (*Paedisca*); Trans. ent. Soc. Lond. 1884: 140. USA: Arizona. Holotype (BMNH); unknown. [female unknown]

Eucosma? *australis* Kuznetzov 1988 (*Eucosma*); Ent. Obozr. 67: 625. North Vietnam: Sonla Province, Naniu. Holotype (ZMAS) female. [female not examined]

Eucosma? ***autochthones*** Walsingham 1897 (*Thiodia*); Proc. Ent. Soc. Lond. 1897: 126. Virgin Islands: St. Thomas. Lectotype (MGAB); male. [outside Holarctic; transferred from *Phaneta*]

Eucosma? *bactromorpha* Diakonoff 1992 (*Eucosma*) Annls Soc. ent. Fr. (N.S.) 28: 38. Madagascar: Madagascar Centre (massif de l'Amkaratra, Manjakatompo). Holotype (MNHN); female. [outside Holarctic]

Eucosma? *betana* McDunnough 1942 (*Eucosma*); Can. Ent. 74: 68. Canada: Saskatchewan, Swift Current. Holotype (CNC); male. [female unknown]

Eucosma? *brigittana* Kennel 1919 (*Semasia*); Mitt. mnch. ent. Ges. 8: 83. Russia: Tannuola. Holotype (MNHU); unknown. [unplaced according to Razowski 1999]

Eucosma? *capnoleuca* Meyrick 1932 (*Eucosma*); Exotic Microlepid. 4: 223. Indonesia: Java. Syntypes (Unknown); male, female. [outside Holarctic]

Eucosma? *chlorobathra* Meyrick 1911 (*Eucosma*); Trans. Linn. Soc. Lond. 14: 268. Seychelles: Aldabra. Holotype (BMNH); female. [outside Holarctic]

Eucosma? *coniogramma* Clarke 1976 (*Eucosma*); Insects Micronesia 9: 53. Micronesia: Kusaie, Pukusrik. Holotype (USNM); male. [outside Holarctic]

> *Eucosma*? *okubiensis* Kawabe 1990 (*Eucosma*); Tinea 12: 141. Japan: Okinawa Island, Kin, Okukubi. Holotype (USNM); male.

Eucosma? *dapsilis* Heinrich 1929 (*Eucosma*); Proc. U.S. natn. Mus. 75: 5. USA: Wyoming, Yellowstone National Park. Holotype (USNM); male. [female unknown]

Eucosma? ***delphinoides*** Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 59. USA: Utah, Juab Co., Eureka. Holotype (USNM); male. [unusual male and female genitalia; transferred from *Phaneta*]

Eucosma? ***delphinus*** Heinrich 1923 (*Thiodia*); Bull. U.S. natn. Mus. 123: 45. USA: California, Lake Tahoe, Deer Park Springs. Holotype (USNM); male. [unusual male and female genitalia; transferred from *Phaneta*]

Eucosma? *directa* Meyrick 1912 (*Eucosma*); Exotic Microlepid. 1: 34. Turkey: Taurus Mountains. Holotype (BMNH); male. [unplaced according to Razowski 1999]

Eucosma? *disjectana* Kennel 1921 (*Epiblema*); Palaear. Tortr.: 558. Latvia: Livonia. Holotype (MNHU); unknown. [unplaced according to Razowski 1999]

Eucosma? *doiinthanonensis* Kawabe 1989 (*Eucosma*); Microlepid. Thailand 2: 61. Thailand: Chiang Mai Province, Doi Chiang Dao. Holotype (OPU); male. [outside Holarctic]

Eucosma? *eburata* Heinrich 1929 (*Eucosma*); Proc. U.S. natn. Mus. 75: 6. USA: Arizona, Mohave Co. Holotype (USNM); male. [female not examined]

Eucosma? *esmodes* Meyrick 1937 (*Eucosma*); Exotic Microlepid. 5: 99. Iraq: Diana. Holotype (BMNH); male. [unplaced according to Razowski 1999]

Eucosma? *fuscana* Kearfott 1907 (*Eucosma*); Can. Ent. 39: 53. Canada: Manitoba. Holotype (USNM); unknown (lost). [female unknown]

Eucosma? *giarabubensis* Turati 1930 (*Eucosma*); Atti Soc. ital. Sci. Nat. 69: 76. Libya: Cyrenaica. Holotype (Unknown); unknown. [unplaced according to Razowski 1999]

Eucosma? *gomonana* Kearfott 1907 (*Eucosma*); Can. Ent. 39: 78. USA: New Jersey, Essex County Park. Lectotype (AMNH); male. [unusual male and female genitalia]

> *Eucosma*? *discipula* Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 34. unnecessary replacement name for *gomonana*; no type.

Eucosma? *graziella* Blanchard 1968 (*Eucosma*); J. Lepid. Soc. 22: 143. USA: Texas, Brewster Co., Big Bend National Park, Green Gulch. Holotype (USNM); male. [female unknown]

Eucosma? *heinrichi* McDunnough 1925 (*Eucosma*); Can. Ent. 57: 19. Canada: Manitoba, Aweme. Holotype (CNC); male. [female not examined]

Eucosma? *hennei* Clarke 1947 (*Eucosma*); Bull. S. Calif. Acad. Sci 46: 51. USA: California, Los Angeles Co., El Segundo sand dunes. Holotype (USNM); male. [female not examined]

Eucosma? *iographa* Diakonoff 1968 (*Eucosma*); Bull. U.S. natn. Mus. 257 (1967): 88. Philippine Islands: Mindanao, Davao Province, La Lun Mountains, Calian. Holotype (CMNH); male. [outside Holarctic]

Eucosma? *jansei* Diakonoff 1956 (*Eucosma*); Proc. Konin. Neder. Akad. Weten. (C) 59: 640. replacement name for *inconspicua*; no type.

> *Eucosma*? *inconspicua* Diakonoff 1953 (*Eucosma*); Verh. Konin. Neder. Akad. Weten. (2) 49 (3): 156. New Guinea: Rattan Camp. (preoccupied) Holotype (RMNH); female. [outside Holarctic]

Eucosma? *jejunana* McDunnough 1942 (*Eucosma*); Can. Ent. 74: 67. Canada: Ontario, Blackburn. Holotype (CNC); male. [female unknown]

Eucosma? *jerusalemana* Amsel 1935 (*Epiblema*); Mitt. zool. Mus. Berl. 20: 291. Israel: Jerusalem. Holotype (LNK); unknown. [unplaced according to Razowski 1999]

Eucosma? *juncticiliana* Walsingham 1879 (*Paedisca*); Illust. typical Specimens Lepid. Heterocera Colln. Br. Mus 4: 75. USA: California, Shasta Co. Syntypes (BMNH), (AMNH); 12 males. [female not examined]

Eucosma? *kurdistana* Amsel 1959 (*Epiblema*); Bull. Soc. ent. Egypte 43: 58. Iraq: Haj Omran. Holotype (LNK); male. [unplaced according to Razowski 1999]

Eucosma? *lioplintha* Meyrick 1920 (*Eucosma*); Exotic Microlepid. 2: 344. India: Madras (Dindigul). Holotype (BMNH); male. [outside Holarctic]

Eucosma? *louisana* McDunnough 1944 (*Eucosma*); Can. Ent. 76: 154. Canada: Alberta, Lake Louise. Holotype (CNC); male. [female unknown]

Eucosma? *metagrapta* Meyrick 1932 (*Eucosma*); Exotic Microlepid. 4: 224. Indonesia: Java, teak-forest. Holotype (BMNH); male. [outside Holarctic]

Eucosma? *micropterana* Turati 1930 (*Epiblema*); Atti Soc. ital. Sci. Nat.. 69: 77. Libya: Cyrenaica [Libya]. Holotype (Unknown); unknown. [unplaced according to Razowski 1999]

Eucosma? *notanthes* Meyrick 1936 (*Eucosma*); Exotic Microlepid. 4: 610. Taiwan: Kagi. Holotype (BMNH); female. [female not examined]

Eucosma? *occipitana* Zeller 1875 (*Paedisca*); Verh. zool.-bot. Ges. Wien 25: 315. USA: Texas. Holotype (BMNH); unknown. [female unknown]

Eucosma? *ophionana* McDunnough 1925 (*Eucosma*); Can. Ent. 57: 18. Canada: Alberta, Nordegg. Holotype (CNC); male. [female not examined]

Eucosma? *pandana* Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 17. USA: Texas, Kerrville. Lectotype (AMNH); female. [female not examined]

Eucosma? *sardiopa* Meyrick 1912 (*Eucosma*); Ent. mon. Mag. 48: 34. unnecessary replacement name for pandana; no type.

Eucosma? *pedisignata* Diakonoff 1948 (*Eucosma*); Bull. Mus. natn. Hist. Nat. (2) 20 (4): 345. Vietnam: Tonkin, Cha-Pa. Syntypes (3) (MNHN); male. [outside Holarctic]

Eucosma? *persolita* Heinrich 1929 (*Eucosma*); Proc. U.S. natn. Mus. 75: 7. USA: Texas, Cameron Co., San Benito. Holotype (USNM); male. [female not examined]

Eucosma? *phaecochyta* Bradley 1965 (*Eucosma*); Ruwenzori Exped. 1952, 2 (12): 93. Zaire: Ruwenzori, Nyamaleju, East Africa. Holotype (BMNH); male. [outside Holarctic]

Eucosma? *plumbaginea* Meyrick 1932 (*Eucosma*); Exotic Microlepid. 4: 127. Uganda: Makindye. Holotype (BMNH); male. [outside Holarctic]

Eucosma? *rhymogramma* Meyrick 1916 (*Eucosma*); Exotic Microlepid. 2: 17. Myanmar: Upper Burma [Myanmar] (Myitkyina). Holotype (BMNH); female. [outside Holarctic]

Eucosma? *sierrae* Blanchard & Knudson 1983 (*Eucosma*); Proc. ent. Soc. Wash. 85: 850. USA: Texas, Culberson Co., Sierra Diablo Wildlife Management Area. Holotype (USNM); male. [female unknown]

Eucosma? *sphalerodes* Meyrick 1934 (*Eucosma*); Ann. Mag. nat. Hist. 14: 404. Africa: West Africa (Principe Island). Holotype (BMNH); male. [outside Holarctic]

Eucosma? *striatulana* Walsingham 1900 (*Tortrix*); Ann. Mag. nat. Hist. (7) 5: 456. China Hubei, Ichang. Holotype (BMNH); unknown. [unplaced according to Razowski 1999]

Eucosma? *tonitrualis* Meyrick 1934 (*Eucosma*); Ann. Mag. nat. Hist. (10) 14: 405. Africa: West Africa. Holotype (BMNH); unknown. [outside Holarctic]

Eucosma? *urnigera* Meyrick 1937 (*Eucosma*); Exotic Microlepid. 5: 158. Canada: NE Baffin Land, Ravenscraig Harbour. Syntypes (Unknown); male. [female unknown]

Eucosma? *vulpecularis* Meyrick 1932 (*Eucosma*); Trans. ent. Soc. Lond. 80: 111. Abyssinia [Ethiopia]: Djem-Djem Forest. Holotype (BMNH); male. [outside Holarctic]

Eucosma? *ursulana* Kennel 1919 (*Semasia*); Mitt. mnch. ent. Ges. 8: 85. Kazakhstan. Holotype (MNHU); unknown. [unplaced according to Razowski 1999]

Pelochrista? *dernina* Turati 1930 (*Epiblema*); Atti Soc. ital. Sci. Nat. 69: 76. Libya: Cyrenaica. Holotype (Unknown); unknown. [unplaced according to Razowski 1999]

Pelochrista? *praefractana* Kennel 1901 (*Epiblema*); Dt. ent. Z. Iris 13 (1900): 287. Russia: Amasia. Holotype (MNHU); female. [unplaced according to Razowski 1999]

Pelochrista? *vandana* Kearfott 1907 (*Eucosma*); Trans. Am. ent. Soc. 33: 24. USA: Florida, Hastings. Lectotype (AMNH); male. [female not examined]

Pelochrista? *pholas* Meyrick 1912 (Eucosma); Ent. mon. Mag. 48: 34. unnecessary replacement name for *vandana*; no type.