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IDENTIFICATION OF INSECTS AND DENSITY DETERMINATIONS OF THE STOMACH CONTENTS OF SMALL MAMMALS

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ABSTRACT

This report presents a microanalytical method for (1) insect identification in small mammal stomachs and (2) visual estimates of the stomach contents in terms of the relative densities of the insects positively identified, and the total plant:animal ratio. Identification of insects by their fragments was verified by T. O. Thatcher, CSU Entomologist; P. H. Baldwin, CSU Ornithologist; and use of reference vials of specific insect fragments. Stomachs used were of the thirteen-lined ground squirrel (*Spermophilus tridecemlineatus arenicola* Howell), Ord's kangaroo rat (*Dipodomys ordii luteolus* Goldman), northern grasshopper mouse (*Onychomys leucogaster arcticeps* Rhoads), prairie deer mouse (*Peromyscus maniculatus osgoodi* Mearns). These animals were trapped at the Pawnee National Grasslands and surrounding areas, located in northeast Colorado.

MATERIALS AND METHODS

Materials

Dissecting scope with zoom device (10X - 80X)

Illuminator and spotlight

Balance with one-tenth gram readings

Ethanol

200 mesh screen

Petri dishes (10mm - 15mm deep) (3)

Plastic sacks (app. 17cm x 8cm)

Spatula (8cm - 12cm wide)

Dissecting scissors

Tweezers (fine pointed) (2)

Teasing needles (2)

Envelopes (app. 15cm x 8cm)

Cards for recording insects and densities

Methods

A very limited amount of material is available concerning micro-analytical techniques for insect identification in stomachs. Harriss (1950) separated some plant tissue fragments from insect fragments by differences in the physical breakdown of their gross portions. He also noted that insect fragments can be distinguished by the absence of cell structure and the presence of varying degrees of translucence. Johnston (1967) preserved his stomachs for study in 95% ethanol solution, and dried his stomach contents at 105°F to obtain oven-dry weight.

In this procedure, the stomachs are removed from the animal and placed in plastic sacks containing an ethanol solution of 70% ethanol and 30% tap water. They remain in this condition until studied. The stomach is

opened along its margin with dissecting scissors, and the contents are deposited on filter paper, placed on a balance, weighed to the nearest one-tenth gram, and recorded. The contents are then placed in a shallow petri dish and sufficient tap water is added to enable the materials to move freely in the dish. In stomachs of larger volumn, the contents can be divided between two or more petri dishes until their density is adequate for sufficient light penetration to clearly distinguish individual insect fragments. If the mixture in the petri dish is not clear, the contents can be strained on a 200 mesh screen under tap water and deposited again in the petri dish.

The petri dish is then placed on a white background under a dissecting scope, preferably one having a zoom device for higher magnification of the more minute insect fragments. A dissecting scope with a zoom device of 10X - 80X ocular power was found satisfactory for this study. The insects are then identified and recorded on the appropriate cards. Tweezers and/or teasing needles are used to manipulate the insect fragments for positive identification. The Appendix contains photomicrographs of insect fragments occurring frequently in this study.

Relative densities of insects positively identified and densities of plant to animal were done by visual estimate, expressed as a volume percent. Five density ranges were used. Where "d" is the relative density;

Density Range	Rank
$20 > d > 0$	1
$40 > d > 20$	2
$60 > d > 40$	3
$80 > d > 60$	4
$100 > d > 80$	5

The rank numbers 1, 2, 3, 4, or 5 were then recorded for each different insect identification and for the plant:animal ratio.

In instances where stomach contents occupy more than one petri dish, the median of the density range can be used for individual estimates for each petri dish. The mean of the sum of the medians can then be found and its representative density range (rank) recorded. Example, using a Coleoptera adult:

	Dish #1	Dish #2	Dish #3
Rel. Density	2	4	0

$$\frac{\text{Medians}}{\text{No. of dishes}} = \frac{30 + 70 + 0}{3} = 33\ 1/3 \text{ (mean)}$$

Because 33 1/3% belongs to the 20% to 40% density range, represented by rank 2, a 2 is recorded for Coleoptera adult in this particular stomach.

After identifications and densities are recorded, the material is strained on the 200 mesh screen, and a spatula is used to place the material in a labeled envelope. The envelopes can then be placed in an oven at 55°C (131°F) for three days and the oven-dry weights of the stomach contents determined.

LITERATURE CITED

Harriss, K. L. 1950. Identification of insect contaminants of foods by the micromorphology of the insect fragments. J. Ass. Offic. Agr. Chem. 33:898.

Johnston, R. F. 1967. Food of the purple martin (*Progne subis*) in Kansas. Ibis 109:8-13.

Kurtz, O'Dean and Kenton L. Harriss. 196 . Microanalytical entomology for food sanitation control. J. Ass. Offic. Agr. Chem., Washington, D.C.

APPENDIX

Photomicrographs (35X)

I. Arachnida: main body

II. Arachnida: tarsus

III. Arachnida: tibia and tarsus

IV. Coleoptera adult: antenna

V. Coleoptera adult: femur, tibia, tarsus

VI. Coleoptera adult: Carabidae--maxilla

VII. Coleoptera adult: tibia

VIII. Coleoptera adult: wing

IX. Coleoptera larva: Scarabaeidae--leg

X. Coleoptera larva: Tenebrionidae--urogomphis

XI. Diptera adult: femur, tibia, portion of tarsus

XII. Diptera adult: robber fly--tarsus, claw

XIII. Homoptera: leafhopper *Cuerma costalis*--wing

XIV. Lepidoptera adult: leg

XV. Lepidoptera adult: palpus

XVI. Lepidoptera adult: tissue and hair

XVII. Lepidoptera larva: Noctuidae (Phalaenidae)--crochet

XVIII. Lepidoptera larva: Noctuidae (Phalaenidae)--tarsus

XIX. Lepidoptera larva: tarsus

XX. Lepidoptera larva: tarsus

XXI. Orthoptera: antenna

XXII. Orthoptera: camel cricket--Ceuthophilus--ovipositor

XXIII. Orthoptera: grasshopper--ovipositor

XXIV. Orthoptera: Portion of tibia and tarsus

XXV. Flea