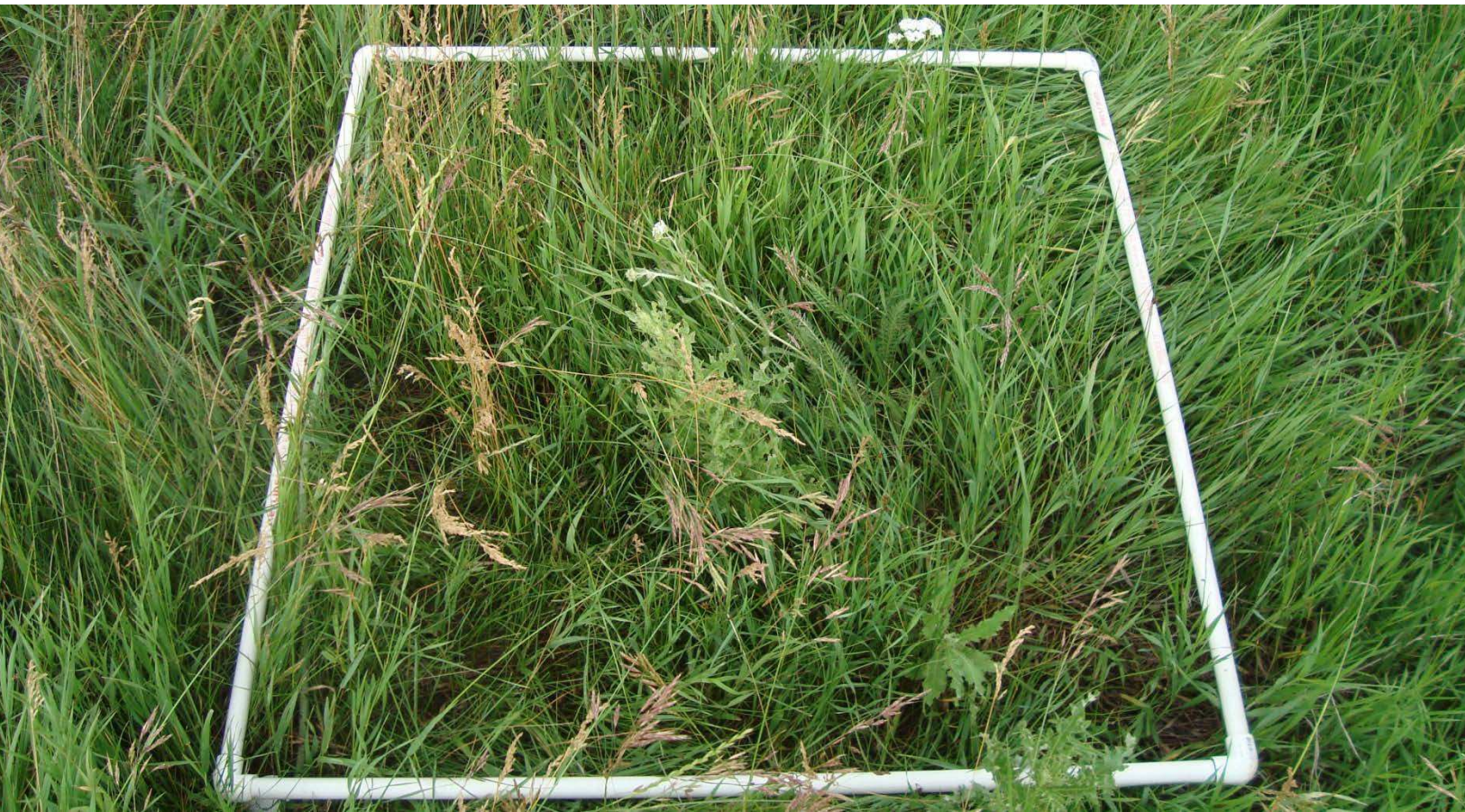


Rocky Mountain National Park



Vegetation Monitoring Division of Resource Stewardship

Project Design
Field Methods
Data Analysis
Reporting



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Rocky Mountain National Park Vegetation Monitoring Plan



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1 Introduction

Vegetation monitoring encompasses a broad spectrum of sampling approaches, ranging from simple species inventories to intensive data collection to answer questions about changes in vegetation composition and ecosystem processes over time. The type of monitoring practiced at Rocky Mountain National Park (RMNP) balances these approaches, allowing for the assessment of a variety of management projects. This monitoring plan provides guidance on how to design studies that are rigorous enough to provide useful data in a timely manner while addressing numerous projects and questions each field season.

The overarching vegetation monitoring objective in RMNP is to determine whether the goals of a particular revegetation or invasive plant management project were met and to use this information to modify and improve our techniques for future vegetation management projects. This method of using monitoring information to inform future management decisions is known as adaptive management (Figure 1). Monitoring and evaluation of revegetation projects can identify both strengths and weaknesses of management actions and allow for reevaluation and adjustments. This ensures that valuable time and resources are used effectively and efficiently in each field season. The purpose of this plan is to ensure revegetation and invasive plant species management in RMNP are consistently and efficiently monitored over successive field seasons. Specific focus is given to a selection of vegetation monitoring methods that are most useful for determining the success of vegetation management projects common in RMNP.

Other work groups within RMNP have well-developed monitoring plans that utilize specialized techniques and address issues related to ungulate impact monitoring and fire effects monitoring that will not be covered here.

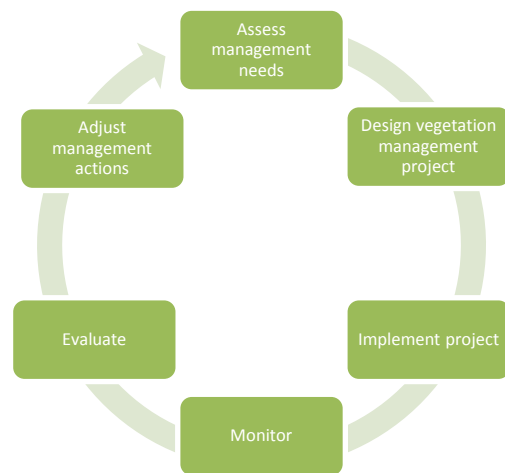


Figure 1. Conceptual diagram of the adaptive management process (Adapted from DOI Adaptive Management Technical Guide 2010).

2 Designing a Monitoring Study

2.1 Defining Study Objectives

Defining the objectives of a monitoring project is a critical component of designing a study to produce information that will inform management decisions. This section will suggest ways to identify what vegetation management projects should be monitored and how to identify what the objectives of a monitoring project are. Monitoring projects in RMNP will typically be conducted in active management sites, and project goals or management actions may change over the lifetime of a vegetation management project. Project objectives should be discussed throughout a field season to determine if project conditions or management objectives have changed.

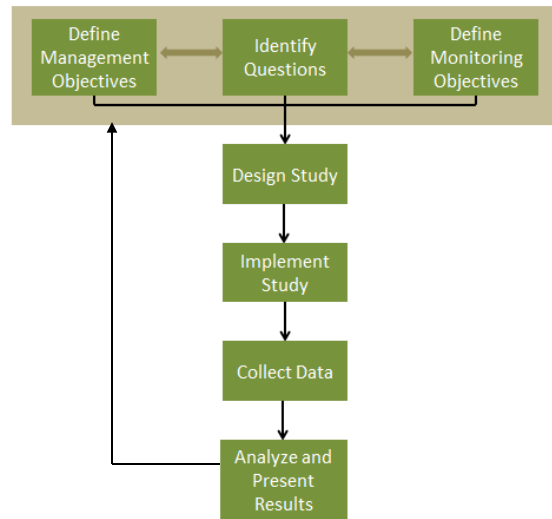


Figure 1. Monitoring project work flow

The objectives of a vegetation management project directly inform the objectives of a monitoring project because the purpose of monitoring is to assess whether management objectives have been met (Table 1).

Table 1. Examples of management objectives, questions and monitoring objectives used in designing a study.

Management Objective	Management Question	Monitoring Objective
Reduce cheatgrass density to <1 plant/m ² while avoiding damage to native plant species.	Does herbicide A effectively reduce cheatgrass to <1 plant/m ² while avoiding damage to native plant species?	Assess changes in native and exotic vegetation abundance (e.g. cover or density) in plots treated with herbicide A and control (untreated) plots.
Establish native plant species on roadside disturbances with cover >50% after 5 years.	Were the species/methods used effective in establishing native plant cover of >50% in 5 years?	Assess changes in planted species cover over 5 years.
Establish native plant species on erosion site with cover >50% and prevent exotic species cover from exceeding 5% after 5 years.	Were the species/methods used effective in establishing native plant cover of >50% and preventing exotics cover from exceeding 5% in 5 years?	Assess changes in planted and exotic species cover over 5 years.

The process of choosing a project, identifying questions, and identifying objectives can start with any one of those components and then progress to the others. For example, if the question about cheatgrass control in Table 1 was formed without a specific project in mind, an upcoming project with the appropriate management objectives could be identified as a candidate for monitoring.

Monitoring projects will increase in complexity as more objectives are added. If the objective of a project is to assess seedling density of seeded forbs on a restoration site, you may only collect data for seeded species. If, in addition to seeded species density, you are monitoring all unseeded volunteer species, the time you spend at each plot will increase substantially. Adding objectives can also increase the number of treatment types you include in your project. A study looking at the effects of an herbicide will require two plot types: plots treated with the herbicide and untreated control plots. If a native species planting component is added to the herbicide study, the study will require four plot types: treated/planted, treated/unplanted, untreated/planted, and untreated/unplanted. The importance of including untreated control plots in your study is discussed on [page 5](#).

Defining objectives clearly will help you design a study that is rigorous and efficient. Knowing that the objective of a study focuses on a single target plant, for example, rather than the entire plant community, will influence what methods are chosen and what data are collected for that project.

Discussing general vegetation management concerns as well as specific project-oriented questions with crew leads and supervisors before and during the study design process can ensure management's monitoring needs are being met.

2.2 Study Design

Once the objectives of the monitoring project have been identified, components of the field study can be selected.

Plot Location

Most of the monitoring studies that will assess vegetation management projects will be located in clearly defined project sites and because of this, locating appropriate project sites is relatively simple.

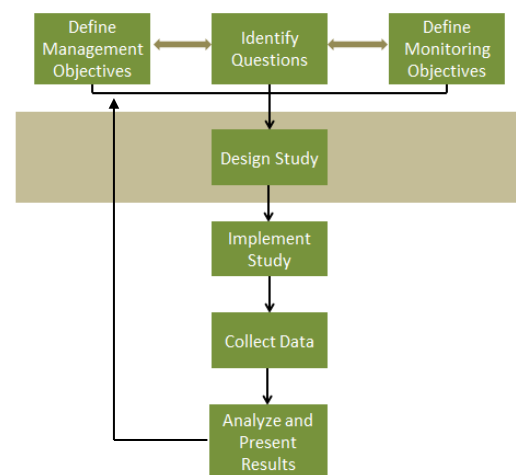


Figure 2. Monitoring project work flow

Wilderness Locations

One of the first things to consider when deciding where monitoring plots will be located is whether these plots will be within designated wilderness area, which comprises more than 95% of the land area of RMNP. Installing permanent plots in wilderness areas requires special permitting from the park Wilderness Coordinator (see *Research Permitting* below). Maps of the designated wilderness area of the park are available on RMNP's shared computer drive. Additionally, GPS coordinates collected at potential plot locations can be entered into a GIS map layer to determine whether they are located within wilderness boundaries. Consult with the RMNP GIS specialist to ensure accurate maps are generated. Detailed information about research permitting can be found on page 16.

Site Topography

Vegetation communities may vary along the environmental gradient of a large slope, making it important to orient transects perpendicular to the direction of a slope. For example, if a roadside project site is being monitored and there is an uphill slope away from the roadside, transects should run up the hill, rather than parallel to the road (Figure 3). The purpose of this orientation is to ensure that any variation due to changes in community composition along the gradient is captured within each transect or plot. If a transect is oriented parallel to the gradient, each transect will have very little variation within the sample, but variation between the transects will be very high (Figure 3).

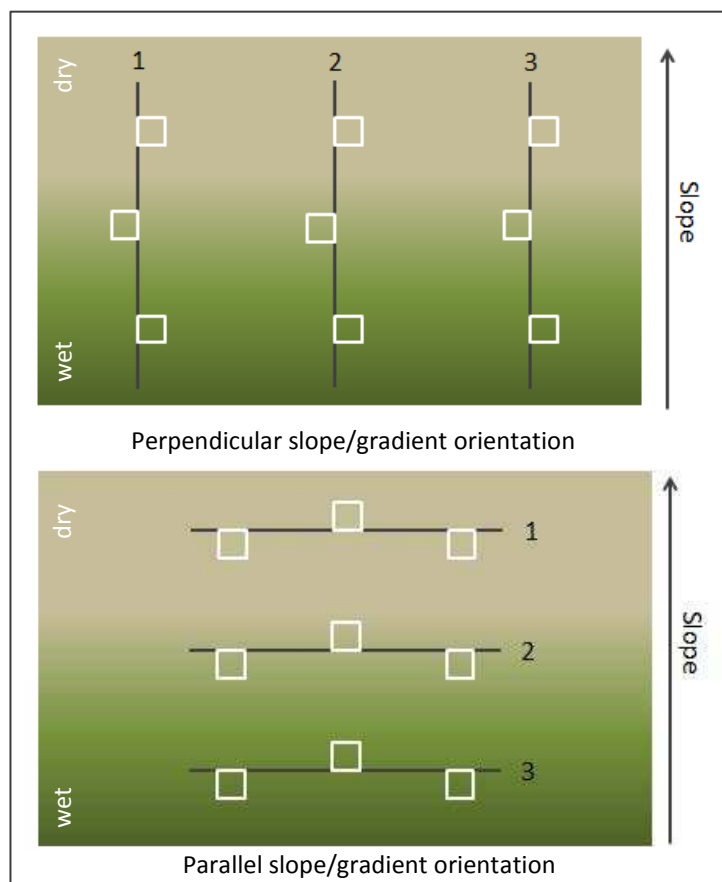


Figure 3. Comparing transect orientation relative to the direction of a slope or gradient

Using the illustrations in Figure 3 as an example, suppose transects 1-3 have different treatments applied to them. Transect 1 is treated at a high seeding rate, transect 2 is treated at a low seeding rate, and transect 3 is an unseeded control. The soil moisture content and plant abundance of this site decreases with increasing elevation along the

slope. When comparing the results of the seeding on this site, the perpendicular transects will all have similar within-transect vegetation variation resulting from the gradient in soil moisture. Differences detected in vegetation in these plots can be interpreted as most likely due to the effects of the seeding rates. On the other hand, the parallel transects do not capture the variation resulting from the moisture gradient, and data will indicate that the vegetation in each transect is different. This makes interpreting the effects of different seeding rates on this site difficult, and can lead to incorrectly inferring that high seeding rates result in reduced plant abundance.

If the objectives of your study are to assess treatment responses at different elevational or environmental gradients, then placing transects parallel to the slope or gradient would be appropriate if suitable control plots were also included.

Treatment and Control Plots

Untreated control plots should be included in a study whenever possible. Comparing untreated plots to treated plots gives you the ability to determine whether changes in vegetation are due to treatment effects or are the result of regular annual variation or another source of variation unrelated to the treatment. For example, if the effects of herbicide application were being monitored using only treatment plots and a significant post-treatment decline in native grass abundance was detected, this could be a response to herbicide exposure or due to normal interannual variation. It is unclear whether native grasses were less abundant due to a direct effect of the herbicide, or some unmeasured factor not related to herbicide application. If treatment and control plots were both monitored and native grass abundance decreased in both plot types, the results more clearly indicate that the effect was not due to herbicide application. Including untreated control plots accounts for the many unmeasured factors that can influence the vegetation communities being monitored. The conclusions made in the interpretation of your results will inspire far more confidence if there is a comparison between treated and untreated control plots.

It may not be possible to include untreated control plots for some projects; particularly those that are controlling highly invasive plant species where the spread of the invader into adjacent habitat is very likely. In this case, collecting pre-treatment data is useful in quantifying and interpreting vegetation changes in response to management actions and should be a priority for the first year of the monitoring study.

Monitoring studies will last between five and ten years; far longer than most vegetation management projects. Therefore it is important to communicate with crew leads and supervisors when monitoring studies that have untreated control plots are completed to determine whether treatment of those plots is needed.

Vegetation Attributes

This section briefly introduces vegetation attribute measurements that can be used when designing a monitoring study. Each of these attributes is described in greater detail later in this document and should be referred to when designing a new study.

Frequency

Frequency measurements are good indicators of general changes in plant communities, but if the objective of your study is to assess plant recruitment, mortality or vigor, you should consider using density or cover measurements. Frequency measurements are useful when monitoring perennial rhizomatous or mat-forming species growing amongst other species where other methods requiring individual plant identification would be difficult. Frequency is also useful for monitoring plant invasions. Collecting frequency data can be done rapidly and with minimal training because there are fewer decisions to be made compared to other attributes like density or cover.

See the frequency section for a detailed discussion of this attribute and its uses.

Density

Density measurements are useful when monitoring a variety of plant growth forms, especially those that are discreet and easily identified as single individuals. Measuring the density of sod-forming or rhizomatous species can be difficult because of ambiguities in identifying individuals amongst dense groups of plants. If the purpose of your study is to monitor these types of plants, the cover attribute may be better suited to your objectives.

Sampling density is appropriate in sites with sparse to moderately dense vegetation, but becomes very time-consuming at high plant densities. Density measurements are sensitive to changes in plant establishment and mortality, making this attribute useful for measuring the effects of management efforts that are intended to introduce (seeding and planting) or eliminate (invasive plant control) certain plant species from a site. Density measurements are not sensitive to changes in plant size or vigor.

See the density section for a detailed discussion of this attribute and its uses.

Cover

Cover measurements are useful for measuring most types of plants and are appropriate to use in sites with moderately sparse to very dense vegetation. Cover measurements can provide a clearer description of community composition than density or frequency because it equalizes measurements of large, uncommon species and small, abundant species.

Cover measurements are highly sensitive to changes in plant size or vigor and will detect short-term changes in plant species caused by annual variation in factors like climate.

Cover measurements can be used to assess all types of plant growth forms, and are especially useful for measuring very dense or mat-forming plants that would otherwise be impractical to measure using other attributes.

See the cover section for a detailed discussion of this attribute and its uses.

Plot Types

There are several plot types to choose from when designing a study, and each plot type can be used to measure a variety of vegetation attributes. When deciding what plot type is most appropriate for a project, a trip to the project site to collect site measurements, take pictures, and briefly assess site characteristics is recommended.

Single line and Baseline Transects

Simple and baseline transects are nearly identical except for how they are arranged on the monitoring site (Figure 4). A single line transect extends from one point to another and data are collected at regular intervals along that line (Figure 4). A baseline transect is a line transect that extends from a baseline installed on the monitoring site (Figure 4). There are typically several line transects extending from a baseline. Data are collected at regular intervals along each line transect on the baseline. Whether single line transects or baseline transects are more appropriate for a particular site will depend on things like site characteristics, treatment distributions, and target plant distributions. Single line transects are useful along long and narrow project sites, where baseline transects would be useful in larger project sites where treatments are applied across a broad portion of the study area. Also, single line transects can be used to strategically locate plots in sites where target species are clumped to ensure efficient data collection of these clumped species. Alternatively well distributed target species can be effectively monitored using baseline transects, since the sampling units are also well distributed within the plot.

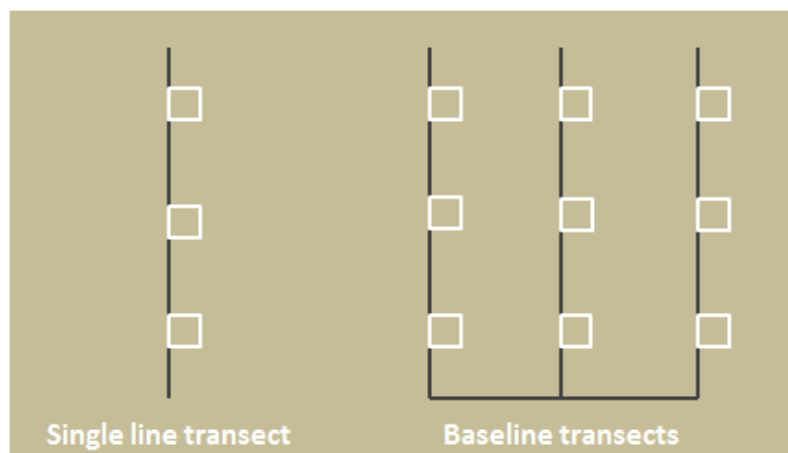


Figure 4. Comparing single line and baseline transects.

Single line and baseline transects can be used to collect frequency, density, or cover data using regularly spaced sampling

plots (quadrats). Cover data can also be collected along line and baseline transects using point-intercept and line-intercept methods. See the vegetation attributes section for more information about specific plant attribute measurements. Section 3 has detailed plot installation methods.

Circular Nested Plot

Circular nested vegetation monitoring plots (CNP) are used to collect cover and frequency data concurrently on project sites. A CNP covers a large area (168.2 m^2 [0.017 ha] or 1810 ft^2 [1/24 of an acre]), making it possible to efficiently monitor large study sites. Cover data are collected in three quadrats using cover-class data while frequency data (i.e. presence/absence) are collected in the remaining area of the CNP. See section 4 for detailed plot installation methods.

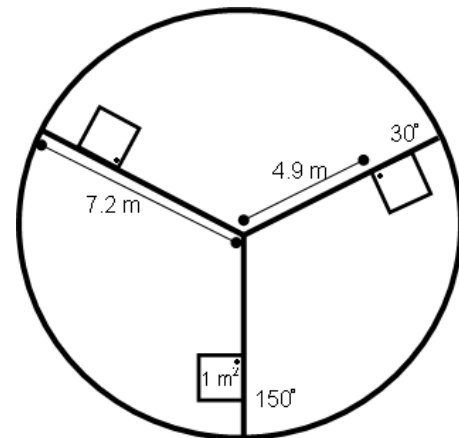


Figure 3. Diagram of a typical circular nested plot design.

This type of plot is appropriate for use in larger project sites where vegetation is distributed evenly throughout the site. If a target species or the vegetation on a site is very clumped, or is distributed along an environmental gradient, the wide spacing between quadrats in the CNP may lead to inadequate sampling of the target species or functional groups. Consider using single line or baseline transects in these cases to ensure cover data are collected efficiently.

Quadrats

Quadrats are used for all three vegetation measurement discusses in this manual and there are several considerations to make when choosing the type and size of quadrat to use for data collection.

Frequency

For frequency data, a general rule of thumb is that a target species should be found in no fewer than 20% of your quadrats and no more than 80% of your quadrats^[11]. This ensures that upward and downward trends in data are detectable. One way to address the need to sample plants at different scales is to use nested frequency quadrats (Figure 4). Data are first collected in the smaller quadrat which is nested within the larger quadrat. Additional species in the large quadrat that were not located in the small quadrat are then recorded, giving a sample at two scales in a single quadrat location. Data are summarized at both scales, and the appropriate scale (the scale that

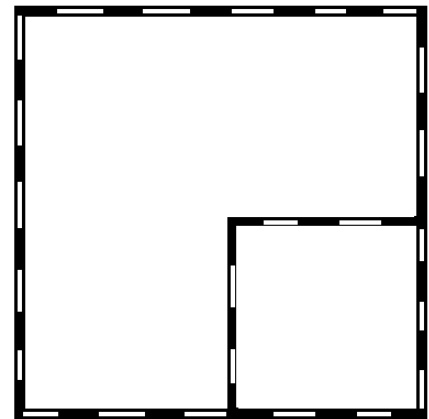


Figure 4. Diagram of a nested frequency plot.

samples the species at a frequency between 20% and 80%) for use in analysis is chosen. When selecting the quadrat size, it is important to be aware that frequency data for a species can only be compared between sites or studies when the quadrats used to collect species data are the same size. If comparing data between sites or studies is part of the study objectives, the same quadrat sizes should be used to collect data across all sites, because plant frequency is a function of quadrat size^[11]. See the frequency attribute in Section 3 for more details about frequency data, and Section 7 for more details about summarizing and analyzing frequency data.

Density

Quadrat size can affect both the efficiency and accuracy of density data collection. For example, when counting every individual of a very densely distributed target species in a 1 m² quadrat, it is some individuals will be missed and others counted more than once due to difficulties in keeping track of individuals at that scale. A smaller quadrat size would be much more efficient in this case. A brief amount of time collecting preliminary density data at the study site using different sized quadrats can inform a decision about the best choice. A rough estimate of the time needed to complete a density study can be made based on the time needed to collect these preliminary data at each quadrat and the number of quadrats that will be monitored at a site. Density data can be compared across sites and studies regardless of the quadrat size used since data are reported in absolute terms, so this is less of a concern compared to frequency quadrat size^[5]. See the density attribute in Section 3 for more details about density data and Section 7 for more details about summarizing and analyzing density data.

Cover

A 1 m² quadrat is recommended for cover-class data collection on both line transects and circular nested plots to avoid difficulties in estimating cover with larger quadrat sizes. See the cover attribute in Section 3 for more details about cover data, and Section 7 for more details about summarizing and analyzing cover data.

Plot Design

This section describes the final steps in designing the monitoring plots chosen for a study. Many of these decisions will depend on the characteristics of the study site and the objectives of a study itself.

Sample Size/Replication

The number of monitoring plots used for a vegetation monitoring project will affect the accuracy and precision of the data collected. Having several replications of monitoring plot data also allows for statistical analyses that measure differences in variation within data sets (e.g. *t*-tests & ANOVA); something not possible with a single replication. Greater numbers of plots also results in increased likelihood of detecting differences or changes in your data. Several methods of determining the number of sampling units needed to achieve a desired statistical power are available. For most vegetation

monitoring projects in RMNP, the number of monitoring plots installed will be a function of the size of project sites and the limited time available for data collection in the field, rather than a number of plots derived from sample size calculations. See *Measuring and Monitoring Plant Populations*^[5] pages 345-362 as well as *Statistical Power Analysis*^[3] for a thorough description of the various sample size and statistical power calculation methods available.

Every monitoring project should include, at minimum, three replications for each treatment type being monitored to allow for statistical analysis of the resulting data. For example, if a monitoring study design includes two different seeding treatments and control plots, there should be at least nine total monitoring plots installed to monitor this project; three for each treatment type. Three replications is the recommended *minimum*, and you are encouraged to install more plots as available field time and project space will allow. When prioritizing plot numbers between projects, greater numbers plots should be installed on project sites where a high degree of variation is observed during initial site assessments or if the objective of a study is to monitor a sparsely distributed or rare plant.

Plot Independence

Plot independence is the likelihood that one plot is not being influenced by processes or treatments in adjacent plots. Plots that are near enough to each other that they cannot be considered independent are more like subplots of a larger macroplot. The simplest way to ensure plot independence is to install plots with adequate distance between them. There are no strict guidelines to determine spacing that guarantees plot independence, so several standards are used in RMNPs vegetation monitoring program:

- CNPs are each considered an independent plot, and the quadrats contained within the CNP are non-independent subplots.
- Single-line transects are considered an independent plot, and the quadrats along the transect are non-independent subplots.
- Baseline transects are each considered independent plots placed along the baseline, and the quadrats along each transect are non-independent subplots

When monitoring more than one treatment type (including untreated control plots) it is preferable to have treatment types interspersed on a monitoring site, rather than grouped into separate locations of a site (Figure 5). In Figure 5, the site shown on the left has good interspersed of treatment and control plots. The example on the right shows plots placed in a way that can lead to pseudoreplication, where the T and C plots are more like subplots of a large macroplot^[9]. As described in the Site Topography section, the goal of having well interspersed plots is to avoid incorrectly concluding vegetation differences between plot types are due to treatment effects rather than attributes correlated with plot location.

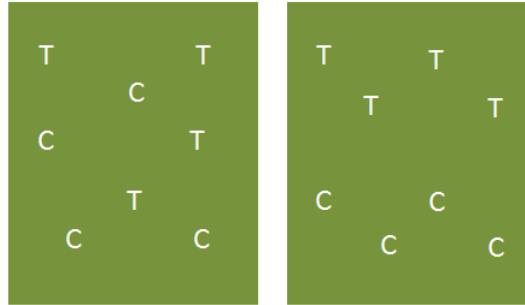


Figure 5. Examples of well-interspersed (left) and poorly interspersed (right) monitoring plots. T:treatment plot, C:control plot.

This complete interspersion of plots is not always possible due to the active management context in which these monitoring projects occur. Revegetation and exotic species control treatments often have predetermined application locations, and communicating objectives with supervisors and crew leads prior to implementation can help determine if it interspersed treatments will be possible. Also, randomly assigning treatments to plots may still produce poorly interspersed plots. If this happens, randomly reassign plots to achieve better interspersion.

Plot and Quadrat Spacing

The distance between quadrats and monitoring plots is another important consideration to make during study design. The ultimate goal of spacing plots and quadrats is to have sampling units distributed evenly throughout a study site. Once the appropriate spacing between quadrats and transects is determined, this spacing should be used systematically throughout the study site.

This section provides several recommendations for the *minimum* distance between quadrats and transects. Plot spacing should be adapted to each study based on the size and shape of the site and the number of plots and quadrats being installed to ensure adequate distribution of sampling units throughout the site.

Circular Nested Plots

Circular nested plots have predetermined quadrat placements within each plot (Section 2). Each CNP on a site should have one meter or more separating them, depending on available space.

Quadrats and Points Along Transects

When using quadrats placed along a transect, every quadrat should be placed at least one quadrat's length away from the next quadrat. For example, if a study is using a square 1 m² quadrat (1 m x 1 m), there should be at least a one meter interval between each quadrat along the transect. If using a rectangular quadrat, use the longer side of the quadrat as the minimum distance between quadrats along the transect. Similarly,

point-intercept points should be systematically placed at intervals along a transect. Points should be spaced far enough that they avoid resampling single individuals^[5].

Single-line and Baseline Transects

When using quadrats along baseline transects or closely spaced single-line transects, there should be at least one quadrat's distance between the quadrats of adjacent transects. A more systematic approach is to keep the distance between transects the same as the distance between quadrats along the transects, which can ensure an even distribution of sampling units across the site. If transects are being used to collect point- or line-intercept data, transects should be placed at least 1 m away from adjacent transects.

Randomization

The placement of plots and quadrats should be randomized to ensure that the data collected remains unbiased, and as a way of including another element of plot independence^[8]. Plot locations for most monitoring projects will be restricted to predesignated revegetation or exotics management areas, and there are some simple ways to include randomization in the placement of these plots and quadrats.

Treatments can be randomly assigned to each plot in a study. For example, a study is monitoring the effects of an herbicide on target and non-target species. If 20 monitoring plots are installed throughout the treatment area, the treatment that is applied to each plot (treated or untreated) can be randomly assigned so that there are 10 plots of each type. As discussed in the Plot Independence section, this type of interspersed randomization may be difficult to achieve for some types of projects where treatments are broadcast over large areas.

When determining quadrat or point placement along a transect, the first placement should be randomly chosen as a distance from the beginning of the transect. For example, a study is using 1 m² quadrats along a transect. There are 10 quadrats along this transect, with a spacing of three meters between each quadrat placement. To randomly determine where the first quadrat will be placed along this transect, a number between 0 and 3 is randomly chosen. If the number 2 is selected, the first quadrat is placed at the 2 m mark of the transect and the remaining quadrats are then placed three meters from each previous quadrat placement (5 m, 8 m, 11 m, etc.). The starting point for quadrat placement should be randomly determined for every transect.

A similar process should be followed to determine the placement of transects along a baseline. If 10 transects are being installed along a 100 m baseline at 10 m intervals, the first transect should be randomly placed at a point along the first 10 m of the baseline. The remaining transects should then be installed at 10 m intervals from the first transect.

Functional Groups

When data are collected for all plant species observed in a monitoring plot, it is often useful to organize plant species into functional groups to make analysis simpler and interpretation of your results easier to understand. A functional group can be defined by plant attributes such as growth form, native status, and other biological or ecological characteristics (e.g., species that fix nitrogen).

Functional groups can range from very broad (e.g., woody and non-woody species) to very specific (e.g., non-native invasive annual grass species). The functional groups that are used will be determined by the objectives of a study.

For vegetation management in RMNP, plants are typically organized into functional groups according to growth habit or morphology and their status as either a native, non-native invasive or non-native, non-invasive species (Table 2). Possible growth habit categories are tree, shrub, forb, or graminoid (grass, rush and sedge species) based on their classification in the USDA PLANTS database^[15]. Species are classified as native or non-native based upon their designation as such in Weber and Wittman's *Colorado Flora: Eastern Slope*^[16]. Species are classified as invasive if they are listed on RMNP's list of invasive species (available on RMNP's shared computer drive) or the Colorado Department of Agriculture's schedule of noxious weed^[2].

Table 2. Example of functional groups used to organize species data.

Native		Tree Shrub Forb Graminoid
Non-native	Non-invasive	Tree Shrub Forb Graminoid
	Invasive	Tree Shrub Forb Graminoid

Any other individual species or functional group of interest can be grouped separately depending on the study objectives. For example, if the effects of an herbicide that is used to control species of the family Asteraceae are being monitored, it would be useful to analyze native forbs and shrubs of the Asteraceae family separately from other species. Also, if it is not possible to reasonably collect data for separate species of the same functional group (e.g., interspersed native graminoids), collecting data for the functional group can reduce the time needed and likelihood of sampling errors. If functional group data are being collected in the field instead of species data, this should be a sampling rule that is recorded and used in all plots for that study (see *Sampling Rules* below). See Section 7 for more information about how to summarize and analyze species data that is organized into functional groups.

Sampling Rules

It is important to have a clearly defined sampling protocol that will be adhered to for the duration of a monitoring study before starting data collection. Establishing a study-specific sampling protocol ensures data are collected consistently across field seasons and with different monitoring personnel. Sampling rules will vary for each study and are different for each vegetation attribute. Recommended sampling rules for each attribute are described in detail in each attribute's respective portion of Section 3.

Site Attribute Data

Recording information about study site attributes in addition to measuring vegetation attributes provides information that is useful in describing site conditions when data are being analyzed and interpreted. Examples of site attribute data include: elevation, slope gradient, slope aspect, and soil information. During data analysis, these site attributes can be included as factors that contribute to the variation in the plant communities and species being monitored. As discussed above in *Plot Design*, whenever possible, it is preferable to minimize site variability between monitoring plots, unless the variation between monitoring plots and sites is a component of a study.

Measuring basic site attribute data can be done quickly by one person. Monitoring plot elevation, slope gradient, and slope aspect should be recorded for each monitoring plot to include in basic site descriptions. Elevation data can be obtained from your GPS unit, the slope gradient can be measured using a clinometer, and the slope aspect can be measured using a compass. A GPS unit and compass are standard equipment that will be carried with you in the field at all times and the Resources Stewardship Division at RMNP has several clinometers that can be loaned out as needed.

There are many other site attributes that can be measured, and the need to collect more than the basic site attribute data will depend on the objectives of a study.

Sampling Timing and Data Collection Regimes

Consistent Sampling Timing

When collecting vegetation over several years, it is important to be mindful of the timing of data collection from season to season. Collecting data during the same period of the growing season across years will ensure consistency in your data and avoid misinterpretation of phenological differences as changes due to management actions. Vegetation data collected in August one year and then in June the following year are likely to yield very different results due to differences in plant life cycles at those points in time. Ideally, data collection for a project would start when indicated by an attribute that is independent of a calendar date but indicative of plant phenology, such as the flowering of a particular species. Realistically, data collection field seasons in RMNP are short with very little down time, making it difficult to wait for exact environmental cues

for when to start data collection. To ensure all projects can be monitored completely and accurately, collecting data for a project during the same general timeframe as in previous seasons can serve as an appropriate proxy for phenology.

Seasonal Time Management

Creating a monitoring season timeline before starting data collection for the field season will help to successfully manage valuable field time. Projects that have been previously monitored will provide a rough idea of how many days or weeks are needed to complete those projects. New projects are more time consuming in their first year due to several factors, including study design time, plot installation time, and the time needed to become familiar with unfamiliar plant communities. There are typically two to three weeks available between completing seasonal training and the start of data collection to plan new projects and prepare for the field season. Comparing a new project to similar projects that have been monitored in previous seasons can provide a frame of reference for estimating the new project's data collection timeframe.

It is good practice to include one day in between projects that is not dedicated to data collection. This provides flexibility to accommodate unpredictable delays in data collection caused by things like inclement weather or difficulty relocating plots. There may be management projects where it is useful to be present during treatment applications to ensure that monitoring plots are correctly treated and control plots are left untreated as indicated by the study design. Coordinate with crew leads and allow time to accompany the respective crews on those days.

Monitoring Regime

Monitoring data can be collected either every year for five years, or over an extended period of time where data are collected in years one, two, three, five, and ten. The choice between the five year or ten year regime depends largely on the type of management project being monitored and the objectives of the study. In general, most herbicide monitoring projects should use the five year monitoring regime to assess the acute effects of herbicide use on target plants. If the objectives of a study are to track long-term recovery of a site restored after disturbance or effects of herbicide use or other methods of control, the ten year monitoring regime would be more appropriate. The regime used when monitoring revegetation projects is more flexible and will depend mostly on the objectives of the monitoring study, and whether short-term or long-term establishment is of greatest concern. Of course, if the need to monitor any project in both the short term and long term is identified, these regimes can be adapted to accommodate those needs.

Table 3. Comparison of five year and ten year data collection regimes. Shaded cells are data collection years. When possible, the first year of data collection should be pre-treatment data.

Data Collection Year	1	2	3	4	5	6	7	8	9	10
5 year regime										
10 year regime										

When using either regime, the first year of data collection should be pre-treatment data. In other words, if plots are going to be manipulated in some way, whether by herbicide application or revegetation, data from those plots should be collected prior to implementation of those treatments. Having pre-treatment data will allow comparison of treatment and control plots before treatment plots are manipulated. If vegetation is different between plot types before treatment, you will avoid incorrectly interpreting this difference as a response to the treatment in your post treatment data.

Communication with supervisors and crew leads about the goals of a monitoring project will aid in choosing the appropriate monitoring regime.

Research Permitting

You will need to submit a research permit to RMNP's Research Administrator for approval of permanent monitoring plot installation before implementing a new study. Installing permanent plots in wilderness areas requires special permitting from the park Wilderness Coordinator and can take longer than acquiring typical research permitting. The process of submitting a research permit for approval should be completed as soon as the details of a monitoring project are finalized. Coordinate with your supervisor to complete and submit a new research permit each year a new project is being implemented.

Safety Considerations

Once a monitoring study is fully designed, you will understand the work environment you will be entering for that project. The safety of you and your crew should be your top priority while working at RMNP. Vegetation management work can be difficult, requiring the lifting and carrying of heavy loads, use of sharp hand tools, working with and around large machinery, and the handling of chemicals and irritants. In addition to the physical demands of this work, the unique environmental conditions of RMNP create an inherent risk any time you are in the field. High elevation, exposed ridges, rapidly changing weather conditions, and rugged terrain expose workers to hazards that may be unique to some of them. Prior to the start of any seasonal field work, you will participate in extensive safety training. Job Hazard Analysis (JHA) descriptions are available to review before any work day begins. A JHA describes common workplace hazards you might encounter and provides recommendations on how to mitigate hazardous situations. You will be trained to use a variety of risk assessment models to evaluate a project and identify ways to mitigate potential risks and work safely every day. Take personal responsibility for everyone's safety, stop and think about potentially dangerous work situations, and if you feel like it may be unsafe, let your coworkers know and stop the activity to assess the situation as a group. Making safety the top priority when planning and executing field work will ensure a productive and enjoyable field season.

3 Vegetation Attributes

The following sections describe in detail the most common types of vegetation attributes that can be measured during a monitoring study.

3.1 Frequency

Applications and Limitations

Frequency of a plant species is the number of quadrats in which the species is present divided by the total number of quadrats in a plot or transect (whatever the experimental unit is) (hereafter, plot). An example of a monitoring plot with a target plant percent frequency of 40% is shown below (Figure 1).



Figure 1. Example of a transect measuring a target plant with a frequency of 40%.

Frequency measurements are most useful when monitoring perennial rhizomatous or mat-forming species growing amongst other species where other measurements would be difficult. Frequency is also useful for monitoring plant invasions. Frequency data are good indicators of general changes in plant communities, but if the objective of your study is to assess plant recruitment, mortality or vigor, you should consider using density or cover measurements. Frequency measurements do not detect changes in plant spatial distribution within the plot (e.g., did some individuals die and did others emerge?) or plant size or vigor, making it more difficult to interpret changes in frequency over time^[4].

Collecting frequency data can be done rapidly and with minimal training as there are fewer decisions to be made compared to other attributes like density or cover, making it useful for projects that need to be completed quickly.

Methods

Study Design

Frequency data are typically collected in regularly spaced quadrats placed along a transect. Alternatively, quadrats can be randomly located in a study site without using a transect, but there are several advantages to using transects. These advantages include more time-efficient plot installation, simplified relocation of individual quadrats in subsequent monitoring seasons, and the ability to accurately repeat measurements in quadrats along permanently installed transects (Section 2).

Transects can either extend from a baseline or be installed as a simple line transect (Section 2), and a 1m² quadrat is used to collect species presence data along the transect. The choice between baseline and simple line transects will depend on characteristics of the monitoring sites as well as your study objectives (Section 2). See Section 3 for transect installation instructions and section 4 for location recording instructions.

Boundary Rules

When collecting frequency data, you should know what criteria you will use to decide whether an individual plant is considered inside or outside your quadrat before going into the field.

For frequency data, if an herbaceous plant is rooted inside the area of the quadrat, that plant is included. Herbaceous plants that overhang a quadrat but are not rooted within the quadrat are excluded from that quadrat. Large woody plants such as shrubs and trees are included if they are rooted inside the quadrat or if they have overhanging canopies, or both.

Materials

- Blank field data sheets or tablet computer with data entry software
- Storage clipboard
- Pens/Permanent marker
- Measuring tape (meters)
- Rebar stakes
- Aluminum rebar caps
- Hammer
- Quadrat/measuring frame
- GPS unit
- Compass
- Specimen bags
- Plant identification materials (ID books, hand lens, dissecting kit)

Field Data Collection

For each transect, record the important site attributes (e.g. elevation or slope) which are determined during planning of the study (Section 2). If there are treatment and control plots, be sure this is easily identified on the data sheet.

Photograph each quadrat (Section 4) on the transect, and record what species are observed within the boundaries of that quadrat. Repeat this for all quadrats on the transect. Remember that only presence or absence is being recorded, not number of individuals. An example of a field data sheet for frequency plots can be found in the appendix .

Data Summary and Analysis

Data Entry

Data should be entered into an Excel spreadsheet following the columnar format example shown in Figure 2. Columns for additional attributes such as elevation or aspect can be added as defined by the individual study. A species “hit” for a quadrat should be indicated as a “1” in the respective cell.

	A	B	C	D	E	F	G	H	I	J	K	L
1	Baseline	Transect	Quadrat	Treatment	Slope	Bare	Litter	Moss/lichen	Rock	Bouteloua gracilis	Poa compressa	Muhlenbergia montana
2	1	1-1	1	herbicide	30	1	1	0	1	1	0	0
3	1	1-1	2	herbicide	30	1	1	0	1	0	1	1
4	1	1-2	1	herbicide	30	0	1	1	1	1	1	1
5	1	1-2	2	herbicide	30	1	1	0	1	1	1	0
6	2	2-1	1	control	15	1	1	0	1	1	0	1
7	2	2-1	2	control	15	1	1	0	1	1	0	1
8	2	2-2	1	control	15	0	1	1	1	0	1	1
9	2	2-2	2	control	15	1	1	0	1	1	1	0

Figure 2. Example of frequency data entry format in Excel. A “1” indicates species presence and a “0” indicates species absence.

Data Summary

To calculate the percent frequency of each species on a plot, add the number of hits observed for a species and divide by the total number of quadrats on that transect and multiply that number by 100.

After calculating the percent frequency of the individual species for each plot, the species frequency data can be grouped by functional group (Section 2). Percent frequency values for species in the functional group can be averaged to calculate the average percent frequency of the overall functional group.

The need to group species by functional group or some other grouping like seeded species or invasive target species will be determined by the unique questions being asked by the study.

Analysis and Figures

You can make comparisons of frequency data based on responses over time, or responses to treatment types or plot attributes like elevation or aspect, or a combination of any of these factors. One of the most powerful ways of presenting your data is in the form of a well-made and easily understood figure or graph. For a discussion of different analytical approaches and instructions on figure and table generation, see Section 7.

3.2 Density

Applications and Limitations

Density of a plant species is the number of individuals of a plant species in a defined area, and these measurements are typically expressed as the number of plants per unit-area (e.g. 4 plants/m², see Figure 1).



Figure 1. Example of a transect using 1 m² quadrats measuring a target plant with a density of 4 plants/m² (12 plants/3 m²).

Density measurements are useful when monitoring a variety of plant growth forms, especially those that are discreet and easily identified as single individuals. Sampling density is appropriate in sites with sparse to moderately dense vegetation, but becomes very time-consuming at high plant densities. Density measurements are sensitive to changes in plant establishment and mortality, making this attribute useful for measuring the effectiveness of management efforts like seeding or herbicide applications that are intended to introduce or eliminate certain plant species from a site, respectively. The data resulting from density measurements are reported in absolute unit-area measurements (e.g. plants/ha or plants/m²) making them easy to interpret and allowing for comparisons of data collected using various quadrat sizes^[5].

Density measurements are not sensitive to changes in plant size or vigor. As a result density is less likely to detect short-term changes in perennial plant species caused by annual variation in factors like climate or sub-lethal changes in vegetation caused by chemical or mechanical damage. Depending on the objectives of your study, this can either be desirable or undesirable. Measuring the density of sod-forming or rhizomatous species can be difficult because of ambiguities in identifying individuals amongst dense groups of plants. If the purpose of your study is focused on monitoring these types of plants, the cover attribute may be better suited to your objectives because identification of individuals is not necessary to estimate cover.

The time needed to collect density data is highly variable and depends on several factors including: plot and quadrat size, plant growth forms and maturity, and whether all species or only a subset or single species are being measured. Many of these study design considerations will be dictated by the objectives of the study (see Section 2). For example, if a study is focused on determining whether a target species is being killed by herbicide applications, it would be acceptable to only measure the density of the target species. On the other hand, if determining the effects of the herbicide on target as well

as non-target species is the focus, collecting density data for all species or a subset of herbicide-sensitive species would be needed.

Methods

Study Design

Density data are typically collected in regularly spaced quadrats placed along a transect. Alternatively, quadrats can be randomly located in a study site without using a transect, but there are several advantages to using transects. These advantages include more time-efficient plot installation, simplified relocation of individual quadrats in subsequent monitoring seasons, and the ability to accurately repeat measurements in quadrats along permanently installed transects (Section 2).

Transects can either extend from a baseline or be installed as a simple line transect (Section 2), and a 1 m^2 , 0.5 m^2 , or 0.1 m^2 quadrat is used to collect density data along the transect. The choice between baseline and simple line transects and quadrat sizes will depend on characteristics of the monitoring sites as well as your study objectives (Section 2). See section 3 for transect installation instructions and Section 4 for location recording instructions.

Boundary Rules

When collecting density data, you should know what criteria you will use to decide whether an individual plant is considered inside or outside your quadrat and how to decide whether a bunchy or clumped plant is a single plant before going into the field.

When a quadrat is positioned along a transect, some plants may either be touching or overlapping the boundary, but not be completely within the quadrat. For density data collection, it is important to have ground rules for deciding whether a plant that is not fully within a quadrat is considered in or out to avoid over- or underestimating species densities. The simplest unbiased way to address this problem is to count any plants touching the quadrat boundary along two adjacent sides as inside, and any plants touching the other two sides as outside the quadrat (Figure 2). This applies to both square and rectangular quadrats.

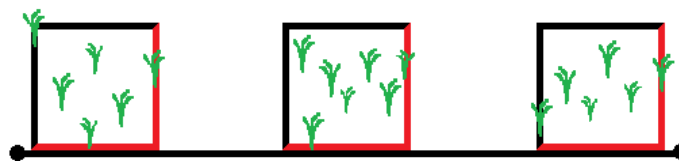


Figure 2. An example of a transect using 1 m^2 quadrats measuring a target plant and using boundary rules that include plants touching two adjacent sides of the quadrat (red) and exclude those that touch the other sides (black). This results in a density of 5.3 plants/m^2 ($16\text{ plants}/3\text{ m}^2$).

Boundary rules that are not as simple or reliable include: counting every other individual touching the boundary as inside or counting plants with more than 50% of their basal or canopy area inside the quadrat as inside.

Defining an Individual Plant

Determining what constitutes an individual plant for counting purposes may be difficult for some plant species; especially for those that are bunchy or rhizomatous. In the simplest cases, a plant will have an easily identified single rooted stem that is counted as one individual. In other cases a decision will have to be made about how a single individual is defined. This problem is most common with bunchy and mat-forming grass species. Typically, collecting bunchgrass data in RMNP is done by counting a single bunch as one individual. When measuring plant density in sites that have mat-forming or otherwise extremely dense grasses, consider using a small quadrat or collecting cover data instead of density data to increase sampling efficiency and reduce the likelihood of sampling errors.

It is important to maintain good records of how individuals were defined for difficult species. For each study be sure to indicate in the materials and methods section what boundary rules were used and how individual plants were identified for counting to ensure consistent data collection across seasons.

Materials

- Blank field data sheets or tablet computer with data entry software
- Storage clipboard
- Pens/Permanent marker
- Measuring tapes (meters)
- Rebar stakes
- Aluminum rebar caps
- Hammer
- Quadrat/measuring frame
- GPS unit
- Compass
- Specimen bags
- Plant identification materials (ID books, hand lens, dissecting kit)
- Toothpicks or small flags for marking already counted individuals

Field Data Collection

For each transect, record the important site attributes (e.g. elevation or slope), which are determined during planning of the study (Section 2). If there are treatment and control (untreated) plots, be sure this is easily identified on the data sheet.

Photograph each quadrat (Section 4) on the transect. Record all species or the target species that are observed within the boundaries of that quadrat. Count and record the number of individuals of each species inside the quadrat, remembering to apply the appropriate boundary decisions and counting rules to maintain sampling consistency. Repeat this for all quadrats on the transect. An example of a field data sheet for frequency plots can be found in the appendix.

Data Summary and Analysis

Data Entry

Enter data into an Excel spreadsheet following the columnar format example shown in Figure 3. Columns for additional attributes such as elevation or aspect can be added as defined by the individual study. At this stage you are simply entering in the raw counts you collected at each quadrat.

	A	B	C	D	E	F	G	H
1	Year	Baseline	Transect	Quadrat	Treatment	Rate	Species	Count
2	2013	1	1-1	1 Seeded	High		Bouteloua gracilis	13
3	2013	1	1-1	1 Seeded	High		Muhlenbergia montana	27
4	2013	1	1-1	1 Seeded	High		Thermopsis divaracarpa	22
5	2013	1	1-1	2 Seeded	High		Bouteloua gracilis	25
6	2013	1	1-1	2 Seeded	High		Muhlenbergia montana	14
7	2013	1	1-1	2 Seeded	High		Thermopsis divaracarpa	22
8	2013	2	2-1	1 Seeded	Low		Bouteloua gracilis	11
9	2013	2	2-1	1 Seeded	Low		Muhlenbergia montana	9
10	2013	2	2-1	1 Seeded	Low		Thermopsis divaracarpa	6
11	2013	2	2-1	2 Seeded	Low		Bouteloua gracilis	8
12	2013	2	2-1	2 Seeded	Low		Muhlenbergia montana	7
13	2013	2	2-1	2 Seeded	Low		Thermopsis divaracarpa	14

Figure 3. Example of density data entry format in Excel.

Data Summary

To calculate the mean density per quadrat of each species on a plot, divide the total count for that species by the total number of quadrats on that plot. In the example shown in Figure 3, *Bouteloua gracilis* was observed a total of 38 times in quadrats 1 and 2 along baseline 1, resulting in a mean density of $38/2 = 19$ plants/quadrat on baseline 1. (In this example baseline 1 and 2 only have a single transect for simplicity, but most baselines have many transects; See section 2).

Convert the density values from quadrat units to unit-area by dividing the mean density per quadrat by the area of the quadrat used to collect the density data. Using the example from Figure 3, assume we used 1 m² quadrats to collect the data.

$$\frac{19 \text{ plants}}{\text{quadrat}} \times \frac{1 \text{ quadrat}}{1 \text{ m}^2} = \frac{19 \text{ plants}}{1 \text{ m}^2} = 19 \text{ plants/m}^2$$

Converting density estimations to other units (e.g. ft², hectares or acres) will allow you to compare density data between studies or to interpret your results at a scale that is more meaningful to vegetation management personnel. All of these calculations are easily performed in Excel.

After calculating the density of the individual species for each plot, the species density data can be reorganized by functional group (Section 2). Density values for species in the functional group can be averaged to calculate the average density per unit-area of the overall functional group.

The need to group species by functional group or some other criteria like seeded species or invasive target species will be determined by the unique questions being asked by the study.

Analysis and Figures

You can make comparisons of density data based on responses over time, or responses to treatment types or plot attributes like elevation or aspect, or a combination of any of these factors. One of the most powerful ways of presenting your data is in the form of a well-made and easily understood figure or graph. For a discussion of different analytical approaches and instructions on data manipulation and figure and table generation, see Section 7.

3.3 Cover

Applications and Limitations

Cover of a plant species is defined as the percentage of an area that is covered by vegetation. Several different approaches to estimating vegetation cover based on both vegetation characteristics and monitoring plot designs are discussed in this section.

Cover measurements are useful for measuring most types of plants and are appropriate to use in sites with moderately sparse to very dense vegetation. There is no need to identify individual plants and there are very few boundary decisions when making cover measurements. Cover measurements can provide a clearer description of community composition than density or frequency because it equalizes measurements of both large, uncommon species and small, abundant species. Cover data are easily described as a percentage, making their presentation and interpretation more straightforward than frequency data.

Cover measurements are sensitive to changes in plant size or vigor and will detect short-term changes in plant species caused by annual variation in factors like climate or changes due to treatment effects. Canopy cover measurements (as opposed to basal cover) are especially sensitive to annual variation. Depending on the objectives of your study, this can either be desirable or undesirable. Cover measurements can be used to assess all types of plant growth forms, and are especially useful for measuring very dense or mat-forming plants that would be impractical to measure using other attributes.

The time needed to collect cover data is highly variable and depends on several factors including: plot size and type, plot number, the number of species that need to be identified in the field, and the experience level of the person estimating cover values. Many of these study design considerations will be dictated by the objectives of the study (Section 2).

Because cover measurements are highly sensitive to the size of a plant during a growing season, the yearly timing of data collection is very important. Ideally, data collection for a project is started when indicated by an attribute that is independent of a calendar date but indicative of plant phenology, such as the flowering of a particular species. Realistically, field seasons in RMNP are short, and to ensure all projects can be monitored, collecting data for a project during the same general timeframe as in previous seasons can serve as an appropriate proxy for phenology.

Types of Cover and Measurements

Types of Cover

Several types of cover can be measured. Canopy cover is the vertical projection of the outermost perimeter of a plant onto the ground, without making an effort to subtract canopy spaces or openings from the cover value. Foliar cover is similar to canopy cover measurements, but attempts to subtract canopy openings from the cover estimation and is not typically used at RMNP. Basal cover is the area of ground surface that is occupied by a plant's stem (Figure 1). Cover of non-living material such as rock, litter, and bare soil can also be estimated and may be important in assessing soil stability.

As shown in Figure 1, depending on the type of plant being measured, canopy and basal cover can provide two very different values for the same plant. This makes the consistent use of either canopy or basal cover across plots and field seasons a critical component of successfully measuring cover across seasons

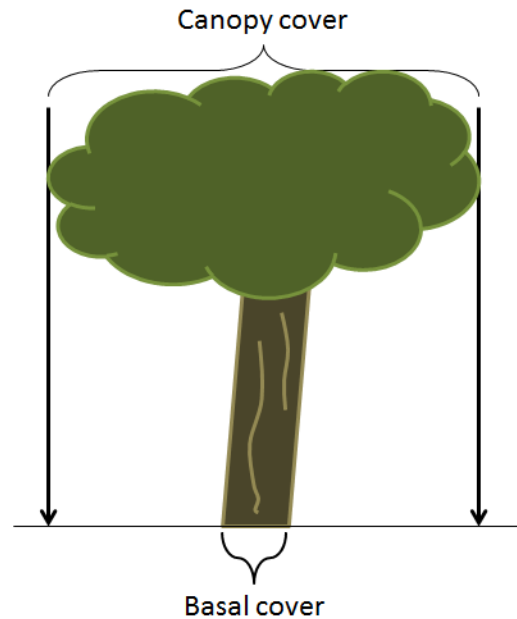


Figure 1. Comparison of canopy and basal cover.

It is recommended that tree, shrub, and forb cover be estimated using the canopy cover method. Most commonly encountered grasses in RMNP grow densely in tufts or bunches, making basal cover simpler to estimate than canopy cover, so it is recommended that grass cover be estimated using the basal cover method. Deciding where the basal area of a bunchgrass or mat of grasses begins and ends is something that should be considered when establishing ground rules for the study and is discussed later in this section.

There are two ways to present cover data for interpretation. Absolute cover is the proportion of ground surface covered by what was measured (plant, rock, litter, particular species, forbs, etc.). The sum of the absolute cover estimates of all species or functional groups can be greater than 100% due to overlapping cover measurements. Relative cover is the proportion of *total plant cover* represented by a species or functional group. The sum of the relative cover estimates for all species or functional groups is 100%. See the data summary and analysis section below for further discussion.

Cover Measurements

Cover-class Estimations

The most common method to measure vegetation cover in RMNP vegetation management projects uses cover-class estimations. Cover-class measurements allow for quick and consistent collection of cover data across monitoring seasons. Cover-classes eliminate the need to estimate an exact cover value, instead providing ranges of species cover (Table 1). Several cover-class methods have been developed, and most are similar in that the lowest and highest cover class values are broken into smaller intervals compared to the other values. A modified version of the Daubenmire cover-class intervals is used in RMNP to collect cover data (Table 1).

Figure 2 is an illustration of a quadrat with three different plant species occupying various amounts of area. Plant species A is represented by two large individuals. Looking at the two individuals we can see that while the two individuals combined take up more than 25% of the quadrat, they do not exceed 50%. Species A has a 10-25% cover value in this quadrat. Similarly, species B occupies 5-10% and species C occupies 3-5%.

Using the cover-class method requires a brief period of time to get comfortable with visually estimating cover, but once this is accomplished cover estimations can be collected rapidly. To make estimations, it is usually helpful to look at the species you are estimating for, and try to imagine what proportion of a quadrat would be occupied by that species if it was consolidated into one portion of the quadrat. In figure 2, exactly 25% of the quadrat is highlighted with a red box. If the canopy projections could be reshaped to fit within the 25% area, it looks unlikely it will be greater, but is clearly greater than the blue 10% box.

Marking the quadrat with 0.1 m graduations can help ensure accurate and consistent cover estimation, since 0.1 m² is equal to 1% of a 1 m² quadrat (Figure 2, Section 2).

Table 1. Modified Daubenmire cover-class values used to estimate cover in RMNP.

Modified Daubenmire Cover-classes
<1%
1-3%
3-5%
5-10%
10-25%
25-50%
50-75%
75-90%
90-95%
95-99%
100%

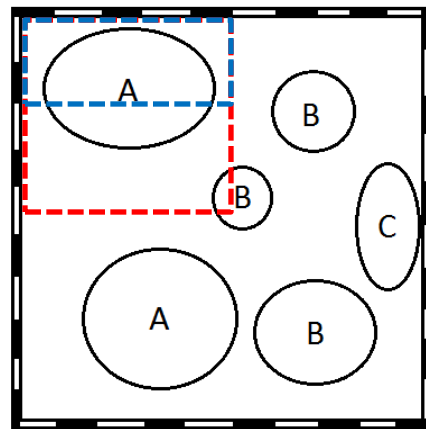


Figure 2. Example of a 1 m² quadrat with three species. Graduations are 0.1 m. The red dashed box is 25% of the area of the quadrat, and the blue box 10%..

Line Intercept and Point Intercept

The line intercept and point intercept methods are useful when measuring cover of very dense plant communities (e.g. alpine tundra) but may be less reliable when used to measure vegetation with more diffuse or less well-defined canopies. The point intercept method is also more likely to miss rare or dispersed individuals unless a high number of transects or points are used^[14].

The line intercept method involves measuring the distance along a transect that a plant's canopy intersects the transect. For example if a plant species canopy is observed along 5.3 m of a 10 m transect, that species has 53% canopy cover on that transect.

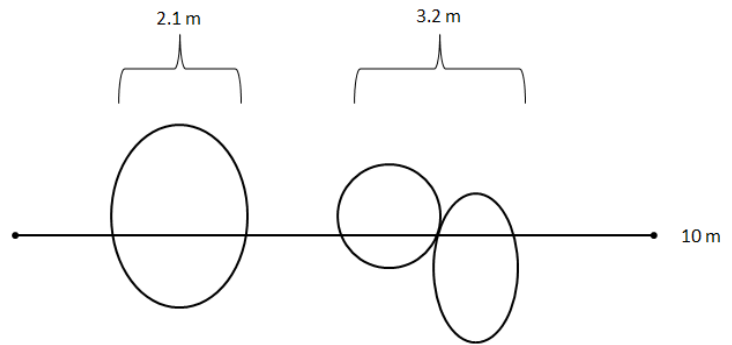


Figure 3. Example of a 10 m line intercept transect measuring a plant species with approximately 53% cover. The view is directly down onto the earth.

The line point method uses the number of points at regular intervals along a transect that coincide with each species or functional group to identify the proportion of points along that transect occupied. A rod or pin with a very sharp tip (to make the point as small as possible) is dropped at each point on the transect and the plants touched by the pin as it is lowered through the canopy are identified and counted as present. A species is only counted once at each point, even if multiple individuals touch the pin. Interval length is determined by site characteristics and size (See Section 2). The pin should be placed vertically at each interval to avoid overestimating cover (See methods in this section for more details). The cover value for a species is the number of times the species was observed divided by the total number of points along the transect. For example, a species that was observed at 5 intervals along a 10 m transect with 0.5 m intervals would have (5 hits/20 possible=0.25) 25% cover (Figure 4).



Figure 4. Example of a 10 m point intercept transect measuring a plant species with 25% cover.

Methods

Study Design

Cover data are collected in regularly spaced quadrats placed along a transect, along a line intercept transect, or along a point intercept transect. Alternatively, quadrats can be randomly located in a study site without using a transect, but there are several advantages to using transects. These advantages include more time-efficient plot installation, simplified relocation of monitoring plots in subsequent monitoring seasons, and the ability to accurately repeat measurements within quadrats placed along permanently installed plots (Section 2).

Transects can either be located within a circular nested plot or CNP (quadrats only), extend from a baseline (quadrats, line or point intercept), or be installed as a simple line transect (quadrats, line or point intercept) (Section 2). We've chosen to use a 1 m² quadrat to collect cover data along transects at RMNP for the purpose of cross-season monitoring consistency. The choice between CNPs, baseline transects and simple line transects will depend on characteristics of the monitoring sites as well as your study objectives (Section 2). See section 3 for transect installation instructions and section 4 for location recording instructions.

Ground Rules and Other Considerations

When collecting cover data, there are few boundary rules to be concerned with since individual plants are not being counted.

When collecting cover data for grasses using the basal cover method you may encounter some grasses that are more sparsely distributed than those commonly encountered (this is often the case with cheatgrass). These situations are where cover-class estimations are helpful in collecting accurate data because there is a range of possibilities for each class. It may be helpful to look at the patch of grass you are trying to estimate and then estimate how much space within that patch is not occupied by a grass stalk, or tiller, to decide what cover value you will assign. Sparse grass cover estimations can be difficult, but the extra time spent estimating cover carefully can ensure accurate and repeatable results.

Many bunch grasses grow radially outward from the center of the bunch over successive growing seasons. This leaves behind a dead and, in some cases, decaying center. If the center of the bunch grass is gray and decomposing it is most likely dead, and this portion of the grass should be included in the litter cover estimation while the green and living portion should be included in the grass cover estimation.

Some shrubs can appear partially dead or denuded on a portion of the plant while the remainder of the plant is green and vegetated. There are several ways to address how to estimate cover on plants like these, and the appropriate approach will depend on

your study objectives. For example, if you want to determine whether non-target plants are being weakened or damaged by herbicide, or what species of plants are thriving after being planted in a restoration site, it may be appropriate to only include the green parts of the shrub canopy cover for that species. The dead sections can then be included with litter estimates. Additionally, litter is typically defined as any dead material from the past year's growth, or persistent woody debris. Past studies at RMNP have typically used this approach. If the primary focus of a study is, for example, strictly on ground cover that will prevent erosion, this choice is less important.

Cover of moss and lichen species is only recorded when those species are found growing directly on the soil surface. Mosses and lichens growing on rock surfaces are not included in these estimates. If, for example, the objective of a study is to assess mosses or lichens specifically, these species should be included regardless of where they grow. Substrate material is included in the 'rock cover' measurement when the particles are larger than pea-sized. This is a rough generalization and rock cover can be broken down to include separate cover measurements of different sizes of substrate particles (e.g. gravel, cobble, small boulder cover) if the study objectives require these measurements. Substrate is included in the 'bare soil' measurement when the particles are smaller than pea-sized.

For each study be sure to indicate in the materials and methods section what ground rules were used to ensure consistent data collection across seasons.

Materials

- Blank field data sheets or tablet computer with data entry software
- Storage clipboard
- Pens/Permanent marker
- Measuring tapes (meters)
- Rebar stakes
- Aluminum rebar caps
- Hammer
- Quadrat/measuring frame
- GPS unit
- Compass
- Specimen bags
- Plant identification materials (ID books, hand lens, dissecting kit)
- 24 foot rope for CNP monitoring
- Long pin for point intercept monitoring
- Camera
- Dry erase board and marker

Field Data Collection

For each transect, record the important site attributes (e.g. elevation or slope) which are determined during planning of the study (Section 2). If there are treatment and control plots, be sure this is easily identified on the data sheet.

If using quadrats, photograph each quadrat (Section 4) on the transect. Record all species or the target species that are observed within the boundaries of that quadrat. Estimate the cover-class of each species inside the quadrat, remembering to apply the appropriate cover estimation methods to maintain sampling consistency. Repeat this for all quadrats on the transect (See Figure 2).

If using the line intercept method, photograph each transect along the length of the transect from the starting point of that transect (Section 4). Follow the length of the transect making note of any species whose canopy is intersected by the transect. Measure the length of the transect that the canopy occupies to the nearest centimeter. A break in the canopy that is greater than 1 cm should be considered the ending point for that portion of the canopy. Repeat this process for all species along the transect. (See Figure 3).

If using the point intercept method, photograph each transect along the length of the transect from the starting point of that transect (Section 4). At each interval along the transect drop a straight pin vertically down to the ground. It is important to orient the pin vertically to avoid overestimating cover. Record any plant species that is touching the vertical pin. A species is only recorded as a 'hit' once for each interval even if there are multiple individuals touching the pin. Repeat this process for all intervals along the transect. However, every species touched by the pin is counted as a 'hit.'

An example of field data sheets for cover plots can be found in the appendix.

Data Summary and Analysis

Absolute and Relative Cover

Absolute cover data are what species or functional groups occupy the proportion of the land area being monitored. The raw cover data that are collected in the field and entered into data sheets are absolute cover data. These data help identify changes in plant communities while taking into account changes in other important attributes like bare soil and litter cover important to informing management decisions. The sum of the absolute cover estimates for all species or functional groups may be greater than 100 percent.

Relative cover is the proportion of total vegetation cover comprised of a species or functional group. Relative cover data provide information about changes in a species or functional group relative to the entire plant community. These data help to characterize

changes in the species composition of plant communities but will not reflect community-wide variation due to factors like annual climate fluctuations that affect the entire plant community. For example, in an unusually dry year, the absolute cover of grass species is 25% lower than the previous season. All other vegetation groups (forbs, trees, and shrubs) experience a similar 25% decline in absolute cover. In this case, the relative cover of grasses will not change from the previous season, since the total proportion of vegetation cover occupied by grasses remained the same (Figure 5)

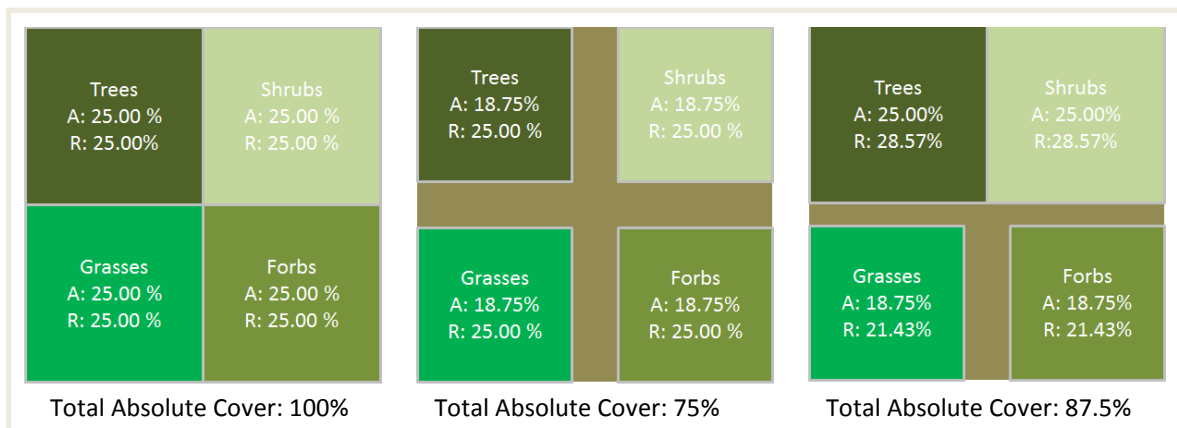


Figure 5. Effects of changing absolute cover (A) on relative cover values (R). Relative cover=Absolute Cover/Total absolute cover.

Whether absolute or relative cover data are presented depends on the objectives of your study. For example, in a study monitoring the success of planted native species on a restoration site, managers may be interested in site stabilization and how quickly these forbs are increasing in ground cover on a site. In this case, absolute cover should be used to describe the changes in forb cover because this measure is in absolute terms of what percentage of ground is covered with vegetation. Relative would be useful in this case to compare changes in community composition of those planted species over time. Relative cover can also be useful for explaining unusual or unexpected results. A 25% decline in percent cover of native grasses in treatment plots may be cause for concern, but if relative cover has not changed, and a similar decline was observed in control plots, it is more likely the effect was not caused by the treatment.

Data Entry

Data should be entered into an Excel spreadsheet following the columnar format example shown in Tables 5-8. Columns for additional attributes such as elevation or aspect can be added as defined by the individual study. At this stage you are simply entering in the raw absolute cover data you collected at each quadrat. These examples only show vegetation data, but bare ground, litter, and rock cover data will also be entered.

When entering cover-class quadrat data, the midpoints for the cover-classes are entered as the cover value. For example, if *Bouteloua gracilis* was estimated to have a

cover-class value of 25-50%, the absolute cover value for *B. gracilis* entered for that quadrat would be $(25+50/2)$ 37.5%.

When entering data from CNPs, there will also be frequency data with no cover value associated. These data are entered the same way the cover data are entered, the quadrat is listed as 'entire', and the cover value cell is left empty (Table 6). These data can be used for species richness calculations.

Table 5. Example of transect quadrat cover data entry format in Excel.

	A	B	C	D	E	F	G
1	Year	Baseline	Transect	Quadrat	Treatment	Species	Cover %
2	2013	1	1	1	Herbicide	<i>Bromus tectorum</i>	0.5
3	2013	1	1	1	Herbicide	<i>Bouteloua gracilis</i>	17.5
4	2013	1	1	1	Herbicide	<i>Helianthus pumilus</i>	12.5
5	2013	1	1	1	Herbicide	<i>Penstemon virens</i>	7.5
6	2013	1	1	1	Herbicide	<i>Thermopsis divaricarpa</i>	12.5
7	2013	1	1	1	Herbicide	<i>Acer glabrum</i>	37.5
8	2013	1	1	2	Herbicide	<i>Bromus tectorum</i>	2
9	2013	1	1	2	Herbicide	<i>Hesperostipa comata</i>	12.5
10	2013	1	1	2	Herbicide	<i>Bouteloua gracilis</i>	12.5
11	2013	1	1	2	Herbicide	<i>Penstemon virens</i>	4
12	2013	1	1	3	Herbicide	<i>Bromus tectorum</i>	0.5
13	2013	1	1	4	Herbicide	<i>Bromus tectorum</i>	2

Table 6. Example of circular nested plot quadrat cover data entry format in Excel.

	A	B	C	D	E	F
1	Year	CNP Name	Quadrat	Treatment	Species	Cover %
2	2013	BLR10	1	Herbicide	<i>Bromus tectorum</i>	0.5
3	2013	BLR10	1	Herbicide	<i>Bouteloua gracilis</i>	17.5
4	2013	BLR10	1	Herbicide	<i>Helianthus pumilus</i>	12.5
5	2013	BLR10	1	Herbicide	<i>Penstemon virens</i>	7.5
6	2013	BLR10	1	Herbicide	<i>Thermopsis divaricarpa</i>	12.5
7	2013	BLR10	1	Herbicide	<i>Acer glabrum</i>	37.5
8	2013	BLR10	2	Herbicide	<i>Bromus tectorum</i>	2
9	2013	BLR10	2	Herbicide	<i>Hesperostipa comata</i>	12.5
10	2013	BLR10	2	Herbicide	<i>Bouteloua gracilis</i>	12.5
11	2013	BLR10	2	Herbicide	<i>Penstemon virens</i>	4
12	2013	BLR10	3	Herbicide	<i>Bromus tectorum</i>	0.5
13	2013	BLR10	entire	Herbicide	<i>Pediocactus simpsonii</i>	

When entering line intercept data, enter the total length of the transect each species occupied for each transect. Dividing each species' value by the total length of the transect and multiplying by 100 gives the absolute cover value for that species.

Table 7. Example of line intercept cover data entry format in Excel.

	A	B	C	D	E	F	G	H
1	Year	Baseline	Transect	Treatment	Species	Length cm	Total cm	% Cover (length/total*100)
2	2013	1	1-1	Herbicide	<i>Bromus tectorum</i>	7	1000	0.70%
3	2013	1	1-1	Herbicide	<i>Bouteloua gracilis</i>	125	1000	12.50%
4	2013	1	1-1	Herbicide	<i>Helianthus pumilis</i>	58	1000	5.80%
5	2013	1	1-2	Herbicide	<i>Bromus tectorum</i>	4	1000	0.40%
6	2013	1	1-2	Herbicide	<i>Bouteloua gracilis</i>	77	1000	7.70%
7	2013	1	1-2	Herbicide	<i>Helianthus pumilis</i>	145	1000	14.50%
8	2013	2	2-1	Control	<i>Bromus tectorum</i>	20	1000	2.00%
9	2013	2	2-1	Control	<i>Bouteloua gracilis</i>	63	1000	6.30%
10	2013	2	2-1	Control	<i>Helianthus pumilis</i>	64	1000	6.40%
11	2013	2	2-2	Control	<i>Bromus tectorum</i>	18	1000	1.80%
12	2013	2	2-2	Control	<i>Bouteloua gracilis</i>	44	1000	4.40%
13	2013	2	2-2	Control	<i>Helianthus pumilis</i>	90	1000	9.00%

When entering point intercept data, enter the total number of hits for each species along each transect. Dividing each species' value by the total number of points along the transect and multiplying by 100 gives the absolute cover value for that species.

Table 8. Example of point intercept cover data entry format in Excel.

	A	B	C	D	E	F	G	H
1	Year	Baseline	Transect	Treatment	Species	Hits	Total points	% Cover (hits/total*100)
2	2013	1	1-1	Herbicide	<i>Bromus tectorum</i>	1	100	1.00%
3	2013	1	1-1	Herbicide	<i>Bouteloua gracilis</i>	30	100	30.00%
4	2013	1	1-1	Herbicide	<i>Helianthus pumilis</i>	44	100	44.00%
5	2013	1	1-2	Herbicide	<i>Bromus tectorum</i>	2	100	2.00%
6	2013	1	1-2	Herbicide	<i>Bouteloua gracilis</i>	77	100	77.00%
7	2013	1	1-2	Herbicide	<i>Helianthus pumilis</i>	45	100	45.00%
8	2013	2	2-1	Control	<i>Bromus tectorum</i>	4	100	4.00%
9	2013	2	2-1	Control	<i>Bouteloua gracilis</i>	63	100	63.00%
10	2013	2	2-1	Control	<i>Helianthus pumilis</i>	64	100	64.00%
11	2013	2	2-2	Control	<i>Bromus tectorum</i>	18	100	18.00%
12	2013	2	2-2	Control	<i>Bouteloua gracilis</i>	44	100	44.00%
13	2013	2	2-2	Control	<i>Helianthus pumilis</i>	30	100	30.00%

Data Summary and Absolute and Relative Cover

Quadrat data are summarized within a transect or within a CNP (Section 7), if those plots can be considered independent (Section 2). To calculate the mean absolute cover per for species on a transect or in a CNP, divide the total absolute cover for that species by the total number of quadrats on that plot. In the example shown in Figure 5, *Bromus tectorum* had a total absolute cover of 5% in four quadrats on transect 1. This results in a mean absolute cover of $5\%/4 = 1.25\%$ on transect 1. This process is repeated for each

species observed in a monitoring plot. (In these examples the data are simplified, but most transects will have more quadrats and species within the quadrats ; See Section 2).

To calculate relative cover plot values for a species, divide a species' mean absolute cover value for that plot by the mean absolute cover of all vegetation on that plot. For example, if *Bromus tectorum* had a mean absolute cover of 1.25% and the mean total vegetation cover for that plot was 75%, the relative cover of *Bouteloua gracilis* would be $(1.25\%/75\%) = 1.67\%$ relative cover.

After calculating the cover values of the individual species for each plot, the species cover data can be reorganized by functional group (Section 2). Cover values for species in a functional group can be averaged to calculate the average absolute or relative cover of the overall functional group.

The need to group species by functional group or some other grouping like seeded species or invasive target species will be determined by the unique questions being asked by the study.

Analysis and Figures

You can make comparisons of cover data based on responses over time, or responses to treatment types or plot attributes like elevation or aspect, or a combination of any of these factors. One of the most powerful ways of presenting your data is in the form of a well-made and easily understood figure or graph. For a discussion of different analytical approaches and instructions on data manipulation and figure and table generation, see section 7.

4 Monitoring Plot Installation

4.1 Installing and Marking Plots

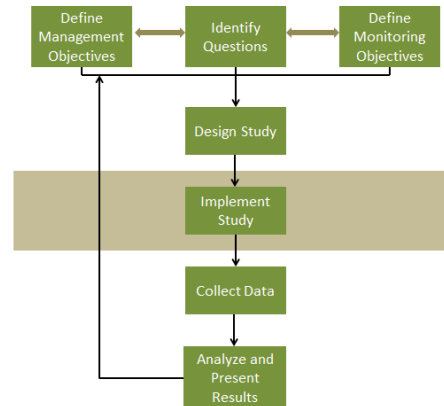


Figure 2. Monitoring project work flow

The process to install both permanent single line and baseline transects is similar, with the exception of including the installation of a baseline for the baseline transects. To install a baseline or single line transect, extend a measuring tape to the predetermined length of the baseline or single line transect. At each end of the measuring tape, hammer a 1-2 ft length of rebar into the ground until approximately 5 cm (2 in) is protruding. For baseline transects, additional rebar stakes are hammered in at the end of the transect opposite the baseline. Additional stakes are not needed for transects along the baseline because transects can be relocated using the measuring tape (Figure 1). Record the compass bearing of single line transects and baselines so that in the event one stake is lost, a new stake can be installed, eliminating the need to abandon the plot. Aluminum caps stamped with an indication that the stakes are for vegetation monitoring purposes should be placed on the top of the protruding ends of the rebar.

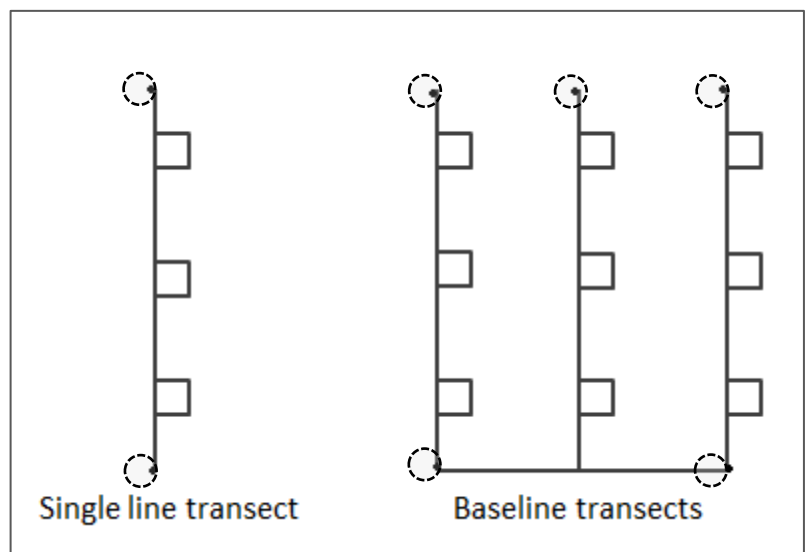


Figure 3. Commonly used line plot types used to monitoring vegetation in Rocky Mountain National Park. Dashed circles indicate placement of rebar stakes in permanent plots.

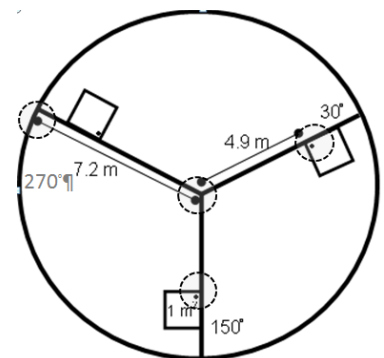


Figure 3. Circular nested plot. Dashed circles indicate placement of rebar stakes in permanent plots.

A circular nested plot is a circular plot with three, 7.2 m (24 ft) spokes at 30, 150 and 270 degrees from the center of the plot. A 1 m² quadrat is located on each of these spokes 4.9

m (16 ft) from the center of the plot (Figure 2). Circular nested plots are installed by hammering a 30 – 60 cm (1-2 ft) length of rebar into the ground at the center point of the CNP and at three spokes within the circle. To determine placement of the spokes, a 7.2 m length of rope is secured to the center stake. A knot tied in the rope at 4.9 m from the center stake allows for the consistent location of the stake placement for each spoke. To orient the first spoke at the 30° azimuth, one person stands over the center stake of the CNP and uses a magnetic compass to determine when the rope that is secured to the center stake is aligned with the compass bearing of 30°. When the rope is correctly positioned, hammer a 30 – 60 cm (1-2 ft) length of rebar into the ground 4.9 m from the center stake (at the knot). This process is repeated for the 150° and 270° azimuths. Aluminum caps stamped with an indication that the stakes are for vegetation monitoring purposes should be placed on the top of the protruding ends of the rebar.

4.2 Relocating Plots

At each plot, GPS coordinates should be recorded. A handheld GPS unit can then be used to locate the permanent plot (Section 4). In some cases, relocating the rebar stakes is very difficult due to dense and high vegetation, or muddy soils. A metal detector can be used to relocate lost or buried rebar stakes.

If a stake is lost, it is usually possible to install a new stake in its place by using compass bearings and measuring the distance from the other stakes that were relocated in the plot.

Materials

- Measuring tape (meters)
- Rope section (7.2 m w/ knot at 4.9 m) for CNPs
- Rebar stakes (30-60 cm)
- Aluminum rebar caps
- Hammer or Mallet
- GPS unit
- Compass
- Random number table or generator
- Camera

4.3 Coordinating with other Crews

At this point of the project, either the monitoring crew or the exotics control or revegetation crews may apply any treatments that are part of the monitoring study. It is important to communicate with crew leads and supervisors about timing and location of these treatments to ensure the study is implemented as intended.

5 Data Collection

In addition to collecting vegetation data for a project, you will also collect site attribute data, sampling unit photographs, and GPS coordinates (if not already completed). Site attribute data, photographs, and GPS coordinates should be recorded first, because it is common to forget these important steps once vegetation data collection is completed.

5.1 Photographing Plots

The utility in having a photographic record of your monitoring plots is twofold. First, annual photographs of a sampling unit can provide useful visual information about changes taking place over the life of the monitoring project. They are especially useful when monitoring personnel changes from season to season. For example, if an unexpected change in vegetation occurs in a plot, you may be unsure of the cause because you did not collect data that year. A quick check of the photograph for that plot may reveal that excessive erosion has removed a portion of the vegetation.

Photographs can also be very useful when relocating plots from previous monitoring seasons. Finally, pre- and post-treatment photographs can be used to effectively illustrate changes that you are presenting in a monitoring report.

Each photograph should show the name of the plot or transect, the name of the individual sampling unit shown, and the data the photograph was taken (Table 1).

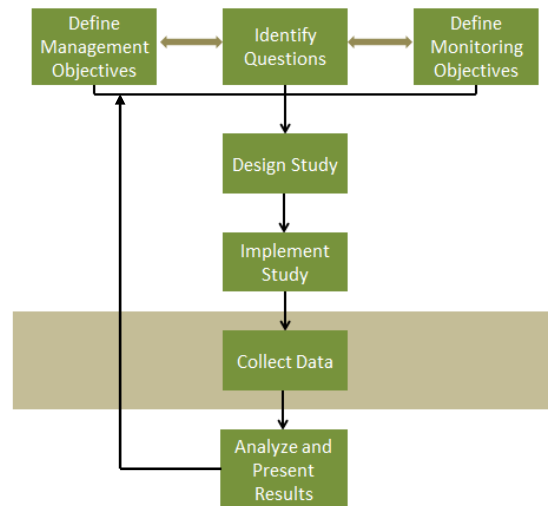


Figure 4. Monitoring project work flow

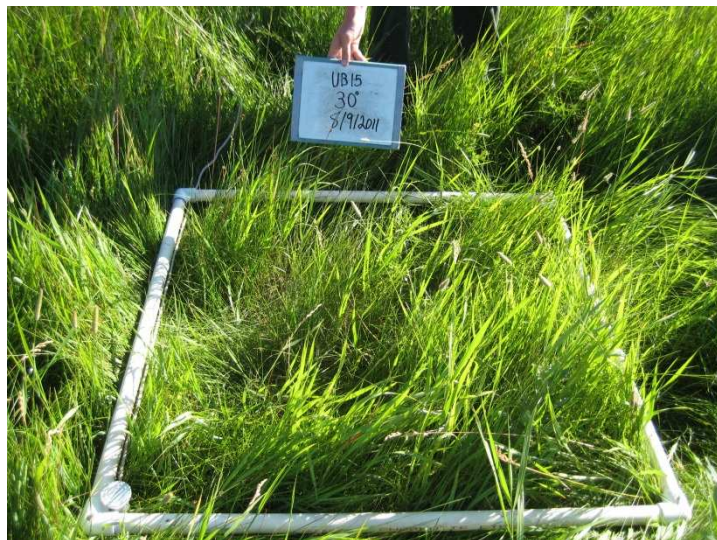


Figure 2. Photograph of a sampling unit (1m² quadrat in a circular nested plot).

Table 1. Information that should be shown in every photograph taken for a monitoring project, according to monitoring plot type with examples. CNP: circular nested plot.

Quadrat on CNP	Quadrat on a transect	Point or line intercept
-Plot name -Quadrat name -azimuth of spoke -Date	-Transect name -Quadrat name -quadrat number or location on transect -Date	-Transect name -Direction of photograph - e.g. 0 m → 30 m -Date
<div> UBM_15T 30 8/4/2014 </div>	<div> BL2_1 2.2 m 6/18/2014 </div>	<div> AVC_3 30 m → 0 m 7/22/2014 </div>

Several rules about the orientation of plot photographs have been established to maintain consistency across field seasons:

1. For quadrats, the marker (e.g. rebar stake, or distance on measuring tape) used to place the quadrat should be located in the lower left hand corner of the photograph. The dry-erase board displaying transect, plot, and date information should be located at the top of the quadrat, or in a place where the plot vegetation is not obscured (Figure 2).
2. For point or line intercept transects where quadrats are not used, a photograph should be taken along the transect from both ends of the transect. In other words, on a 30 m transect, a picture should be taken while standing at the 0 m end of the transect while facing the 30 m end, and again at the 30 m end while facing the 0 m end. The dry-erase board displaying transect, plot, and data information should be located at the top of the quadrat, or in a place where the plot vegetation is not obscured.

Since there will be a large number of pictures taken for most projects, it is important to save photographs using a consistent file-naming protocol to make relocating needed files possible (Section 6).

5.2 GPS Coordinates

Recording GPS coordinates for a monitoring plot will allow you to rapidly relocate plots in subsequent seasons. GPS coordinates are also useful for identifying plots to other crew members that will be applying treatments in the study site. GPS coordinates only need to be recorded once; usually when plots are first installed.

At RMNP, GPS coordinates are recorded using the **UTM Region 13** (Universal Transverse Mercator) system of coordinates using **NAD83 datum** (North American Datum 1983). Your GPS will automatically select UTM Region 13 based on your location, but always check these settings before marking GPS coordinates for a plot, or entering coordinates to relocate plots. Incorrectly recording or entering coordinates will make relocating plots based on GPS directions very difficult or impossible.

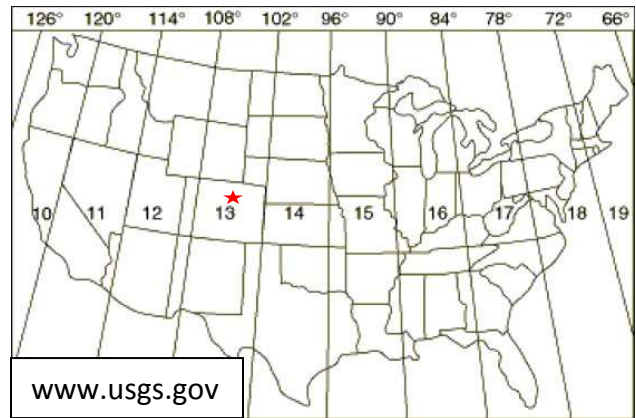


Figure 3. UTM projections on the United States. RMNP is located in Region 13.

For circular nested plots, GPS coordinates should be recorded at the central stake of the plot. For baseline or single line transects, GPS coordinates should be recorded at the starting point or 0 meter mark of the transect.

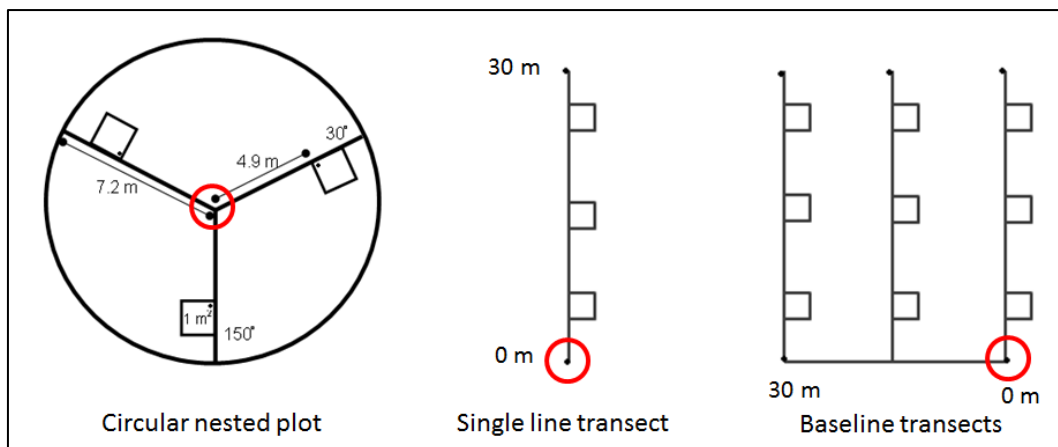


Figure 4. Recommended GPS coordinate locations for different plot types.

It is usually not necessary to collect more than one GPS location for each plot, since your plots will be set up systematically and you can use your knowledge of the plot layout to locate the remaining stakes. When baseline and single line transects are installed, the compass bearing of the direction of the transect should be recorded, making location of the other stake possible without GPS coordinates.

GPS coordinates should be recorded in the Easting (UTME) by Northing (UTMN) format as shown in Table 2.

Table 2. Example of GPS coordinates recorded in UTM format using NAD83 datum.

Study: South Lateral Moraine		Datum: NAD83
Plot name	UTME	UTMN
BSLME_001	0450465	4466552
BSLME_002	0450450	4466528
BSLME_003	0450419	4466533

There are other methods of recording location data and the theories behind the different coordinate systems can be complex. You will have extensive training on proper GPS usage during summer seasonal training before entering the field.

5.3 Plant Species Identification

One of the most common tasks you will encounter while in the field collecting data is identification of unknown plant species. Before your field season begins, you will be given training on identifying the most common species of plants in RMNP, but you will encounter plants that must be identified in the field or greenhouse. You should attempt to identify plants down to the genus and species level

You will be provided with several plant identification tools which you should have with you at all times when collecting vegetation data:

- Plant identification keys (Beidleman et al.'s *Plants of Rocky Mountain National Park*^[1], Weber's *Colorado Flora: Eastern Slope*^[16], Wingate's *Illustrated Keys to the Grasses of Colorado*^[17], and Shaw's *Grasses of Colorado*^[12] are highly recommended)
- Hand lens and dissecting kit including forceps and probe
- Specimen collection bags for difficult to identify species

It is a good idea to bring previous years' data sheets when heading out to a monitoring plot. Having this species information can provide clues as to what species you might expect to find in a plot, and sometimes help quickly identify a species in the field that might otherwise need to be examined under a microscope.

If you are having difficulty identifying a particular plant species, collect a sample in a sealable specimen bag. The bag should be marked with the plot and sampling unit the species is in and the date it was collected. It is often useful to note some of the site characteristics (e.g., slope or meadow, wet or dry soil, shaded or sunny, subcanopy) for help in keying later. Collect the entire individual plant including the root structure if possible, and it is good practice to collect two individuals if available. Also, collecting

individuals of the unknown plant from outside your sampling unit will avoid confounding your data by altering plant presence within the plot. Removing plants from your sites should be considered as a last resort only after several attempts at keying in the field are unsuccessful. Samples collected in the field can then be examined under a dissecting microscope and compared with herbarium specimens from the working herbarium located in the Resources Stewardship office.

In the event that a plant species cannot be identified in the field and there are only one or two individuals that can be found in the surrounding area, do not remove the unknown species. Instead, record the species as 'Unknown' and indicate its growth habit (e.g. Unknown forb 1). Take notes on the characteristics of the unknown plant and take a picture to include in the records for that transect. It is preferable to not destroy plants in the park whenever possible, especially when those plants are rare.

5.4 Vegetation Attribute Data Collection

The materials and methods for collecting vegetation attribute data vary depending on the particular attribute being measured. The vegetation attribute sections in this manual describe in detail the process of collecting and recording data for each attribute. See the Frequency, Density, and Cover sections for further instructions.

When using a digital data entry device such as a tablet computer to collect data, it is recommended that you still include paper data sheets with your materials to ensure efficient use of your time in the event of equipment failure.

6 Data Management

Saving project files using consistent naming and organization conventions ensures files can be easily located and identified in future monitoring seasons.

6.1 Naming Data Files

File names should be concise and descriptive and named in a way that they can be identified if separated from their original file folder. File name length restrictions vary with software and operating systems, so file names should be restricted to fewer than 30 characters to ensure compatibility with some older programs with limited file name sizes.

File names should contain these basic components:

- **The date the file was created** in a YYYYMMDD (year, month, day) format. Files with the YYYYMMDD date format will automatically sort chronologically within a file folder, making it easier to locate the most recent version of a file if multiple versions exist.
- **A descriptive and unique identifier** that includes the project and transect/plot name and file type (e.g. raw data, statistics output, figures, tables). In most cases, data files for a monitoring project should contain the data for all plots or transects for that data collection year. Having descriptive and unique file names allows for the identification of files that have been separated from their original file location.

Other general rules to ensure ease of location and file name compatibility across platforms include:

- Use identifiable abbreviations in files names to limit characters used.
- Use underscores instead of spaces (e.g. ProjA_RawData).
- Do not use special characters (e.g. “ , : ? ! @ /) which can be reserved for special functions in some programs.
- Alternate upper- and lower-case characters to make reading file names easier (e.g. RawData instead of rawdata).

For example, consider a revegetation project in Upper Beaver Meadows with 12 treatment and 12 control plots. This is the first revegetation project that has been monitored on this site, and the file that is being created contains the raw treatment and control plot data. A file name such as *Upper beaver meadows treatment data 2012* is a poor file name. A file name such as *20120812_UBR1_AllPlot_RawData* contains much more information, including when the data were entered, the specific project these data are from, what plots these data are from, and what form the data are in (Figure 1).

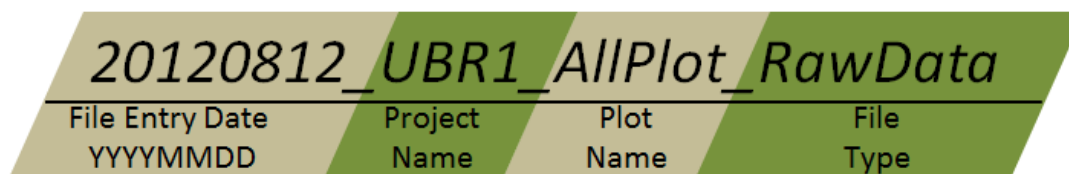


Figure 5. Components and format of a good file name

The descriptive identifier portion of file names will be different for each project, but it is very important to consistently name files across monitoring seasons *within* a project. This will avoid confusion when compiling project data to compare across multiple monitoring seasons.

Files containing plot and project photographs should follow the same file naming convention described for data files. Because there can be hundreds of photographs taken in a single monitoring season, renaming the files in bulk using Windows Explorer is recommended. This is accomplished by highlighting all of the file names in the folder containing photograph files for that project in that year and renaming the first file with a descriptive file name as described above. Windows will then rename all of the files with that name and append a sequential number after each file name (Figure 2).

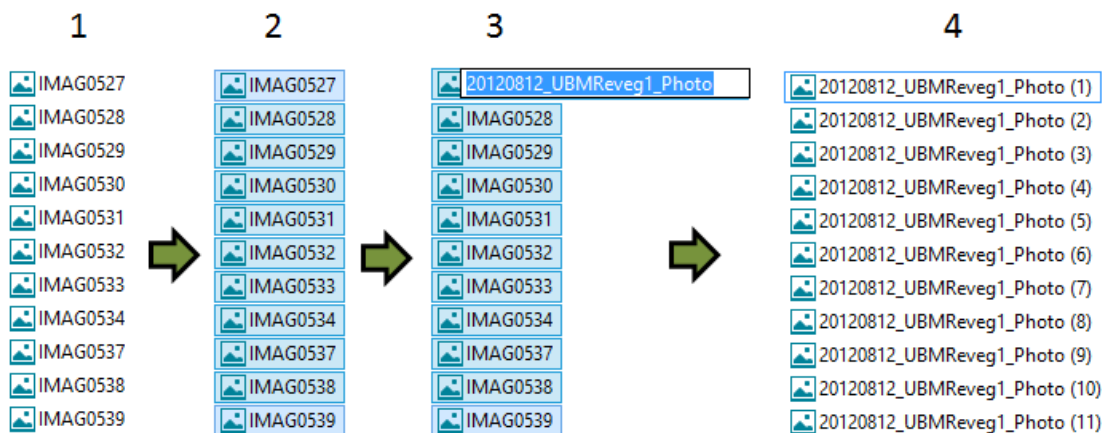


Figure 6. Changing photograph file names in bulk. (1) Windows automatically names uploaded image files. Copy and paste image files to the appropriate file location. (2) Highlight all file names by clicking the first file, holding the Shift key, and clicking the last file. (3) Right-click the first file, click Rename File, and rename the file. (4) Pressing enter will rename all the highlighted files with the same name with sequential numbers appended.

Renaming files in this way saves time while still making files identifiable to the project and data collection year. Pictures should include a dry-erase board with the date and plot details in the frame, and particular plot photographs can be located by browsing the image previews.

6.2 File Folder Organization

Consistent file folder organization across projects and seasons makes navigating the many files that accumulate over monitoring seasons less confusing. Having file folders organized in a hierarchical fashion makes saving and finding specific files simpler for large, multi-year monitoring projects.

Files are organized in the Vegetation Monitoring folder on RMNP's shared computer drive in a way that moves from a broad to specific context. The Vegetation Monitoring folder location can be found by following this file pathway:

O: drive>resmgmt>Resources operations>Vegetation monitoring

The general organization of Vegetation Monitoring file folders on the RMNP shared drive are shown below.

1 General file folder organization

- Vegetation monitoring
 - Project files→see 2
 - Completed Projects
 - Current Projects
 - Pictures
 - Other files
 - Vegetation monitoring database
 - Yearly Monitoring Reports
 - Year

2 Project file folder organization

- Project files
 - Completed Projects
 - Follows current project structure
 - Current Projects
 - Timelines and Schedules
 - Year
 - Project A
 - Year
 - Data
 - Analyses
 - Figures and Tables
 - Yearly Project Report
 - Literature

Other file folders can be added as needed, and should be placed logically within the file folder hierarchy described here.

7 Reporting Results

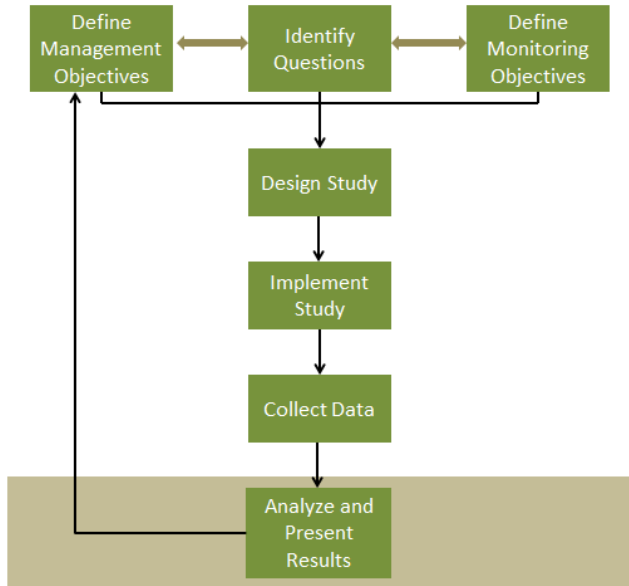
Describing the results of a monitoring project in a clear and informative manner ensures that your findings can be used by crew leads and supervisors to inform current and future management decisions.

7.1 Data Analysis

This section will describe analytical methods that are used in RMNP to interpret the most common types of data sets resulting from monitoring projects in the park.

Summarizing Data

The first step in the data analysis process is to summarize the raw data that you have collected. Summarized data are used for statistical analysis of the data as well as the creation of figures and tables to illustrate your results. All of these calculations can be easily performed in an Excel spreadsheet. Also, MS Access database combined with Excel functions can be used to quickly organize and group data by functional groups prior to calculating summary data. See Appendix 3 for more details.



To calculate the **sample mean**, or average, for a variable, divide the sum of all the values by the number of values added (Figure 1). Mean values are calculated for individual monitoring plots and again across all plots of a treatment type. For example, consider a monitoring project with three plots treated with herbicide and three untreated control plots. Mean cover values are calculated for the target plant in each of the three treatment plots (Figure 2). The mean of these three plot values gives the mean target plant cover in treatment plots. This process is repeated for control plots, and the mean value of the target plant in treatment and control plots can be compared.

$$\bar{x} = \frac{\sum x_i}{n}$$

Figure 1. Sample mean formula. \bar{x} : sample mean, $\sum x_i$: sum of individual values, n : number of individual values (sample size).

The variables you will most commonly be summarizing will be one of the three main vegetation attributes described in Section 5. Also, you will be analyzing these vegetation attribute data by individual species, by functional groups, or a combination of both, depending on your project objectives (Section 2). Each vegetation attribute section describes in detail how to calculate the sample mean for that attribute under the *Data Summary* subheading.

In Excel, the [=average(range)] command will calculate the mean of a range of values.

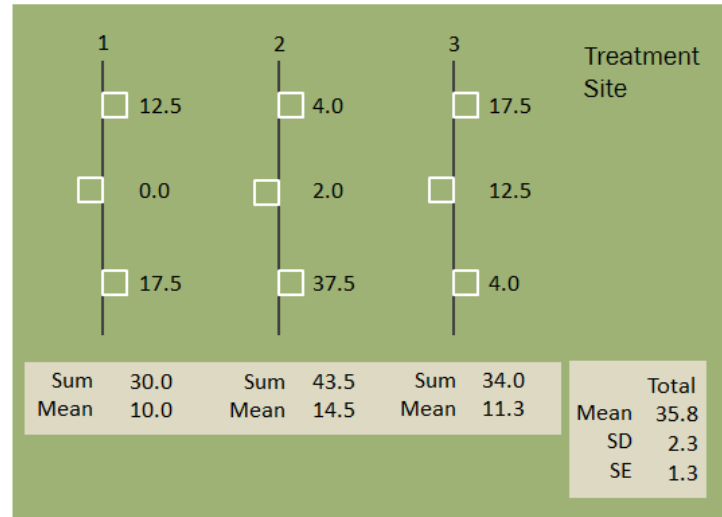


Figure 2. Example of mean, standard deviation, and standard error of the mean calculations for three monitoring transects in a treatment site. The same process is applied to circular nested plots.

The plot mean values are used to calculate the **standard deviation** and the **standard error of the mean**, which are estimates of the amount of variation that exist in a data set and are used for data analysis and figure and table generation.

Standard deviation is calculated using the formula shown in Figure 3.

$$s_x = \sqrt{\frac{\sum (x_i - \bar{x})^2}{n - 1}}$$

Figure 3. Standard deviation formula. s_x : standard deviation; x_i : individual plot values; \bar{x} : sample mean; n : sample size.

In Excel, the [=stdev(range)] command will calculate the standard deviation of a range of values. The range of values should be the same as that used to calculate the mean.

The standard error of the mean is calculated by dividing the standard deviation by the square root of the sample size (Figure 4).

$$SE_x = \frac{s_x}{\sqrt{n}}$$

Figure 4. Standard deviation formula. SE_x : standard error of the mean; s_x : standard deviation; \sqrt{n} : square root of the sample size.

There is no command in Excel for calculating the standard error of the mean but it can be calculated manually by entering the formula [= (standard deviation value)/(sqrt(sample size))].

Statistical Analyses

This section will briefly describe some commonly used approaches to data analysis in the vegetation monitoring program.

Testing the Difference Between Two Sample Means

The ***t-test*** is used to compare the means of two samples. If comparing mean vegetation cover in treatment and control plots for a single data collection year, a *t-test* can be used to determine if the values are different. The null hypothesis of a *t-test* is that the means of the two samples are equal. A *t-test* resulting in a statistically significant *p-value* ($p < 0.05$ for $\alpha = 0.05$) indicates that the means being compared are not equal. This type of test is normally only used in the first year of data collection when only two means are being compared. Other methods are used to analyze data from the same plots measured multiple times, which are not independent and must be analyzed as repeated measures.

A *t-test* can be performed in Excel using the function [=TTEST(array1,array2,tails,type)]. *Array 1* and *2* are the columns of data from your two samples. *Tails* indicates whether you want to perform a one (enter a 1) or two (enter a 2) tailed *t-test*. A one-tailed test is used when a directional change (*either* an increase or decrease) is expected in mean values, and a two-tailed test is used to detect both increases and decreases in mean values. We are usually concerned with any vegetation changes that are happening in a study site, the two-tailed test should be used. The *type* of test most commonly used is a type two (enter a 2) test for two samples with equal variance, since the *t-test* is not very sensitive to unequal variances (Sokal and Rolf 1995). A type one test could be used if comparing the sample mean of plots before and after a treatment or manipulation (Table 1).

Table 1. Types of *t-tests* in Excel

Type	Test Performed
1	Paired (same plots before and after)
2	Two-sample equal variance (independent plots)
3	Two-sample unequal variance (independent plots)

Testing Differences Between More Than Two Sample Means

One-way or single-factor **Analysis of Variance (ANOVA)** is used to compare means of more than two independent subjects. For example, if a study is comparing the effectiveness of three different exotic species control methods, the mean vegetation values of for the three plot types could be simultaneously compared using ANOVA.

The null hypothesis of an ANOVA is that the means of all the groups are equal. An ANOVA resulting in a statistically significant *p-value* ($p < 0.05$ for $\alpha = 0.05$) indicates that at least one of the means being compared is different.

In Excel, a single-factor ANOVA can be performed by using the *ANOVA:single factor* function in the data analysis tab. See Table 2 for detailed instructions.

Table 2. Step-by-step instructions for performing single factor ANOVA in Excel

Enter data in columnar format

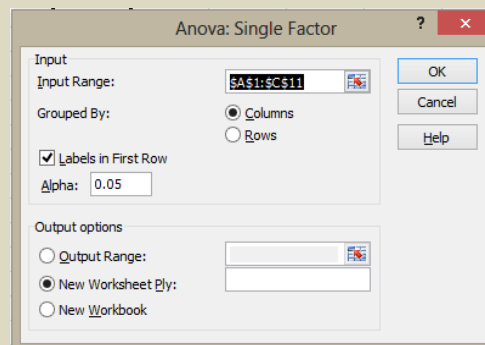
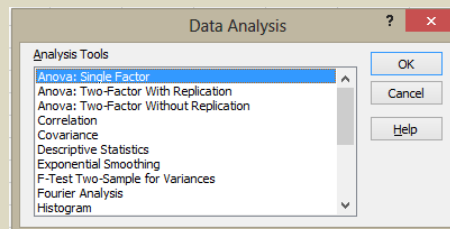
Select Data Analysis in the Data tab in Excel

Select ANOVA: Single Factor from Analysis Tools

Select the columns of data for the Input Range. Indicate whether you included data labels in the first row. Press OK.

ANOVA output. A *p-value* of 0.02, less than 0.05, indicates that at least one of the group means is different than the others.

	A	B	C
1	Herbicide	Manual	No treatment
2	6	6	16
3	3	8	15
4	9	11	12
5	8	5	5
6	1	3	14
7	9	8	13
8	0	6	2
9	5	0	16
10	4	8	19
11	6	3	10



	A	B	C	D	E	F	G
1	Anova: Single Factor						
2							
3	SUMMARY						
4	Groups	Count	Sum	Average	Variance		
5	Herbicide	10	38	3.8	6.4		
6	Manual	10	44	4.4	6.711111		
7	No treatment	10	121	12.1	24.76667		
8							
9							
10	ANOVA						
11	Source of Variation	SS	df	MS	F	P-value	F crit
12	Between Groups	118.0667	2	59.03333	4.675565	0.018048	3.354131
13	Within Groups	340.9	27	12.62593			
14							
15	Total	458.9667	29				

Excel's ANOVA output does not indicate which group means are different from each other, so *t-tests* comparing the pairs of means (e.g. Herbicide vs Manual, Herbicide vs No Treatment, etc.) can be used to interpret your results in more detail. This is a rudimentary method of means separation, but should be sufficient for detailing observed differences.

Complex data sets

Single-factor or one-way ANOVA is only appropriate to use for data collected in the same year. If data have been collected in the same plots for successive years, **repeated measures ANOVA** must be used. More complex ANOVA techniques like **multivariate analysis of variance (MANOVA)** are used when multiple factors are included in your study design. MANOVA can also accommodate repeated measures analysis, making it a versatile tool for analyzing complex data sets. For example, if the goal of a study is to determine how herbicide application affects vegetation cover in different vegetation communities over time, repeated measures ANOVA or MANOVA techniques will be required. As discussed in the study design section, adding multiple factors to a study increases the complexity a study's design as well as the analysis and interpretation of the resulting data.

Repeated measures ANOVA, MANOVA and other sophisticated types of data analyses cannot be performed in Excel, but can be performed in dedicated statistics packages like SAS, R, SPSS and JMP. Discuss your data analysis needs with your supervisor to coordinate a plan to obtain statistics assistance for complex data sets. Consult the texts cited in the Further Reading section below or any statistics textbook for detailed descriptions of the methods discussed here.

Notes on Statistical significance

Statistical significance is the probability that an observation or effect is not due to random chance alone (Gotelli and Ellison 2012; Ott and Longnecker 2004). The most commonly used level of significance, called alpha (α), is 0.05 or 5%. A statistical test that produces a *p-value* less than 0.05 is considered a statistically significant result. In other words, a *p-value* of less than 5% indicates that there is a greater than 95% chance that the observation or effect you tested was not due to random chance.

Sometimes you may obtain *p-values* that are a several percentage points above or below the accepted α -level. In a report, these results should be reported as either significant or non-significant based on your α , but make note of their marginally significant nature to avoid overstating their significance.

In other cases you may encounter statistically significant results due to minute changes in an attribute. Statistical significance does not necessarily mean a result is biologically significant. For example, an uncommon species or functional group may have a very small mean density in a site. Any moderate change in density will likely return a

significant result. If the change over a single year is only several plants per square meter this may be due to normal annual variation. The value in having monitoring projects that span five to ten years is that it allows us to track both rapid and gradual changes on monitoring sites. Remember to use your biological intuition when interpreting these results, and make note of whether these changes appear to be biologically significant. The exception to this recommendation is when any invasive species is observed in plots previously unoccupied by that species. This is a situation where early intervention can be most successful in preventing the spread of invasive species into uninvaded sites.

Further Reading:

-*An Introduction to Statistical Methods and Data Analysis*, R. Lyman Ott and Michael Longnecker, 2008 ^[10]

-*Biometry*, Robert Sokal and F. James Rohlf, 1995 ^[13]

-*A Primer of Ecological Statistics*, Nicholas Gotelli and Aaron Ellison, 2004 ^[7]

7.2 Report Writing

Results of monitoring projects are reported in a yearly vegetation monitoring report. This report is written in the style of a scientific journal article with an abstract, introduction, materials and methods, results, and discussion sections. The report for each monitoring project is written as a stand-alone paper and the individual project reports are compiled into one yearly report. This section briefly describes the recommended content and organization of a yearly monitoring report. A style guideline outlining recommended formatting details for the yearly monitoring report can be found in the Appendix.

Sections described in green boxes are written once for the entire yearly report.

Sections described in gray boxes are written separately for each individual monitoring project.

Title Page

In addition to the title of the yearly report, the title page indicates the authors of the paper and has a simple table of contents.

Introduction

This section introduces the reader to the basic study topic and describes the purpose of the project. This section should begin with a broad outline of the background of the components of the study and proceed to describing the questions and hypotheses of the study. Referencing peer-reviewed studies and relevant agency documents to support your introduction is recommended.

Materials and Methods

The details of how a study was conducted in the field and how the data were analyzed are presented in a narrative description in this section. The reader should be able to repeat each study based on the descriptions given.

Results

The results of data summary and analysis are described in text and illustrated in either figure or table format in this section. When presenting ANOVA results, be sure to include an ANOVA table. Any figures and tables should be directly referred to in the text of the Results section. Avoid interpreting the results in this section; simply describe trends and the outcomes of data analysis.

Discussion

In this section, the results are interpreted in the context of the original questions and hypotheses. The specific conclusions of the study should be described (were the study questions answered?, etc.) before moving on to making conclusions about the broad management implications of the study and recommendations for future management.

actions and monitoring techniques. This section should cite peer-reviewed studies and relevant agency reports to support the conclusions made.

Alternative option: Results/Discussion

If a monitoring study is very simple and separate Results and Discussion sections are not needed for clear organization and description of study results, a combined Results/Discussion section is recommended.

Literature Cited

List the resources cited throughout the report using the Council of Science Editors (CSE) Name-Year citation format, shown below. See Appendix for in-text citation format.

Baker, W.L., J. Garner and P. Lyon. 2009. Effects of imazapic on cheatgrass and native plants in Wyoming big sagebrush restoration for Gunnison sage-grouse. *Natural Areas Journal*. **29**:204-209.

Appendices

The Appendix includes any material that is not a component of a scientific report, any results and figures that are interesting but not necessary for a report, or any highly detailed methods descriptions that would be inappropriate for a report but are helpful for subsequent monitoring season activities. Additionally, this section should include tables with the GPS coordinates of all monitoring plots that are still active.

It is useful to review previous seasons' reports to understand expected standards for seasonal reports.

7.3 Figures and Tables

This section briefly describes some recommended approaches to presenting data in graph and table formats. Typically, graphs are more effective at displaying data in a way that is easily and quickly understood compared to tables of summarized data. A good figure should still make sense if it were to be viewed separately from the rest of the report.

Bar graphs are useful when comparing data collected in a single monitoring season (Figure 1). Standard error of the mean (SEM) bars show the variability observed in the data sets.

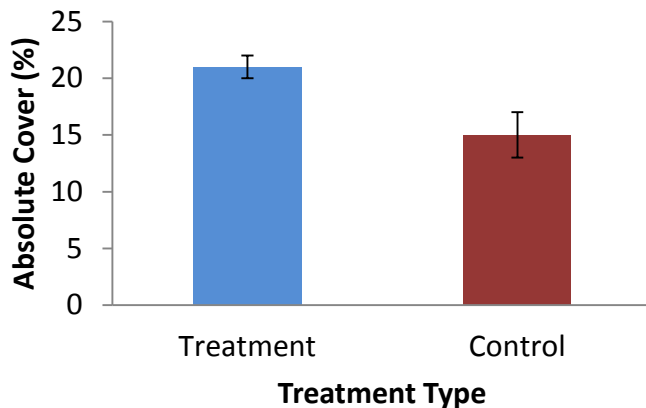


Figure 1. Example of a simple bar graph. Error bars show \pm SEM (standard error of the mean).

Line graphs are useful for comparing data that were collected repeatedly over time (Figure 2). This is the type of figure you will most commonly be generating for the yearly monitoring reports.

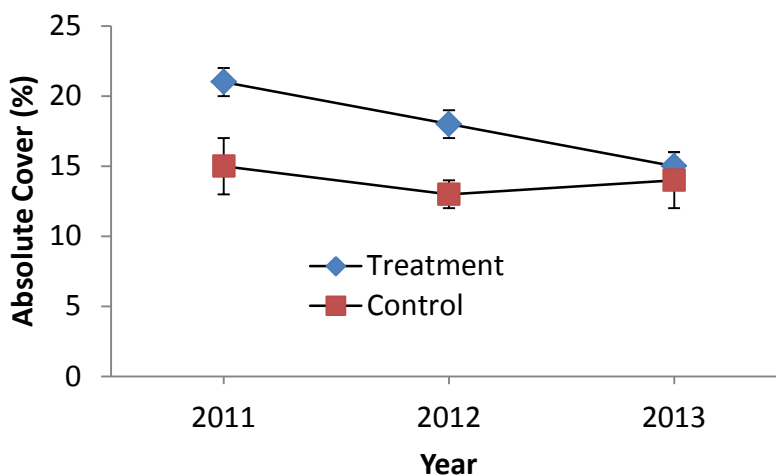


Figure 2. Example of a line graph with markers. Error bars show \pm SEM (standard error of the mean).

Tables are used to present information that can't be displayed in a graph and would be difficult to understand if written in the body of the report. One common use of tables in monitoring reports is to display the results of an ANOVA (Table 1). The format and organization of a table depends largely on the content of the table, so there are few guidelines to follow. Maintain consistency in table formatting throughout the entire report to maintain a professional look. Keeping tables as simple and streamlined as possible will ensure their readability and usefulness to the reader.

Table 1. Example of a table used in vegetation monitoring reports. Statistically significant *p-values* are shown in bold.

Source of Variation	df	F-statistic	P-value
Between Subjects			
Veg Type	2	4.10	0.08
Treatment	1	51.82	>0.01
Veg Type x Treatment	2	4.62	0.06
Within Subjects			
Time	3	9.32	0.03
Time x Treatment	3	8.82	0.04
Time x Veg Type	6	3.46	0.05
Time x Veg Type x Treatment	6	2.97	0.08

8 References and Further Reading

- 1** Beidleman, L.H., R.G. Beidleman and B.E. Willard. 2000. *Plants of Rocky Mountain National Park*. 1st ed. revised. Helena, MT: Falcon Publishing.
- 2** CDA [Colorado Department of Agriculture]. Noxious Weed Species [Internet]. Accessed 3/2014. Available from: http://www.colorado.gov/cs/Satellite/ag_Conservation/CBON/1251618874438.
- 3** Cohen, J. 1988. *Statistical Power Analysis for the Behavioral Sciences*. 2nd ed. New York, NY: Lawrence Erlbaum Associates.
- 4** Coulloudon, B., K. Eshelman, J. Gianola, N. Habich, L. Hughes, C. Johnson, M. Pellant, P. Podborny, A. Rasmussen, B. Robles, P. Shaver, J. Spehar, J. Willoughby. 1996. *Sampling Vegetation Attributes*. Denver (CO). Available from: Bureau of Land Management National Business Center. BLM technical reference 1734-4.
- 5** Elzinga, C.L., D.W. Salzer, and J.W. Willoughby. 1998. *Measuring and Monitoring Plant Populations*. Denver (CO). Available from: Bureau of Land Management National Business Center. BLM Technical Reference 1730-1.
- 6** FHA [Federal Highways Administration]. 2007. *Roadside Revegetation: An Integrated Approach to Establishing Native Plants*. Vancouver (WA), Available from: Technology deployment program, Western federal lands highway division, Federal highway administration. Report number: FHWA-WFL/TD-07-005.
- 7** Gotelli, N. J. and A.M. Ellison. 2004. *A Primer of Ecological Statistics*. 1st ed. Sunderland, MA: Sinauer Associates Inc.
- 8** Greig-Smith, P. *Quantitative Plant Ecology: Studies in Ecology Volume 9*. 3rd ed. Berkeley, CA: University of California Press.
- 9** Hurlbert, S.H. *Pseudoreplication and the Design of Field Experiments*. Ecological Monographs. **52**:187-211.
- 10** Ott, R.L. and M. Longnecker. 2008. *An Introduction to Statistical Methods and Data Analysis*. 6th ed. Belmont, CA: Brooks/Cole.
- 11** Ruyle, G.B. *Some Methods for Monitoring Rangelands and other Natural Area Vegetation*. Tuscon (AZ). Available from: Division of range management, The University of Arizona, College of Agriculture. Extension Report 9043.
- 12** Shaw, R.B. 2008. *Grasses of Colorado*. Boulder, CO: University Press of Colorado.

- 13** Sokal, R.R. and F.J. Rohlf. 1995. *Biometry*. 3rd ed. New York, NY: W.H. Freeman and Company.
- 14** Stohlgren, T.J. 2007. *Measuring Plant Diversity: Lessons from the Field*. 1st ed. New York, NY: Oxford press.
- 15** USDA [United States Department of Agriculture]. PLANTS Database [Internet]. Accessed 8/2013. Available from: <http://plants.usda.gov/>.
- 16** Weber, W.A. and C.W. Wittman. 2002. *Colorado Flora: Eastern Slope*. 3rd ed. Boulder, CO: University Press of Colorado.
- 17** Wingate, J.L. 1994. *Illustrated Keys to the Grasses of Colorado*. Denver, CO: Wingate Consulting.

Appendices

A1 Sampling Design Decision Tables

Summary of Vegetation Attributes and Their Applications

Attribute	Monitoring Objective	Vegetation Characteristics	Plot types
Frequency (page 17)	Monitor general changes in vegetation. Can be completed rapidly. Especially useful for monitoring movement of annual species and plant invasions.	Good for use with most growth-forms; especially when monitoring rhizomatous or mat-forming species that are difficult to identify as individuals.	Single-line or Baseline transects with quadrats
Density (page 20)	Monitor recruitment or mortality of individuals with less focus on detecting short-term changes in vigor or size.	Good for use with vegetation communities or target species that are easily identified as individual plants. Not efficient for monitoring very dense or rhizomatous species.	Single-line or Baseline transects with quadrats
Cover (page 25)	Monitor changes in plant vigor and size with ability to detect sub-lethal changes in vegetation with less focus on tracking recruitment or death of individual plants.	Good for use with most growth-forms. Useful for communities where species may be difficult to identify as individuals or when a community is composed of both large, less common species and small, abundant species.	Single-line or Baseline transects with quadrats
			Circular Nested Plots
			Single-line or Baseline transects using point-intercept
			Single-line or Baseline transects using line-intercept

Summary of Monitoring Plot Types and Their Applications

Plot Type	Site and Vegetation Characteristics	Attributes	Measurement Unit
Single Line Transect	Long and narrow sites, or when monitoring a target species or group with a clumped distribution. Single line transects can be strategically located to ensure clumped target species data are being collected efficiently.	Frequency	Quadrat
		Density	Quadrat
		Cover	Quadrat
			Point-intercept
			Line-intercept
Baseline Transect	Larger sites with moderately well distributed target species.	Frequency	Quadrat
		Density	Quadrat
		Cover	Quadrat
			Point-intercept
			Line-intercept
Circular Nested Plot	Larger sites with moderately well distributed target species. Also useful for collecting species richness data.	Cover	Quadrat and frequency count

A2 Report Formatting

General Report Formatting

Individual project reports are each written as a standalone report in the style of a scientific journal article (page 53). The following formatting recommendations are intended to maintain consistency of the presentation of the reports across monitoring seasons.

Text

- Body of text single-spaced
- Sentences separated by a single space
- Paragraphs not indented
- Paragraphs separated by a single 12 pt line
- Single or double space before and after tables and figures as needed for clarity
- See following table for contextual text formatting guidelines

Text Type	Font	Size	Format	Example
Major headings (Paper and project titles)	Franklin Gothic Book	26	Normal	Title
Section Headings	Calibri	16	Bold	Results
Subheading	Calibri	14	Italic	<i>Native Shrubs</i>
Body	Calibri	12	Normal	The data show...
Latin species names	Calibri	12	Italic, Genus capitalized	<i>Viola selkirkii</i>
Figure and Table Captions	Calibri	10	Normal	This figure shows changes in...
Literature Cited	Calibri	10	Normal	Restoration techniques...

- Literature cited follows the CSE name-year citation format. Ex:
Baker, W.L., J. Garner and P. Lyon. 2009. Effects of imazapic on cheatgrass and native plants in Wyoming big sagebrush restoration for Gunnison sage-grouse. *Natural Areas Journal*. **29**:204-209.
- In text citations formatted as:
 - Single author (Darwin 1859)
 - Two authors (Watson and Crick 1953)
 - More than two authors (Gleason et al. 1926)
 - Citing multiple sources at once (Clements 1938; Carson 1940)

Figure Formatting

The following guidelines are for figures and tables created in Excel and are intended to improve their readability and professional appearance.

Once you have formatted a figure, you can save that figure format as a design template by clicking the Design tab in Chart Tools, and selecting Save as Template. That template can be selected from the Insert tab under Other Charts and All Chart Types. This reduces the amount of time spent reformatting figures when dozens of the same type are being generated.

Graphs

General

- Omit (right-click and delete) horizontal gridlines
- Omit outer figure border (shape outline)
- Add x- and y-axis titles in bold
- Axis titles and dependent/independent axis values in 12 pt font
- Figure number and descriptive caption *below* every figure in 10 pt font
- Figures numbered consecutively and independently from tables (i.e. Figure 1, Table 1, Figure 2; *not* Figure 1, Table 2, Figure 3)
- Add error bars showing the standard error of the mean for each individual mean value (see below for instructions on adding unique error bars in Excel)
- Avoid using 3-dimensional figures unless it aids in the interpretation of the figure

Bar graphs

- Delete redundant data legend, columns should be labeled on x-axis
- Choose different column colors for clarity or patterns if presenting in black and white
- Right click the x-axis and open the format axis menu to change default x-axis tick marks from Major tickmarks:outside to Major tickmarks:none

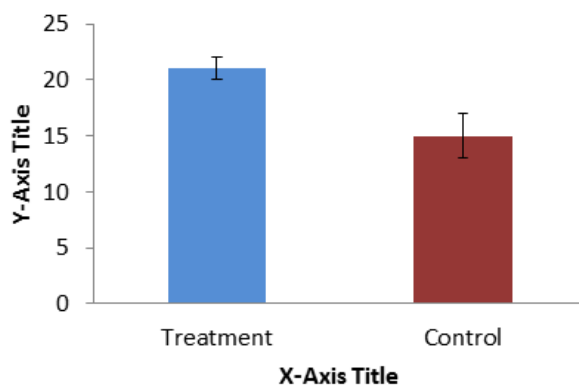


Figure 1. Example of a simple bar graph. Error bars show \pm SEM (standard error of the mean).

Line Graphs

- Include markers at mean values along the line
- Markers should be different shapes for each variable
- Right click the x-axis and open the format axis menu to change default x-axis tick marks from Major tickmarks:outside to Major tickmarks:none, and Minor tickmarks:none to Minor tickmarks:outside
- Reformat lines: Black color, 1 pt width
- Reformat markers: Marker size=8
- The default format for Excel line graphs is to have the legend separated to the right of the graph, creating a large empty white area in the figure. Change this by left-clicking the plot area to highlight it and dragging right side of the highlighted plot to the right, filling in the empty space. Reorient the data legend within the plot area.

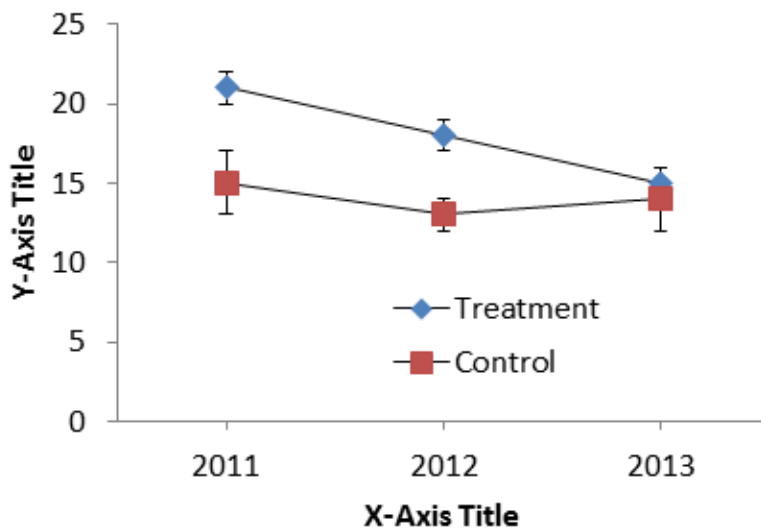


Figure 2. Example of a line graph with markers. Error bars show \pm SEM (standard error of the mean).

Tables

Since the format of a table depends largely on its contents there are fewer clearly defined rules to adhere to when formatting. The best approach is to maintain consistent formatting that is simple and easy to understand to make the table readable and useful to readers.

- Use a 10 pt font to keep tables from becoming crowded and difficult to read
- Tables numbered consecutively and independently from figures (i.e. Table 1, Figure 1, Table 2; *not* Table 1, Figure 2, Table 3)
- Table number and descriptive caption *above* every table in 10 pt font

Adding Unique Error Bars in Excel

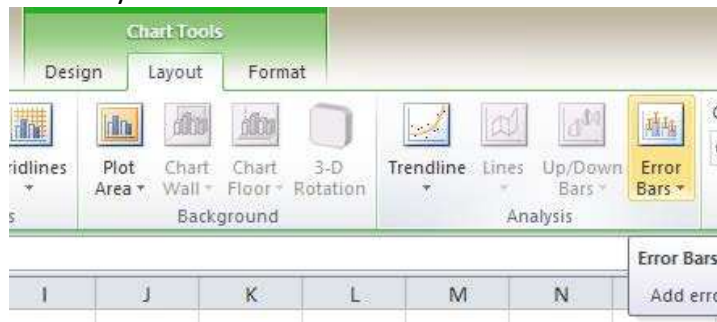
Excel can add generic error bars across all variables, but there is not a simple function for adding unique standard error bars to excel figures. The following steps need to be followed to add unique error bars to figures.

Bar Graphs

After the bar graph is generated:

-Click anywhere in the graph area

-Click Layout tab in Chart Tabs



-Click Error Bars

-Select More Error Bar Options

-Click Custom and Specify Value

Error Amount

☐ Fixed value: 0.1

☐ Percentage: 5.0 %

☐ Standard deviation(s): 1.0

☐ Standard error

☒ Custom: Specify Value

-Click red arrow under Positive Error Value, highlight cells containing SEM; repeat for Negative Error Value, Click OK

	A	B	C	D
1		Treatment	Control	
2	mean	21	15	
3	sem	1	2	
4				
5				
6				
7				
8				
9				
10				
11				
12				

Custom Error Bars ? X

Positive Error Value

Negative Error Value

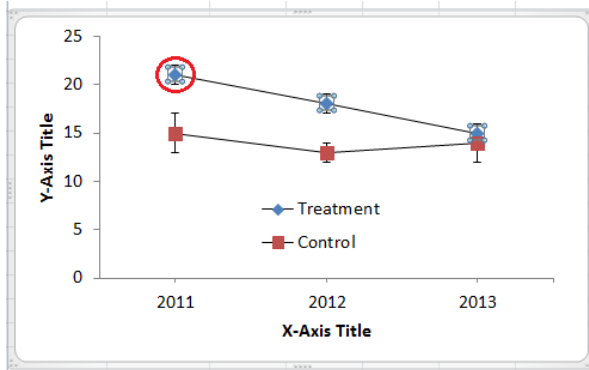
OK Cancel

Line Graphs

The process for adding error bars to line graphs is more complex.

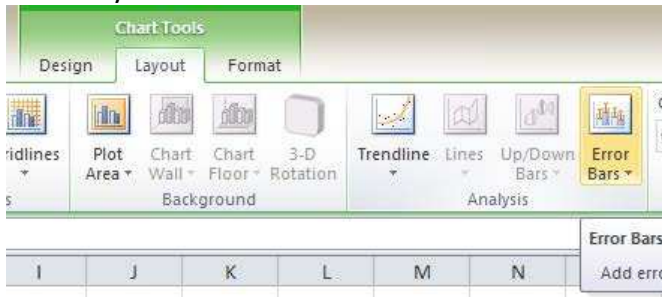
After the line graph is generated:

-Click in the graph area on the line with the markers you want to add error bars to. This will highlight the entire data series for that variable.



	A	B	C	D
1		2011	2012	2013
2	Treatment	21	18	15
3	sem	2	1	2
4	Control	15	13	14
5	sem	1	2	1

-Click Layout tab in Chart Tools



-Click Error Bars

-Select More Error Bar Options

-Click Custom and Specify Value

Error Amount

☐ Fixed value: 0.1
☐ Percentage: 5.0 %
☐ Standard deviation(s): 1.0
☐ Standard error
☒ Custom: Specify Value

-Click red arrow under Positive Error Value, highlight cells containing SEM; repeat for Negative Error Value, Click OK. In this example error bars are being added to the treatment markers first, so those values are highlighted.

	A	B	C	D	E	F	G	H
1		2011	2012	2013				
2	Treatment	21	18	15				
3	sem	2	1	2				
4	Control	15	13	14				
5	sem	1	2	1				

Custom Error Bars ? x

Positive Error Value
=Sheet2!\$E\$2

Negative Error Value
={1}

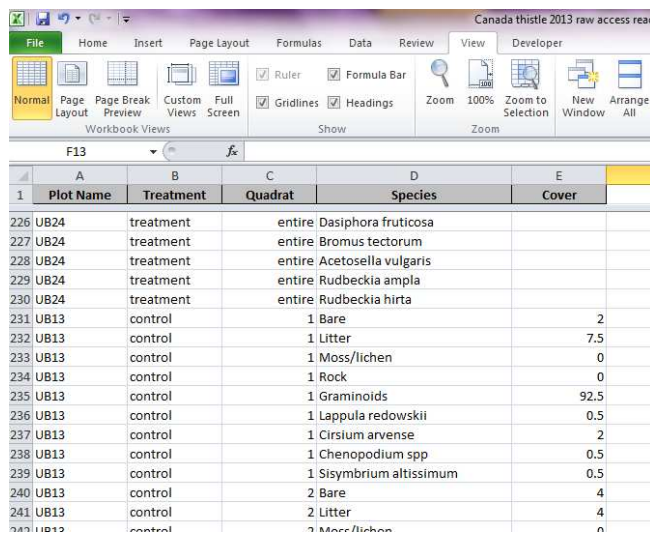
OK Cancel

-Repeat these steps for each variable with a line on the graph.

A3 Data management in Access

Using the Access database to sort and summarize data can save time and prevent data entry errors before data analysis. The following example uses the 'Canada thistle/Milestone herbicide' project data as an example, and the resulting data sets and outputs for different projects will vary. The process described will take your data from a completely raw state to a format where data are grouped by Plot, Species, Growth Habit, and Native Status. Species data will also be summed within each plot. Using this approach can save many hours of manual data manipulation and organization.

1. Enter data from paper field sheets into an Excel sheet in columnar format (Step 1 may be skipped due to tablet data entry and automated data sheet population from data entry software).



	A	B	C	D	E
	Plot Name	Treatment	Quadrat	Species	Cover
226	UB24	treatment	entire	Dasiphora fruticosa	
227	UB24	treatment	entire	Bromus tectorum	
228	UB24	treatment	entire	Acetosella vulgaris	
229	UB24	treatment	entire	Rudbeckia ampla	
230	UB24	treatment	entire	Rudbeckia hirta	
231	UB13	control	1	Bare	2
232	UB13	control	1	Litter	7.5
233	UB13	control	1	Moss/lichen	0
234	UB13	control	1	Rock	0
235	UB13	control	1	Graminoids	92.5
236	UB13	control	1	Lappula redowskii	0.5
237	UB13	control	1	Cirsium arvense	2
238	UB13	control	1	Chenopodium spp	0.5
239	UB13	control	1	Sisymbrium altissimum	0.5
240	UB13	control	2	Bare	4
241	UB13	control	2	Litter	4
242	UB13	control	2	Moss/lichen	0

2. In Access, import Excel data into Access database by clicking Excel in the External Data tab.



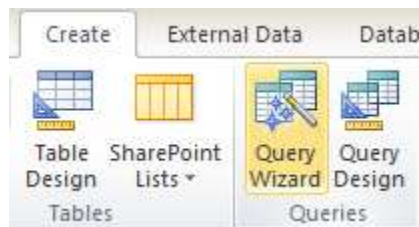
The resulting data table will look similar to this:

Canada thistle 2013 with richness						
ID	Plot Name	Treatment	Quadrat	Species	Cover	
1	UB12	treatment	1	Bare	7.5	
2	UB12	treatment	1	Litter	7.5	
3	UB12	treatment	1	Moss/lichen	0	
4	UB12	treatment	1	Rock	0	
5	UB12	treatment	1	Taraxacum officinale	2	
6	UB12	treatment	1	Graminoids	82.5	
7	UB12	treatment	1	Solidago canadensis	2	
8	UB12	treatment	1	Phleum pratense	7.5	
9	UB12	treatment	1	Lepidium densiflorum	2	

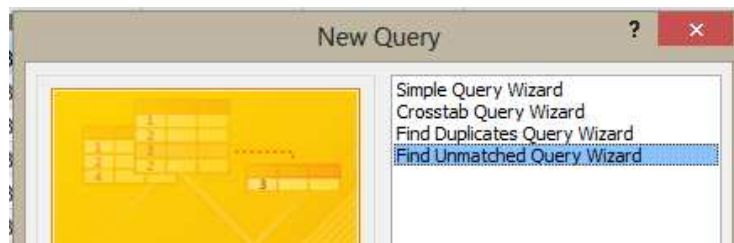
3. The Vegetation Monitoring Access database has a Plant Associations table that lists plant species, their growth habit, their native status, their family classification, and any other attribute (e.g. seeded on Project X) that can be used to organize and group species through Access queries.

Plant Species Associations						
Species	Growth Habit	Native Status	Spp Numb	Seeded on BL	Family	
Acer glabrum	tree	native	1	N	Aceraceae	
Acetosella vulgaris	forb	non native	2	N	Polygonaceae	
Achillea lanulosa	forb	native	3	N	Asteraceae	
Achillea millefolium	forb	native	4	N	Asteraceae	
Achillea rydbergia	forb	native	190		Asteraceae	
Achnatherum hymenoides	graminoid	native	199		Poaceae	

4. Use the Query Wizard under the Create tab to Find Unmatched species names between the newly imported Data Table and the Plant Associations data table.



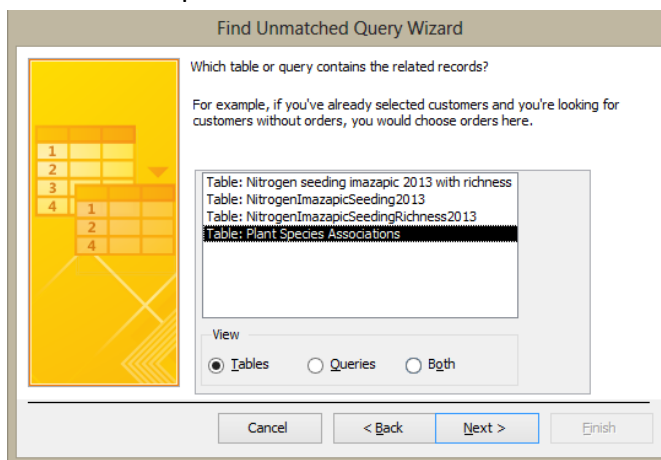
Select Find Unmatched.



Choose the Data Table you are checking.



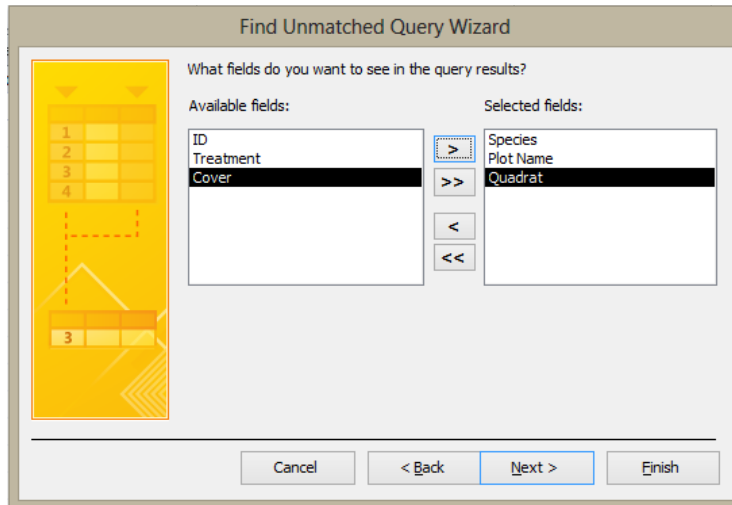
Select Plant Species Associations.



Select species for both tables being queried.



Select the Fields that will make locating the unmatched entry possible.



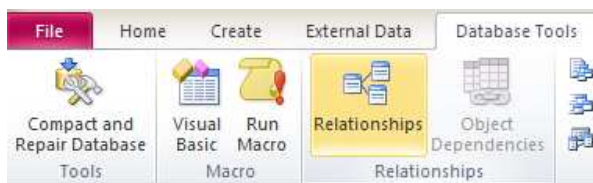
Access outputs the following table containing all unmatched species names.

Canada thistle 2013 with richness Without Matching Plant Species			
Plot Name	Treatment	Quadrat	Species
*			

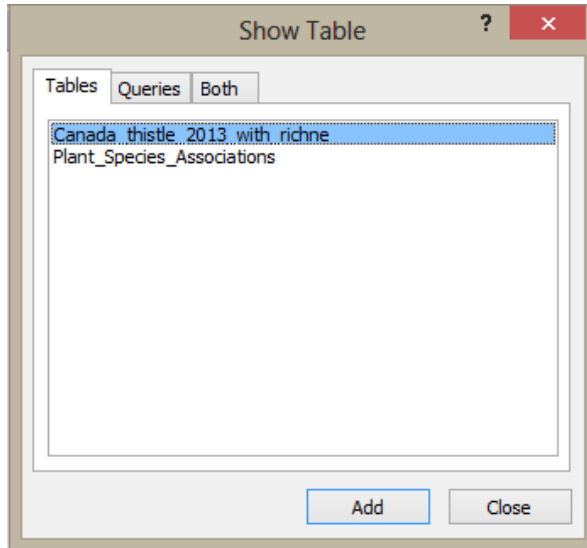
This effectively checks for misspelled or new plant species names and allows you to correct or add species and plant association information to Plant Associations as needed. This step is crucial, since Access will omit any plant species names and their associated data in later steps if there is not an exact match between the Data Table and the Plant Associations Table. Also, any new species that have not been observed or entered in previous monitoring seasons can be added to the Plant Associations Table at this point.

5. If the data you have imported is brand new (i.e. data from the first year of a study), the relationships between the Data Table and the Plant Associations table must be identified so they can be cross-referenced by Access.

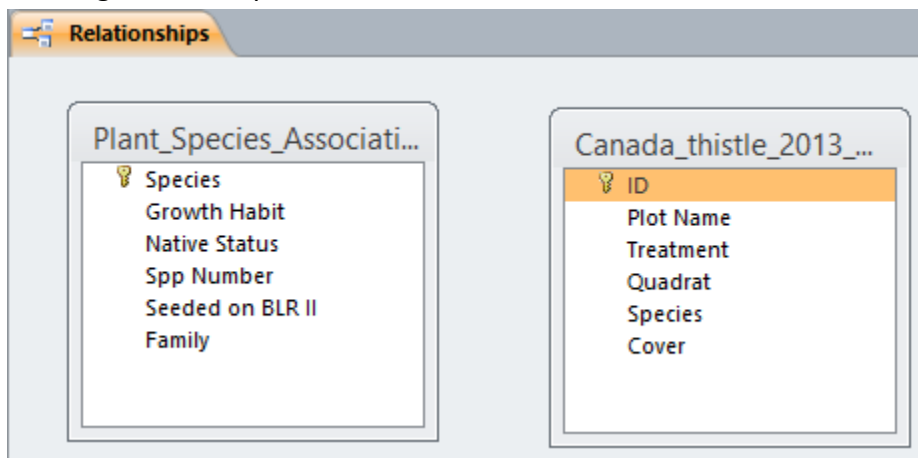
Under the Database Tools tab, click Relationships.



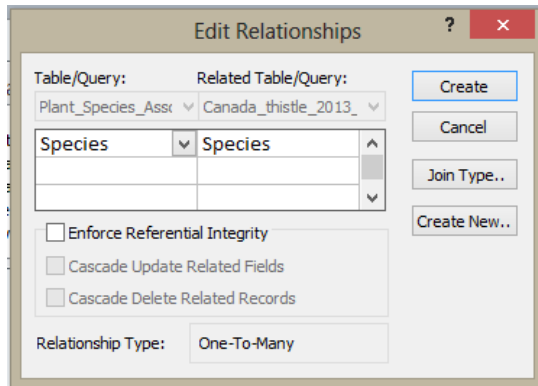
Any tables that exist in the database will be displayed in a pop-up window. Add the New Data Table (Canada thistle... in this example) to the relationships. If using the Vegetation Monitoring Access Database, the Plant Associations table should already be added to relationships.



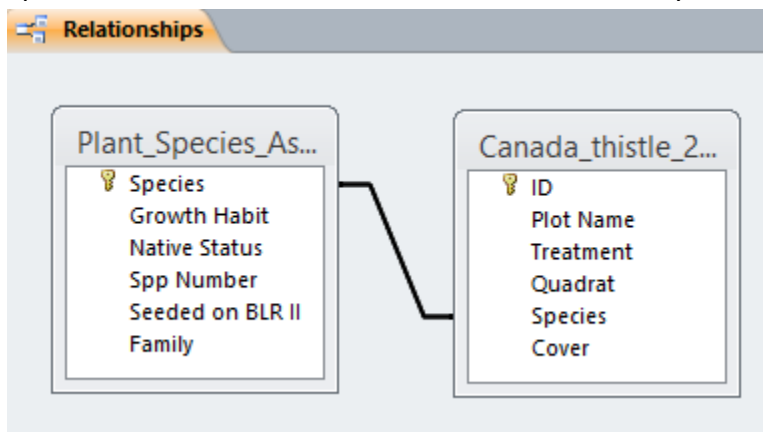
The relationship tables you create will look like this and show the column headings in the respective data tables.



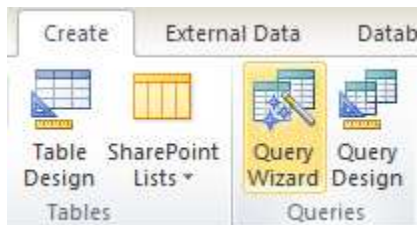
Identify that the species information from the plant associations table is related to the species information in the New Data Table by clicking Species in the Plant Associations table and dragging and dropping to Species in the New Data Table. Click Create in the resulting window.



Species information can now be cross-referenced by Access.



6. Use the Query Wizard to run a Simple Query Wizard between the New Data Table and the Plant Species Associations table to group species data by plot, treatment, native status and growth form, or any combination of the attributes you would like your data to be grouped by.



You can select what data columns you would like cross-referenced from each table using the drop-down Tables/Queries menu and the Available Fields. The Species field should be selected from both Tables to ensure correct groupings based on cross-referencing. When fields with the same name are added (in this example, Species) the name of the table that field originated in is also shown in Selected Fields. In this Example we are asking Access to group the Canada thistle

cover data for each plot and treatment type by the growth habit and native status associations indicated in the Plant Associations table.

Simple Query Wizard

Which fields do you want in your query?
You can choose from more than one table or query.

Tables/Queries
Table: Plant_Species_Associations

Available Fields:
Spp Number
Seeded on BLR II
Family

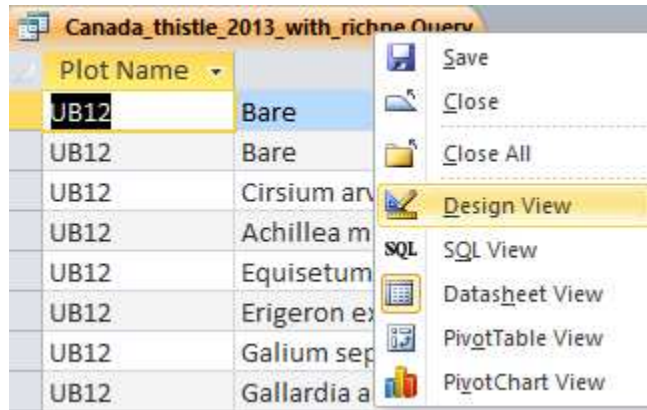
Selected Fields:
Canada_thistle_2013_with_richne.
Plot Name
Treatment
Cover
Plant_Species_Associations.Species
Growth Habit
Native Status

Cancel < Back **Next >** Finish

Access outputs a data table such as this, combining the data from the Data Table and the Plant Associations table. Rearrange data columns by dragging and dropping column headings. With Plot Name as the first column, you can right click the column heading and select order A->Z ascending, which will group all of the data by plot name.

Canada_thistle_2013_with_richne Query						
Plot Name	Canada_thistle_2013_with_richne_Species	Treatment	Cover	Plant_Species_Associations_Species	Growth Habit	Native Status
UB12	Moss/lichen	treatment	0	Moss/Lichen	ml	moss
UB12	Rock	treatment	0	Rock	rk	rock
UB12	Litter	treatment	7.5	Litter	ltr	litter
UB12	Graminoids	treatment	82.5	Graminoids	graminoid	native
UB12	Phleum pratense	treatment	7.5	Phleum pratense	graminoid	invasive
UB12	Lepidium densiflorum	treatment	2	Lepidium densiflorum	forb	native
UB12	Litter	treatment	37.5	Litter	ltr	litter
UB12	Rock	treatment	0	Rock	rk	rock
UB12	Taraxacum officinale	treatment	2	Taraxacum officinale	forb	non native

- Right-click the query tab and select design view.



The design view table allows you to fine-tune all aspects of the query output datasheet. Right-click anywhere inside the design table and select Totals from the pop-up menu. The Totals row is added. Using this function you can sum the vegetation attribute data for each species while they remain organized by plot and growth habit, etc. In this example, Sum is selected for the Cover attribute by clicking on the cell that says “Group By” in the Cover column.

Field:	Species	Plot Name	Treatment	Cover	Plant_Species_Associa	Growth Habit	Native Status
Table:	Canada_thistle_2013_	Canada_thistle_2013_	Canada_thistle_2013_	Canada_thistle_2013_	Plant_Species_Associa	Plant_Species_Associa	Plant_Species_Associa
Total:	Group By	Group By	Group By	Sum	Group By	Group By	Group By
Sort:		Ascending	Ascending			Ascending	Ascending
Show:	<input checked="" type="checkbox"/>	<input checked="" type="checkbox"/>	<input checked="" type="checkbox"/>	<input checked="" type="checkbox"/>	<input checked="" type="checkbox"/>	<input checked="" type="checkbox"/>	<input checked="" type="checkbox"/>
Criteria:							
or:							

Under the Design tab for query tools, select Run. Access sums all species data for that attribute and outputs a new query data table. What Access has done is find any Species that was observed multiple times within a plot, and summed the individual data values.



The resulting species data are now:

- Grouped by Plot, Growth Habit, and Native Status.

b) Summed by species within Plot, Growth Habit, and Native Status.



8. Remember that the species attribute values that were summed by Access are *still only summed values for that plot*. You will need to calculate the average species value by *dividing the summed species plot values by the number of subplots* in the monitoring plot. For example, if *Solidago canadensis* has a total cover of 12% in the three subplots of Plot A, the mean cover is $12/3=4\%$ cover.
9. Export this new query table to Excel (under External Data tab) and save.
10. Some projects will have a series of automated Excel tables that will allow you to paste in the new Excel output for that years data and automatically produce a table containing mean species plot values which are then grouped by functional group. These files are saved with file names with "Step 1", "Step 2", etc. prefixes so the order of their use is clear. New projects will require new automated tables to be created, since these automated tables rely on subplot numbers that can change with each project.

A4 Sample Data Sheets

A4.1 Sample Circular Nested Plot Data Sheet

Site Name:

Field Data Collection Date:

Data Collectors:

Elevation:

CNP Center Point

Coordinates Datum: NAD83

Easting (X):

Northing (Y):

Notes:

[illegible]

Site Name:	Transect GPS Coordinates NAD 83 Datum	Notes:
Treatment: (if applicable)	Easting (X):	
Data Collection Date:	Northing (Y):	
Data Collectors:		
Baseline ID: (if applicable)	First quadrat placed at: (e.g. 2 m)	
Transect ID:		

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4.3 Sample Transect Data Sheet for Point Intercept Cover Data

Site Name:

Treatment: (if applicable)

Data Collection Date:

Data Collectors:

Baseline ID: (if applicable)

Transect ID:

Transect GPS Coordinates NAD 83 Datum

Easting (X):

Northing (Y):

First point placed at: (e.g. 2 m)

Notes:

[illegible]

4.4 Sample Transect Data Sheet for Line Intercept Cover Data

Site Name:

Treatment: (if applicable)

Data Collection Date:

Data Collectors:

Baseline ID: (if applicable)

Transect ID:

Transect GPS Coordinates NAD 83 Datum

Easting (X):

Northing (Y):

Notes:

[illegible]

A5 Monitoring Plot Terminology

The following definitions are provided to clarify the usage of these words in this document. Other sources may use these words in a slightly different or interchangeable manner.

Plot/Monitoring Plot- A general term for a monitoring unit that contains sampling units or subplots. Examples of monitoring plots include circular nested plots (CNPs), single line transects, and baseline transects.

Transect- A line along which sampling units are placed at regular intervals. Transects can be installed as standalone units or along a baseline.

Baseline- A line along which transects are placed at regular intervals. Sampling units are then placed along each transect. Typically transects along a baseline are treated as independent plots.

Subplot- A single measurement unit within a monitoring plot. Examples of subplots include quadrats along a transect or within a CNP. Values for subplots are averaged over the entire monitoring plot.

Sampling unit- Similar to subplots, but also includes points along a transect when using the point-intercept cover method as well as transects using the line-intercept cover method.

Quadrat- A four-sided frame used to identify an area from which vegetation data are collected. Quadrats placed along a transect or in a CNP are subplots of those plots. (Many sources refer to quadrats as plots. This document avoids this usage for the sake of clarity.)

Interspersion- Generally refers to the even distribution of monitoring plots and sampling units across a study area. As defined, there are two levels of interspersion: the overall distribution of plots throughout the study area and the distribution of treatment types across those plots. See Section 2 for a detailed discussion of interspersion.

Independence- Plots that are separated by enough distance that they are not affected by treatments or vegetation changes on adjacent plots are independent. There is not a strict mathematical method of determining independence, and several guidelines to achieving plot independence are discussed in Section 2.