Identification of Potential Monitored Elements and Sampling Protocols for Terrestrial Arthropods

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Abstract

Terrestrial arthropods are the most diverse group of organisms in forests and play an essential role in ecosystem functioning. However, patterns of biodiversity in the terrestrial arthropod component of Alberta's forested regions remain largely unstudied. A range of natural resource management practices, including forestry, fire suppression, and petroleum development, affect biodiversity in Alberta. The monitoring of arthropod biodiversity patterns at different spatial and temporal scales should yield useful information for developing natural resource management options. However, a good monitoring program must have clearly articulated objectives, be carefully planned, and be executed in a rigorous standardized manner.

This document outlines the steps required to implement a terrestrial arthropod monitoring program including the need for long-term planning and the resources required to achieve success. Steps addressed are: an introduction to monitoring of terrestrial arthropods; selection of taxa to monitor; data integration with a seven-step analysis package; a synthesis of the sampling protocols and logistics appropriate for monitoring; detailed costing for time, equipment, processing and storage of data, archiving target groups and residues; the infrastructure needed to complete the entire field and laboratory segments of a project; data management considerations; scenarios for a recommended pilot project; and suggestions for longer-term research.

Introduction

Canada ratified the Convention on Biological Diversity (United Nations Environment Programme 1992) which requires each contracting party to identify components of biodiversity important for conservation. The ecological, economic and social importance of sustaining forest ecosystems has been clearly outlined in several documents (e.g., Canadian Biodiversity Strategy 1995). In order to meet the stated goals of sustainable management and retention of biodiversity, ecological research and biomonitoring are needed. We currently assess the effects of disturbance by a patchwork of biomonitoring programs (Rosenberg 1998); none of, which are well, developed to look at terrestrial forest arthropods (Winchester 1997b; Winchester and Ring 1999). Biomonitoring can be defined as the systematic use of living organisms or their responses to perturbations in an effort to determine environmental quality (Rosenberg 1998). A wide variety of biotic groups can be used in a biomonitoring program as has been documented for aquatic invertebrates (e.g., Hellawell 1991; Rosenberg and Resh 1993). Progress integrating terrestrial forest arthropods into monitoring programs has been relatively slow. A survey of the literature using the BIOSIS database, search engine Biosystematics and Life Sciences Resources (Biodiversity) from 1993 to 1999 using the following terms: terrestrial, arthropods, monitoring, indicators, and boreal forests, did not list any reference material that was not already included in this report.

In temperate forests, a range of natural resource management practices, such as forestry, urbanization, and petroleum development affect terrestrial arthropods (Vlug and Borden 1973, McIver et al. 1992, Niemelä et al. 1993; Roland et al. 1997; Spence et al. 1997, Winchester 1997b). Present knowledge shows clearly that most invertebrate species are affected, to some degree by habitat change. All species are probably affected, either directly or indirectly, by natural and anthropogenic disturbance, although not all species will be adversely affected (Redford and Richter 1999). Efforts to ascertain which species are affected by which disturbance types, and to estimate the magnitude of biodiversity change (May 1986, Myers 1988), must include the documentation of long-term trends in populations and community assemblages.

Biological diversity, considered at the species level, is a function of both species richness (number of species) and abundance (evenness, number of individuals) (Gaston 1996). All of the protocols presented in this report provide some qualitative/quantitative measure (species richness) when applied at a site, and if applied in a consistent, replicated manner, can provide data suitable for comparative analyses (i.e., of site, ecozone, or other stratum) (see Biological Survey of Canada 1994, Winchester 1997b, Finnamore et al. 1998). Most of the protocols also provide measures of relative abundance in the form of relative trapability of organisms, if sampling is performed in a consistent and replicated manner. In addition, a few of the protocols also provide scaleable abundance measures for each species. The protocols presented in this report are designed to give maximum flexibility in sampling terrestrial forest arthropod communities while providing adequate resolution to examine change at different spatial and temporal scales.

Improved sampling methodology has enabled researchers to incorporate taxonomic inventories into broader-based ecological programs, providing detailed information at the species level. Biodiversity exists within species, populations, individuals, and genes. Higher taxonomic classification is a human construct that reflects evolutionary relationships, and any biological reality they possess is usually accidental. Meaningful arthropod monitoring programs require detailed knowledge at the species level (see Danks 1996). A comprehensive terrestrial arthropod monitoring program should consider the following two points: (1) Sampling techniques to capture arthropod groups that characterize the diversity associated with different habitats at different spatial scales (i.e., sample site - landscape level). (2) Avoidance of single-species or indicator-species approaches (see Lawton et al. 1998). Attempts to understand ecosystem change using one, or a limited number of indicator species may be misleading (see Lawton et al. 1998), although other studies point out the utility of the indicator-species approach (McGeoch 1998).

Selection of Taxa for Monitoring

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Because detailed study is costly, many workers have tried to identify “indicators” — characteristic or surrogate species or sets of species — that could serve as a “magic bullet” to describe and evaluate ecological conditions (see McKenzie et al. 1990; McGeoch 1998). Valid indicators would greatly reduce the need to cope with the true diversity. Unfortunately, the criteria used to select indicators are seldom based on objective information. Scientific findings or methodology generally has not supported specific indicator choice, although useful procedural steps for justifying the choice are given by McGeoch and Chown (1998) and Dufrêne and Legendre (1997). At present there is little evidence to suggest that any taxon or group of organisms qualifies as a universal biodiversity indicator. This is true from a broad faunal viewpoint, but also for more specific questions. For example, in the study by Neave (1996), species confined to old-growth forests were present, but not among the carabid beetles, the group most often studied in such habitats in Canada. Results for one taxon do not predict results for another over different spatial scales (e.g. Lawton et al. 1998; Prance 1994; Williams and Gaston 1994). This finding is not surprising, because ecological patterns and processes depend on scale and different groups differ in local species richness, endemism, habitat association or specificity, vagility and so on (e.g. Prendergast et al. 1993). Some insects are relatively insensitive to environmental factors of potential interest (e.g. Whitford et al. 1999). In other words, ecosystems are complex, so that multi taxa approaches (e.g. Hammond 1994) are more realistic.

Given these difficulties, there is no clear consensus as to which groups or species best allow environmental changes to be assessed. Few attempts have been made to contrast attributes that might confer susceptibility or resilience to habitat change. Individual species responses are the key to understanding and measuring these impacts, but species so far examined in biodiversity studies in northern temperate forests might be atypical of the majority of invertebrates because they are taxonomically convenient (e.g., carabid beetles: Huhta 1971; Holliday 1991; Niemela et al. 1993; Spence and Niemela 1994; Spence et al. 1997), conspicuous (e.g., butterflies: see Davidar et al. 1995), broadly distributed geographically (see Platnick 1991; Danks, 1993, 1994), or have specific habitat requirements (cf. Danks 1979). Moreover, experiments have not always been designed to resolve the effects on local biodiversity of factors such as habitat loss, degree of spatial or temporal isolation, and dispersal capabilities, which influence different taxa to widely different degrees.

Based on reviewer's suggestions and consultations with entomologists, the suggestion for monitoring arthropod elements project is:

- Use a multiple trap approach (see text) to effectively sample the greatest number of arthropods (e.g., habitat and functional group associations). From these samples target taxa will be selected for further analysis. The remaining arthropods should be stored and properly curated as residues since they are likely to hold the answer to many questions. Because we do not necessarily know in advance which group might be suitable to seek answers, long-term preservation of residues is key, especially since biodiversity concepts, questions and analytical tools evolve over time.

Initial target taxa are:

- All Coleoptera, with species identification initially proceeding for species in the families Staphylinidae, Carabidae, Scolytidae, and Cerambycidae.
- All Hymenoptera, with species identification initially proceeding for species in the superfamilies Siricoidea, Tenthredinoidea, Apoidea (Apiformes, Spheciformes), Chrysidoidea, and Vespoidea. Species identifications should also be considered for the belytine Diapriidae.
- All Arachnida identified to species.
- All Hemiptera, with species identification initially proceeding for species in the family Cicadellidae. Species identification should be considered for the families Miridae, Lygaeidae and Cercopidae.

The arthropods listed above will provide taxonomic and functional (e.g., ecological roles/habitats) information that will be used to monitor the effects of environmental impacts on species, populations and communities in the forested regions of Alberta. This data can be used to address questions concerned with fragmentation, disturbance, habitat modification and ecological disruption (see Kremen et al. 1993).

Site Selection

The AFBMP technical and steering committees will choose the selection and number of sites. Recognition of the need for reference or benchmark areas is acknowledged. For example, refer to Chapter 4 of this volume (Lee and Herbut 200?).

Specific Goals

This chapter provides the information needed to monitor patterns of arthropod biodiversity at different spatial and temporal scales. Detailed goals, questions and hypotheses of the overall project are provided in Chapters 1 and 2 of this volume (Farr et al. 200?a; Farr et al. 200?b).

Integration and Data Analyses

To integrate terrestrial arthropod monitoring protocols with monitoring protocols for other flora and fauna it is recommended that the following steps be used. These steps provide suggestions for a logical way to proceed with data analyses and are not meant to preclude other avenues of investigation and analysis. Statistical programs are often one of personal choice and those listed below are used in the arthropod biodiversity work at the University of Victoria. Several other comparable programs exist and can be used to analyze terrestrial arthropod biodiversity data.

A seven-step package of analytical techniques is proposed that could be specially tailored for analyzing terrestrial arthropod data. If possible, all of the steps should be completed with detailed information at the species level being of most value.

Coarse-grained

1. Total arthropod density
2. Total arthropod biomass and energy cycling
3. Trophic group analysis for all arthropods
4. Species richness and estimated total regional biodiversity of target taxa
5. Changes in trophic group proportions within target taxa
6. Species complementarity
7. Relating species composition to environmental gradients

Fine-

Each successive step adds an increasing level of complexity (and cost) to the analysis, but at the same time provides an increasing level of sensitivity to detect the impact of finer-scale disturbance events on biodiversity. The first three levels of analysis require arthropod sorting to ordinal- or family-level, but only give coarse-grained resolution. The other four levels of analysis require more detailed sorting to species-level for the designated target taxa, but provide fine-grained sensitivity to even minor environmental disturbances.

Total Arthropod Density

Arthropod samples are initially sorted to Class, Subclass, Order and Family, all individuals are counted, and then each taxonomic grouping is placed in a separate specimen vial. Given equal sampling effort and at least three traps per site, the mean number of arthropods within each taxon can be compared spatially across groups of sites, and also monitored from year to year to detect changes. Statistical analysis is by ANalysis Of VAriance (ANOVA) (Sokal and Rohlf 1995). Ideally such comparisons would be based on the density of individuals per unit area, however few activity-based arthropod trapping methods are capable of providing this type of data. A basic assumption in this analysis, then, is that equal trapping effort (number of trap days) allows direct comparison of relative abundance between groups of sites and between years.

For monitoring purposes, a more robust method than ANOVA for detecting trends in arthropod abundance from year to year is to use time series analysis. Time series analysis will require at least five years of data. As with most of the other analyses considered below, a minimum of three to five years of data are required to form the baseline for a long-term monitoring program. Arthropod populations may be naturally highly variable from year to year (Southwood 1968).

Proportional representation of taxonomic groups can be monitored spatially and temporally as well. Proportional representation is calculated by dividing the abundance of each taxonomic group by total arthropod abundance. Statistical analysis is by log-linear analysis (Sokal and Rohlf 1995). After severe disturbance events there can be a major shift in the proportional representation of certain forest arthropod groups, e.g., Collembola and Acari (Vlug and Borden 1973). However, proportional changes in higher taxonomic groups are relatively insensitive to minor disturbances (pers. obs.).

Total Arthropod Biomass and Energy Cycling

Arthropod biomass is a much better indicator of rate of energy utilization and cycling through the ecosystem, and potential available energy for insectivorous organisms, than is the abundance of arthropods. Dry-weight arthropod biomass can be accurately estimated from known body length to weight ratios available in the literature (e.g., Rogers et al. 1976, Marcuzzi 1987). From a representative sample of arthropods from each site, draw *at random* a sub-sample of 100 individuals from each taxonomic group and measure the body length of each individual to the nearest 0.1 mm using an ocular micrometer on a microscope. From this body length, calculate dry weight using the mathematical equation for that taxonomic group. To calculate total arthropod biomass within each taxonomic group, simply multiply the average dry weight biomass per individual in the sub-sample by the number of individuals in the total sample. Ideally this should be calculated separately for each sample because the size distribution of arthropods may change with season and site. Spatial and temporal changes in arthropod biomass can be analyzed statistically using ANOVA and time series techniques. Again, these data will be useful for detecting coarse-grained changes in arthropod biomass, energy flow and prey availability following major disturbance events.

Trophic Group Analysis for All Arthropods

Assigning arthropods to different trophic groups adds an important component to the analyses, contrasting absolute and proportional changes in both abundance and biomass at different levels within the forest food web. For example, forest fragmentation has been shown to affect the relative proportions of different arthropod trophic groups in tropical forests (Didham et al. 1998b).

Recognized trophic groups are: fungivores, herbivores, predators, saprophages (scavengers), xylophages (wood feeders), xylomycetophages (specialists on “ambrosia” fungi inside wood), and parasitoids (Hammond 1990).

Once arthropods have been sorted to family-level they can be coarsely assigned to trophic feeding groups based on their taxonomic classification. Some arthropods can confidently be assigned to trophic groups based on their taxonomic Class (e.g., all millipedes are scavengers, and all spiders are predators). In other more trophically diverse groups, such as beetles (Coleoptera), trophic groupings have been well characterized to the Family and Subfamily levels by Hammond (1990) and others. Inevitably, however, at this level of analysis there will be some margin of error in assigning trophic groups (e.g., Elateridae, click beetles, are approximately one-third herbivores, one-third predators and one-third xylophages, but there is no way of telling without identifying the individual species, in which cases individuals can be assigned at random to one of the three groups).

These data provide a more detailed picture of spatial and temporal changes in the abundance and biomass of arthropods by focusing on individuals in different trophic groups within the food web (see also Didham et al. 1996). Proportional changes within trophic groups can be monitored using log-linear analyses (Sokal and Rohlf 1995).

Species Richness and Estimated Total Regional Biodiversity of Target Taxa

Once arthropods have been counted and a sub-sample of each taxonomic group measured for body length, the majority of specimen vials can be archived. Target taxa designated for sorting to species level can then be processed by taxonomists. The aim in species sorting is to produce a list of identified (or at least coded) species or recognizable morphospecies and their abundances in each sample. It is critical that all samples are cross-referenced so that the identities of species at any particular location are checked against an appropriate reference collection.

The total number of different species collected from a site during the sampling season is the species richness, *S*, for that site. Species richness is perhaps the most readily comprehensible expression of the diversity of a site. However, *S* is greatly affected by sample size (Southwood 1978) so that a larger sample of arthropods taken from one site will intrinsically contain a greater number of species, even though there may be no true difference in species richness between sites. To control for sample size, Sanders (1968) devised a technique known as "rarefaction" which calculates the expected number of species in a sample of a given size (a form of sub-sampling).

Indices of species diversity, notably the Shannon-Wiener index (Shannon and Weaver 1949), have been widely used in biodiversity studies without regard for their biological significance. The Shannon-Wiener index, in particular, has been variously described as a dubious index of doubtful biological interpretation, poor at site discrimination, and more of a distraction than an asset (Hurlbert 1971, Goodman 1975, Southwood 1978, Taylor 1978, Magurran 1988). Rarefied species richness is far simpler to calculate and more readily interpretable than any of the species diversity indices available.

The next step following species-level sorting is to compute a randomized species accumulation curve, giving the accumulated number of species with increasing sampling effort at each site. Usually these curves are plotted with number of individuals captured along the x-axis and number of species captured along the y-axis. Alternatively, curves can be plotted with number of trap-days sampling effort along the x-axis to show the trapping effort required to accumulate species of that target taxon. Initially the curve rises rapidly with sampling effort as most of the common species are caught, and then reaches an asymptotic plateau where the addition of each new species takes an increasing amount of effort.

Fitted curves can be used to estimate the accumulated number of species at which the curve plateaus, and hence the estimated total local biodiversity within the target taxa (given unlimited sampling effort). Samples from all sites within an ecozone can be combined to calculate a species accumulation curve for the entire ecozone, and hence to estimate total regional biodiversity for the target taxon. Similarly, species accumulation curves can be calculated across years.

Characteristically, the total number of species in an area is the measure that most people consider as "biodiversity". While this is partially true it is also important to note that it can be a red herring, because the identities of the species involved are vitally important. Forest species are more likely to be in need of conservation monitoring and protection than widespread disturbance-adapted species (Didham et al. 1998a).

Changes in Trophic Group Proportions within Target Taxa

For target taxa which are trophically diverse (e.g., beetles), appropriate trophic designations can only be accurately made following species sorting. Thus, species richness within specific trophic levels can be monitored from year to year. This can be useful, for example, in cases where disturbance has been shown to alter predator to prey ratios in fragmented forests (Didham et al. 1998b).

For species-level trophic assignments, all species within a Family can be easily assigned if only one feeding-biology is known for that Family. In cases where multiple feeding biologies are known to occur within a Family, species can be assigned individually based on mouthpart and general morphological characters, as well as published details of the feeding biology of the genus, or of related genera. Statistical analysis is by log-linear analysis (Sokal and Rohlf 1995).

Species Complementarity

The simplest way to monitor a change in species composition over time is to use a species similarity index and compare the similarity of species composition from one year to the next. It is important that sampling effort be roughly the same each year. In a thorough comparison of species similarity indices Wolda (1981, 1983) found that the only index not greatly affected by sample size or the diversity of the sample is the Normalized Expected Species Shared (NESS) index developed by Grassle and Smith (1976). NESS is a quantitative similarity index (taking account of both the number of species as well as their abundances) and can be calculated with confidence intervals for hypothesis testing, making it far superior to all other similarity measures (Wolda 1983).

Spatially, mean species similarity can be compared between groups of sites using ANOVA. The inverse of species similarity is obviously the degree of dissimilarity, or species turnover, between groups of sites. The greater the physical distance between sites, or groups of sites, the lower the value of species similarity. Because of random sampling effects it is important to remember that no two samples (even if taken from the same site) will ever be exactly the same. Intrinsically, there will always be some degree of sampling variance in measures of species similarity, so comparisons between sites are relative, not absolute.

For monitoring purposes, the same caveats regarding sampling variance apply, and each successive year's species composition should be compared with that of the first year of sampling. Hence, the first year's sample becomes the baseline against which future changes in species composition are measured. If sample sizes for the target taxon are small, then year to year species similarity may only be 50-60% due to intrinsic sample variance. However, it is the relative change in this value over time that is important, not the absolute value. It is important that sampling effort is approximately equal each year. Temporal data can be analyzed using regression techniques, with time (years) on the x-axis and similarity to initial species composition on the y-axis. At least three to five years of data are required to fit regression lines.

Relating Species Composition to Environmental Gradients

The most comprehensive class of analytical techniques available for the analysis of multivariate arthropod data is community ordination. In principle, community ordination techniques identify key elements in the species composition and species abundance patterns of different sites and attempt to group these sites based on variation in the association of species. Sites are associated in multidimensional ordination space. Simplistically, sites are ordered along a primary axis in terms of their level of species-abundance similarity. Axis 1 explains the greatest amount of variation in species-abundance patterns between sites. Sites are then secondarily ordered along axis 2, which explains the next greatest amount of variation in species-abundance patterns between sites, and so on for multiple axes. Different ordination techniques vary depending on whether they are *direct gradient* (incorporating environmental data directly into the analysis) or *indirect gradient* analyses (species relationships to environmental variables are measured subsequent to ordination, in an *ad hoc* manner), and in their assumptions regarding species responses to environmental gradients. Early ordination techniques, such as Principal Components Analysis (PCA), assumed a linear relationship between species responses and environmental gradients, whereas more recent ordination techniques make the more reasonable assumption that responses are non-linear (e.g., Detrended Correspondence Analysis, DCA). However, these techniques are indirect gradient analyses. By far the best available ordination method is Canonical Correspondence Analysis (CCA) which assumes non-linear responses to environmental gradients and is a direct gradient method, allowing any number of environmental variables to be incorporated into analyses. Variation in species-abundance patterns between sites can then be related directly to variation in environmental gradients, including habitat disturbance (Jongman et al. 1995).

The CCA computer package CANOCO (Ter Braak 1986, 1988) could be used for analysis. A list of species and their abundances at each site are entered into the program, along with a list of environmental measurements for each site.

CANOCO is a multivariate technique to relate community composition to known variation in the environment. It provides an integrated description of species-environment relationships by assuming the existence of a single set of underlying environmental gradients to which all species respond (Ter Braak 1986, Jongman et al. 1995). CANOCO uses a forward selection procedure to rank environmental variables in order of their importance for determining species composition, in much the same way as forward stepwise multiple regression (Ter Braak and Verdonschot 1995). Most importantly, in terms of monitoring arthropod species composition, time in years can be added as an environmental variable along with other more conventional variables such as temperature or amount of forest cover in the landscape. CANOCO will identify whether there is significant variation in arthropod species composition from year to year taking all sites and environmental conditions into consideration. Confounding environmental factors (such as temperature variation from year to year) can be factored out by adding all significant environmental variables into the analysis as co-variables, and focusing solely on species-abundance changes through time. These analyses can be performed within specific sites or specific ecozones to look for temporal changes at a certain spatial scale, or more broadly to see if there are general changes in arthropod species composition across the whole province. Didham et al. (1998a) provide good examples of CCA ordinations and partial-CCAs to control for co-variables in the analysis of arthropod species responses to forest fragmentation.

Analysis and presentation of data using CANOCO is technically complex, but once the data are formatted correctly and the techniques mastered, analysis-using CANOCO is flexible and powerful. CANOCO provides the most robust statistical analysis of multivariate arthropod community data and allows unparalleled integration of arthropod and other faunal data, plant data and environmental measurements across numerous sites. Monitoring temporal changes in biodiversity is confounded by numerous environmental factors beyond the control of the investigators, but CANOCO can effectively separate this confounding variation to identify any true change in biodiversity over time.

Survey Design

A well thought-out sample design (e.g., scale, replication) must be implemented in order to gather meaningful data that can be extended to answer monitoring questions. There are numerous examples of relationships between insect diversity, forest structure and abiotic parameters that serve to structure arthropod communities. It is not the purpose of this report to itemize these relationships, however it should be noted that if the broad-based protocols, sample frequency and recording of abiotic elements are undertaken then the prescribed analysis presented in this section will give maximum flexibility and resolution to monitor change at sample sites and ecozones.

In order to achieve maximum efficiency, a monitoring program should be fully designed before any field work is initiated. Previous arthropod biodiversity studies have generally been the purview of either of two scientific disciplines: systematics or ecology. Systematic work tends to be highly proficient in maximizing an estimate of species richness and relating data in the form of sample catch per unit effort. Usually the end result of this "museum collecting" (see Coddington et al. 1991) is a list of species encountered. Conversely, under the umbrella of ecologically oriented sampling, quantification of habitat, richness and evenness measures offer objective methodologies for the comparison of true relative abundance and sampling effort. However, ecological methods are not as proficient in cataloguing the maximum number of species. Ideally, the desired monitoring design should be a combination of both sets of techniques. In this manner, inventory information can be maximized and quantified to answer basic questions about biodiversity (e.g., monitor species assemblage changes in response to habitat changes). It is important to note that simple species lists do not provide enough information to answer questions concerned with monitoring change.

Related Elements to Monitor

Many abiotic attributes of an ecosystem are crucial to comparative studies or to standardize background variables so that other parameters can be considered. Some attributes that can be positively associated with insect distribution and diversity have been incorporated into other programs and it would be beneficial for the Alberta monitoring project to establish linkages with the International Tundra Experiment (ITEX) or to the Long-Term Ecological Research (LTER) sites and their international counterparts (ILTER) in the United States and elsewhere. Abiotic measures which could be included in the monitoring program for terrestrial arthropods are summarized in Table 14.1.

Monitoring Protocols and Logistics

Introduction

Terrestrial forest arthropods (mainly insects, mites and spiders) have several biological features that predispose them to diversity in response to environmental heterogeneity (Southwood 1978). The ability of insects to exploit a variety of habitats coupled with their diversified behaviour means that few traps are equally efficient for the capture of different groups. With increasing scientific interest in terrestrial forest arthropods numerous sampling methods have been developed, but as Finnamore et al. (1998) noted it is important to standardize techniques in order to address similar questions at different locations. However, owing to the diverse nature of the taxa to be surveyed, and their varied habits and life cycles, no single survey method or sampling technique is appropriate to overcome the numerous challenges that arise when studying forest arthropods. For example, pyrethrum knockdown is a productive method for examining arthropod communities in trees and shrubs with respect to factors such as guild, biomass, body size, and species-abundance patterns (Stork and Hammond 1997), but this method would be unsuitable to study aspects of arthropod life histories, such as diel activity (Basset et al. 1997).

Depending on the taxa of interest, usually more than one sampling method should be used. The entomological literature comparing efficiency of different traps is large. This section will examine general concerns with ground-based sampling and itemize the advantages and disadvantages of each sampling method, as well as the rationale for its use. Since a wide range of techniques has been developed over the last several years this section will cover basic ground-trapping techniques that have been suggested by the Biological Survey of Canada and field tested in Canadian biodiversity studies of arthropods in forests. For an excellent review and additional information on invertebrate sampling see New (1998).

NOTE: It may not be logistically feasible to service traps every two weeks for a long-term, large-scale study. Tradeoffs with sampling frequency will be considered and modified where appropriate by the AFBMP.

Trap Protocols

Where applicable, information on trap protocols is reproduced with permission from Winchester and Scudder 1993, and Finnamore et al. 1998.

Malaise Traps

General Trap Information

The most common passive flight-intercept trap is an open-sided tent-like structure called a Malaise trap. Insects strike the central panel of the trap and either drop to the ground, or fly/crawl to the highest point. Those flying upward are contained by the roof and concentrated in the collecting head of the trap where they are killed. Generally the trap is dark in colour with a light roof which reinforces any positive response to light in diurnal insects. The collecting head is attached to the higher end of the trap to take advantage of insects moving to the highest/lightest area. Insects caught in the collecting head are killed by drowning in a solution of preservative.

Ground Malaise traps catch flying insects up to one metre above ground level, in or above surface vegetation. They are most effective at sampling arthropods along forest edges, but can be used in forest interiors, and in associated habitats such as forest wetlands. A common modification of a Malaise trap is the inclusion of a liquid-filled container below the trap along the middle panel to collect insects that drop to the ground after contact with the panel. The containers can be the same as those used for pan traps but they should be light gray in color and placed on top of the ground (i.e., not sunken in the ground). This ensures that insects intercepted by the Malaise trap that end up falling to the ground are kept separate from any ground trapping program.

Use

Malaise traps collect a diverse sample of primarily winged and vagile insects from the Orders Diptera (flies) and Hymenoptera (bees, wasps, ants). A variety of other orders are also caught in Malaise traps (e.g., Coleoptera). Traps tend to collect large numbers of non-habitat-associated species, however habitat associations at a larger scale can be determined (forest interior, forest edge).

Malaise Trap Equipment

Many Malaise trap designs are available (e.g., see Townes 1972) and could be used in a monitoring program. The Malaise trap design suitable for this project should closely follow the traps used by A.T. Finnamore in his grassland study at the Canadian Forces Base, Suffield. Fabric: reverse locknit tricot warp knit, ultraviolet stabilized polyester, fine mesh (11/cm wales, 10/cm courses) with individual fibre diameter about 40 microns.

- Colour: central and end panels black: roof white with black strips on open edges.
- Collecting head: translucent white plastic with fine-screen vent
- Poles: 2 per trap, wooden poles are recommended
- Cord: approximately 30 m per trap
- Stakes: metal stakes, 15 cm in length, 14 per trap

Malaise Trap Installment

The Malaise trap is suspended between vertical poles placed at each end of the trap. The poles are held upright and in place by 3 cords tied to stakes in the ground. The cheapest and strongest poles are made of wood that can be purchased from local hardware stores. Each of the four corners and centre panel should be staked to the ground. Each of the four elevated corners can be tied with cord to nearby trees or poles.

Malaise traps are most efficiently installed by two people. Malaise trap efficiency also varies with aspects of installation. Factors such as likely flight paths, prevailing winds, and Malaise head position can affect both the numerical and taxonomic composition of the catch. The following points should be adhered to when setting up Malaise traps. (1) The collecting head (high end of the trap) should be oriented consistently with the head pointing to the lightest part of the forest habitat; toward the open area (clearing or water) when placed in a forest edge. (2) Record the compass direction of trap orientation and use it, where possible, to standardize trap placement. (3) Shrub vegetation directly beneath the trap should be cut back to a standard height of several centimetres above the surface. (4) Perturbations by large mammals (e.g., bears) can be a problem with Malaise traps.

Field Installation Protocol

- Suspend the Malaise trap between vertical poles placed at each end of the trap.
- Support the 2 end poles using 3 cords tied to one of 3 stakes hammered into the ground to form a triangular support. Trees or shrubs, if available can be used instead of stakes to anchor and support the end poles.
- Tie the sides to the lateral stakes used for end poles.
- Adjust the tension on the cords so that no part of the trap sags.
- Place gray pan traps on top of the ground along the bottom of the centre panel. Do not sink pans into the substrate.
- Fill the Malaise head and pans about 3/4 with propylene glycol mixture.
- Place an identification marker containing the site and trap numbers (in water proof ink or pencil) near or on the trap.

Trap Servicing

Service the trap by removing the Malaise head and screwing on the Malaise trap cap. A new pre-filled, Malaise head can then be added. An immersible label should be added to the Malaise sample. The label should have the collection date, trap number and site number.

Pan traps under the Malaise central panel are serviced by scooping out the contents with an aquarium net and then either reusing the old solution or replacing it with fresh solution. Before scooping remove large objects such as leaves, twigs, small animals, etc., that may have fallen into the trap. The net should be dragged gently near the bottom in one direction several times until the contents are recovered. Top up the pans with solution (if necessary). Samples from the Malaise pans may be pooled but should be kept separately from the Malaise trap catch. Pan trap labels should contain the same information as the Malaise trap and be associated with the Malaise trap catch.

Specimen Processing

Specimens collected in the field (avoid sloshing of specimens) can remain in the trap solution until they can be processed back at the laboratory. Each Malaise trap should be emptied into a sieve (approximately 140 micron mesh size) and rinsed thoroughly with tap water. Contents can then be transferred into whirl-pak bags, to which 80% ethyl alcohol is added to completely immerse the specimens. Samples should be double bagged. Fresh water washing is essential because the propylene glycol and dirt will form a film on the insects if they are placed directly in alcohol. This film is difficult or impossible to remove once specimens are in alcohol, thus rendering the specimens much less useful. Sample labels (immersible, written in pencil) must be placed inside the bag with the specimens. Change the alcohol in about 7 days, and if possible, refrigerate or freeze the samples to await further processing. Remember that all samples in storage should be double bagged.

Sampling Frequency

In order to fully sample any site or habitat, trapping should be continued throughout the active season, usually April to October in temperate latitudes. Such an extended trapping period is needed to accommodate the different phenologies exhibited by the various taxa. Traps should be serviced every 2 weeks. Refer to the section entitled "Recommended Pilot Studies and longer-term research" for the Malaise sample program prescription.

Further Reading

Malaise (1937), Roberts (1972), Townes (1972), Disney et al. (1982), Darling and Packer (1988), Marshall et al. (1994).

Pitfall Traps (unbaited)

General Trap Information

Pitfall traps are deep, small containers (cups) that are sunk into the ground to sample surface-active arthropods. They function in a manner similar to pan traps by collecting arthropods that fall into the container and drown in the collecting fluid at the bottom of the trap. Pitfall traps collect smaller samples of arthropods than pan traps because of the smaller trap perimeter of the pitfall trap. They are easier to install and service because of the smaller volume of collecting fluid needed to operate the trap, less expensive in materials to construct the trap, and less expensive in labour to process the smaller samples. Covered pitfall traps not only differ from pan traps in the size of the sample collected but also in the light response of the diurnal (day-active) species collected. Covered pitfall traps tend to collect diurnal arthropods with a negative response to light while pan traps tend to be neutral for diurnal surface-active arthropods.

The most common modification to pitfall traps is a cover. The cover creates a cave-like microhabitat that is attractive to surface-active diurnal arthropods with a negative response to light. The cover also protects the trap and its contents from climatic conditions like heat and precipitation, thereby providing some buffering capacity from decomposition of the sample. The cover should be opaque to avoid a greenhouse effect in the trap, and light grey in colour so as not to attract low-flying insects or to attract a different suite of species than that captured by yellow pan traps. The cover is made of ceramic tile, a material heavy enough not to be dislodged by wind, or affected by rain or sun, and unpalatable to termites. The cover is supported 2.5 cm above the pitfall trap by 4 galvanized nails or plastic rods. The supporting nails or rods should be environmentally neutral so as to minimize any effect on the soil chemistry and biota. Pitfall traps must be sunk into the ground flush with the top rim of the plastic cup. A propylene glycol mixture is placed in the trap and acts as the collecting medium and preservative.

Pitfall traps can be used in any ground level substrate except water and rock. They can be used to sample arthropods in a variety of microhabitats such as rotting wood, surface litter, or peatlands, but the most common application is in a soil substrate for surface-active arthropod fauna.

Use

Pitfall traps collect a diverse sample of primarily ground-dwelling arthropods, including Coleoptera, Collembola, Arachnida--Araneae, Acariformes.

Pitfall Trap Equipment

The Pitfall trap design suitable for this project should closely follow the traps used by N.N. Winchester in his long-term forest monitoring project at Rocky Point, British Columbia and A.T. Finnamore in his grassland study at the Canadian Forces Base, Suffield.

- Pitfall traps: 450 ml plastic cups (2 per trap). Cups must fit snugly inside each other with rims touching.
- Dimensions: 92 mm top diameter X 100 mm depth.
- Cover: 152 mm X 152 mm ceramic tiles (1 per trap).
- Tile colour: light gray (e.g., Munsell Book of Colours, Glossy Collection - N7.5 on the Neutral Value Scale, 50.7% reflectance).
- Cover supports: 152 mm galvanized nails or plastic rods (e.g., chopsticks, skewers) (4 per trap).

Pitfall Trap Installation

Pitfall traps should be sunk into the ground with top rim of the plastic cup flush with the soil or litter surface. If abundance measures are desired choose either litter or soil and use that surface consistently to standardize pitfall trap placement.

Field Installation Protocol

- Use a trowel to cut through and remove the root mat and soil to create a hole which should closely approximate the size of the plastic cup. Care should be exercised in removing soil and root mat material so as to minimize disturbance in the adjacent microhabitat. The excavated material must be removed about 10 m from the site to minimize disturbance. Some soil should be retained to fill around and level the top of the trap.
- The pitfall trap is placed in the hole and the outside surrounding edge filled with soil and carefully worked flush with the rim. When setting the trap, be careful not to puncture the container (i.e., avoid sticks or rocks in the excavated hole). Any material that falls into the trap during placement can be blown out, larger particles must be removed by hand.
- Place a second cup inside the first. It is important to select cups that fit snugly inside each other with rims touching since a second plastic cup is placed inside the first so that the outside cup will not be disturbed during trap servicing. Note that cups from different manufacturers are often of different dimensions. The outer container can then be left in the ground permanently, and the inner container removed for servicing.
- Insert 4 cover supports (e.g., galvanized nails) into the soil surrounding the trap leaving 2.5 cm of each support visible above the substrate to support the cover.
- Fill the traps about 3/4 with collecting fluid and place the cover on the supports.
- Place a marker containing the site and trap numbers (in waterproof ink or pencil) near the trap.

Trap Servicing

Service traps by removing the tile cover and lifting the inner cup, containing the sample, from the outer cup. Remove large objects such as leaves, twigs, small mammals, etc. that may have fallen into the trap (small animals can be retained for the vertebrate portion of the project). The contents of the cup are gently swirled to lift specimens off the bottom, then the entire contents are poured through an aquarium net, the liquid is collected in another cup to avoid microhabitat disturbance around the trap. The collecting fluid can then be reused. If reused, the liquid and the servicing cup can be placed back in the outer container of the trap. Specimens are transferred to plastic whirl-pak bags from the net using a squeeze bottle of water to remove the last specimens. Each trap should be processed separately and a label containing the site, trap identifiers, and date of collection placed in the bag. Contents should be double bagged. Once all the traps have been cleaned, and replaced, ensure that they are flush with the ground, are 3/4 full of collecting fluid and that the cover has been replaced.

Specimen Processing

Specimens collected in the field can remain in the trap solution (e.g., stored properly and in a cool environment) until they can be processed back at the laboratory. Processing should occur within one week of obtaining the samples—the sooner the better! Each pitfall trap should be emptied into a sieve (or a fine mesh aquarium net) and rinsed thoroughly with tap water. Contents can then be transferred into whirl pak bags, to which 80% ethyl alcohol is added to completely immerse the specimens. Samples should be double bagged. Fresh water washing is essential because the propylene glycol and dirt will form a film on the insects if they are placed directly into alcohol. This film is difficult or impossible to remove once specimens are in alcohol, thus rendering the specimens much less useful. Sample labels (immersible, written in pencil) must be placed inside the bag with the specimens. Change the alcohol in about 7 days, and if possible, refrigerate or freeze the samples to await further processing. Remember that all samples in storage should be double bagged.

Sampling Frequency

In order to fully sample any site or habitat, trapping should be continued throughout the active season, usually April to October in temperate latitudes. Such an extended trapping period is needed to accommodate the different phenologies exhibited by the various taxa. Traps should be serviced every 2 weeks. Refer to the section entitled "Recommended Pilot Studies and longer-term research" for the Pitfall trap sample program prescription.

Further Reading

Briggs (1961), Rivard (1962), Mitchell (1963), P.J.M. Greenslade (1964), Greenslade and Greenslade (1971), Gist and Crossley (1973), P. Greenslade (1973), Luff (1975), Newton and Peck (1975), Southwood (1978), Baars (1979), Anderson (1982), Dennison and Hodkinson (1984), Halsall and Wratten (1988), Dunn (1989), Niemelä et al. (1990), Clark (1992), Marshal et al. (1994), Spence and Niemelä (1994), and Scudder (1996b).

Pan Traps

General Trap Information

Pan traps are shallow, relatively large containers (compared to pitfall traps) that are placed on or sunk into the ground to sample arthropods. In their simplest form they function in nearly the same manner as pitfall traps, collecting arthropods that fall into the container and drown in the collecting fluid. Pan traps not only collect larger samples of arthropods than pitfall traps, they also differ with respect to the light response of the diurnal (day-active) arthropod species that are captured. Pan traps collect more specimens than pitfall traps because of the increased surface perimeter of the trap. Although both pitfall and pan traps are neutral with respect to the light response of nocturnal (night-active) species, they differ in the light response of the diurnal species captured. Covered pitfall traps collect diurnal, surface-active species with a negative response to light. Unmodified pan traps remain neutral for surface-active diurnal arthropods, collecting diurnal species with either positive or negative response to light.

The most common modification to pan traps is the colour. Insects, particularly flying insects, are differentially attracted to a variety of colours, but most are attracted to bright yellow. As a result pan traps are usually coloured bright yellow to attract a broad spectrum of low-flying insects. Pan traps are most effective if sunk into the ground flush with the top rim of the trap. For a discussion on specificity to colour response see Disney et al. (1992), and New (1998).

Pan traps can be used in just about any terrestrial habitat including water surfaces of wetlands, ponds, or lakes (floating pan traps) or suspended in tree canopies, but the most common application is in a soil substrate for surface-active fauna.

Use

Pan traps collect a diverse sample of ground-dwelling arthropods (similar to pitfall traps) but also collect a diverse sample of low-flying, vagile arthropods. Typically, ground-dwelling arthropods collected include Collembola, Coleoptera, Diptera, Arachnida--Araneae, Acariformes. Yellow pan traps are particularly effective for sampling vagile Hymenoptera and Homoptera.

Pan Trap Equipment

The Pan trap design suitable for this project should closely follow the traps used by N.N. Winchester in his long-term forest monitoring project at Rocky Point, British Columbia and A.T. Finnamore in his grassland study at the Canadian Forces Base, Suffield.

- Pan traps: plastic bowls.
- Dimensions: 273 mm diameter X 79 mm depth.
- Colour: Safety yellow in North America (Munsell Book of Colours, Glossy Collection - 5Y 8/12).

Field Installment Protocol

Pan traps should be sunk into the ground with their rims level with the soil or litter surface. If abundance measures are desired choose either litter or soil and use that surface consistently to standardize pan trap placement.

- Use a shovel to cut through and remove the root mat and soil to create a depression which closely approximates the size of the pan trap. Care should be exercised in removing soil and root mat so as to minimize disturbance in the adjacent microhabitat. The excavated root mat and soil must be removed about 10m from the site to minimize disturbance to the trap site. Some soil should be retained for fill around the outside of the trap.
- The pan trap is placed in the depression and the outside surrounding edge filled with soil from the excavation and carefully worked flush with the rim. The fill should provide a surface for microarthropods to walk unimpeded up to the rim of the trap. Fine soil which falls into the trap during placement can be blown out, larger particles must be removed by hand.
- Fill the traps about 3/4 with collecting fluid.
- Place a marker containing the site and trap numbers (in waterproof ink or pencil) near the trap.

Trap Servicing

Traps are serviced by scooping out the contents with an aquarium net and then either reusing the old solution or replacing it with fresh solution. Before scooping, remove large objects (check for arthropods before discarding) such as leaves, twigs, small animals, etc., that may have fallen into the trap (small animals can be retained for the vertebrate portion of the project). The net should be dragged gently near the bottom in one direction several times until the contents are recovered. Check the trap to see if it is still level and top up the collecting solution (if necessary). Ensure that the solution is transparent enough for the yellow background to show through. Specimens are transferred to plastic whirl-pak bags from the net using a squeeze bottle containing collecting fluid to wash out the last specimens. Each trap should be processed separately and a label containing the site and trap identifiers and date of collection placed in the bag. Contents should be double bagged.

Specimen Processing

Specimens collected in the field can remain in the trap solution until they can be processed back at the laboratory. Each pan trap should be emptied into a sieve (or a fine mesh aquarium net) and rinsed thoroughly with tap water. Contents can then be transferred into whirl pak bags, to which 80% ethyl alcohol is added to completely immerse the specimens. Samples should be double bagged. Fresh water washing is essential because the propylene glycol and dirt will form a film on the insects if they are placed directly into alcohol. This film is difficult or impossible to remove once specimens are in alcohol, thus rendering the specimens much less useful. Sample labels (immersible, written in pencil) must be placed inside the bag with the specimens. Change the alcohol in about 7 days, and if possible, refrigerate or freeze the samples to await further processing. Remember that all samples in storage should be double bagged.

Sampling Frequency

In order to fully sample any site or habitat, trapping should be continued throughout the active season, usually April to October in temperate latitudes. Such an extended trapping period is needed to accommodate the different phenologies exhibited by the various taxa. Traps should be serviced every 2 weeks. Refer to the section entitled "Recommended Pilot Studies and longer-term research" for the Pan trap sample program prescription.

Further Reading

Greenslade (1964), Luff (1975), Disney et al. (1982), Snider and Snider (1986), Finch (1991), Topping (1993), Blades and Marshall (1994), Finnamore (1994), Marshall et al. (1994), Masner (1996), Aguiar and Sharkon (1997).

<u>Berlese-Tullgren Extraction</u>

General Extraction Information

Berlese-Tullgren extraction is used to separate arthropods from litter and soil samples. Arthropods can be extracted from almost any moist substrate (decaying wood, fungi, etc.) using a Berlese-Tullgren or similar type of apparatus. Unlike many of the arthropod sampling protocols presented here, the Berlese-Tullgren apparatus extracts arthropods from a standard volume of substrate. The quantitative data (number of specimens) collected are scaleable abundance measures for each species extracted from the sample.

A modified Tullgren apparatus, based on the Berlese funnel (thus often called Berlese-Tullgren funnel), and its various modifications, is the most commonly used method for separating arthropods from soil and litter. The essential component of these extractors is a sample container with wire mesh or screening on the bottom, a metal or plastic funnel in which, or over which, the sample container is placed and a collecting vessel below the funnel which usually contains a liquid preservative, generally 80% ethanol. A source of heat and desiccation (light bulb) is placed above the sample. The objective is to create a steep gradient of temperature and moisture throughout the sample. Arthropods react to the heat and desiccation by moving downward (away from the heat) and eventually fall through the screen at the funnel bottom into the preservative. Cheesecloth below the sample and/or a baffle in the funnel is necessary to reduce debris from the sample falling into the preservative as the sample dries out, or is agitated by the movement of larger organisms. The wattage of light bulb used in the Berlese-Tullgren funnel depends on the size and water content of the sample and on the distance of the bulb from the sample surface.

Commonly used modifications of the Berlese-Tullgren funnel, in which the extraction efficiency is improved by enhancing humidity and temperature gradients, include the Macfadyen high-gradient funnel, the Kempson apparatus and the Merchant-Crossley extractor. For qualitative sampling of large volumes of litter a light-weight, collapsible and easily transportable Berlese-Tullgren funnel is available.

Behavioural extractors extract only the active stages of arthropods. Eggs, prelarva, and quiescent pre-molt stages remain in the sample. In arid solid, deep soils, and for arboreal habitats such as twigs, behavioural extractors are inefficient. Processing using behavioural extractors should be carried out as soon as possible after field collection of samples.

Use

Behavioural extractors collect a diverse sample of soil/litter dwelling invertebrates, primarily Annelida, Arachnida--Araneae, Acariformes, Diplopoda, Diplura, Collembola, Heteroptera, Coleoptera.

Berlese-Tullgren Equipment

The Berlese-Tullgren design, suitable for this project should closely follow the design used by V. M. Behan-Pelletier in a number of soil extraction projects.

- Berlese funnel: diameter at least 30-38 mm.
- Heat source: electric light bulb, 60-80 watts.
- Power source.

Litter Sampling

Litter is the decaying plant and animal material that accumulates on the soil surface. The quantity and quality of litter and the associated arthropod species assemblages vary with the plant community, time of year, precipitation, temperature, and humidity.

- The litter is raked to the level of the soil surface and put through a sifter to remove large fragments of debris and to concentrate the arthropods until 6 litres of sifted litter are obtained.
- The sifted litter is place in a pillowcase, labeled and tied.
- Samples can be kept in the pillowcases for up to 4-5 days as long as they are turned frequently and not exposed to excess moisture or extreme temperatures.

Specimen Processing

Samples are processed by dividing each 6 litre sample into 3 parts of approximately 2 litres each. Each 2 litre sample is place in a separate Berlese-Tullgren funnel and the arthropods extracted according to the instructions given below under Lab Sample Sorting (Litter and Soil). The specimens extracted from each 2 litre portion of a sample (3 funnels) represent a single sample and can be recombined for ease of storage and sample tracking.

Soil Sampling

The sample unit can be a core or cube, ranging from 2.5 cm to 10 cm in diameter, or can be a specific volume of substrate. The most common core size used in recent publications on quantitative sampling for soil arthropods in agro-ecosystems is 5 cm diameter, to a depth of 15 cm in soil. However, the core size used will depend on the habitat, although the size mentioned above is suitable for most temperate forest sampling. There are a variety of corers available, but simple bulb planters constructed from case-hardened steel with 5 cm internal diameter are available and less costly.

- If necessary, gently remove growing vegetation from the sample plot using scissors or a knife.
- Use the standard 5 cm diameter corer to remove soil sample to depth of A horizon (usually 5-20 cm depth). Take 5 replicate cores.
- Divide sample into layers of 5 cm depth and bag (brown wax paper bags or cloth bags) and label separately.
- Keep samples cool until arthropods can be extracted, preferably the same day as collected. If samples cannot be extracted on the same day they should be maintained at 4-10°C. Arthropod extraction techniques follow under Lab Sample Sorting (Litter and Soil).

Lab Sample Sorting (Litter and Soil)

Use Berlese-Tullgren extraction or any extractor modifications by following these protocols:

- Record wet weight of sample.
- Unless sample is very compact, place a layer of cheese-cloth or fine screen over the extractor screen to prevent soil particles being dislodged into the extractant.
- Invert 5 cm soil sub-samples or litter samples over the extractor screen, the top of the sample should be closest to the screen.
- Ensure an air-space between sample and collecting funnel, to avoid condensation in the funnel.
- Extract into 80% ethanol in a collecting jar (or shell vial) placed at the bottom of the funnel.
- Use a dimmer (rheostat) attached to the heating source to establish a gradient between the top and bottom of the sample; this gradient should reach about 30°C by the end of the extraction period. NOTE: dimmers should be temperature calibrated for several days to ensure uniformity between funnels. Alternatively, place extractor in refrigerated cold room (less than 10°C) and a suitable gradient will be established.
- Extract for 4 to 7 days depending on moisture and organic matter content of sample; if in doubt, extract for 7 days.
- Record dry weight of sample.
- Retain sample for determination of soil abiotic characteristics.
- Sample labels must be placed inside the collecting container with the extracted specimens. Change the 80% alcohol in about 7 days and if possible refrigerate or freeze the samples to await further processing.

Sampling Frequency

In order to fully sample any site or habitat, general litter samples and soil cores should be collected every two months throughout the active season, usually April to October in temperate latitudes. Such an extended trapping period is needed to accommodate the different phenologies exhibited by the various taxa. Refer to the section entitled "Recommended Pilot Studies and longer-term research" for the litter/soil core sample program prescription.

Further Reading

Macfadyen (1962), Kempson et al. (1963), Merchant and Crossley (1970), Martin (1977), Krantz (1978), Seastedt and Crossley (1978), Norton (1986), Steyskal et al. (1986), Walter et al. (1987), Wheeler and McHugh (1987), Norton and Kethley (1988), Crossley and Blair (1991), Edwards (1991), Kethley, (1991), Marshall et al. (1994), Anderson (1996), Behan-Pelletier et al. (1996).

Lindgren Traps (baited)

General Trap Information

Lindgren traps are a series of black funnels (8 or 12 funnels are available) that are connected one on top of the next. They function in a manner similar to flight interception traps by collecting flying arthropods that hit the funnels and then drop into the collecting container attached to the bottom of the funnels. Lindgren traps primarily catch flying Coleoptera and take advantage of the fact that when most Coleoptera hit an obstruction they fold their wings and drop. Lindgren traps come pre-assembled, are easy to install and samples are less expensive to process because the trap contents are free of debris and are dominated by the order Coleoptera. Unmodified Lindgren traps remain neutral for active, dispersing beetles.

Lindgren traps are normally provided with a ‘dry collection’ container. A ‘wet option’ is available upon request (S. Lindgren, pers. comm.). The most common modification to Lindgren traps is to use a bait, which is usually a pheromone that is attached in a pre-sealed rubber lure. A variety of lures are available, however a simple ethanol lure attracts the greatest diversity of Coleoptera. As a result, ethanol baited Lindgren traps can be used to assess the diversity (richness and evenness) of Coleoptera that are associated with forest habitats.

Use

Baited Lindgren traps collect mainly flying Coleoptera but also collect Diptera and provide incidental catches of Hymenoptera.

Lindgren Trap Equipment

The Lindgren trap protocol suitable for this project should closely follow that used by Humble and Winchester in their long-term Coleoptera monitoring project at the Montane Alternative Silvicultural System site on Vancouver Island.

- Lindgren trap (8 funnel model) and ethanol lures.
- ‘Wet’ collection container.
- Cord for trap suspension, approximately 2 m per trap.
- Short ladder for trap installation.

Field Installation Protocol

Lindgren traps should be hung about 1.5 m from ground level. If abundance measures are desired choose consistent microhabitats in which to place the traps (e.g., standardize edge effect).

- Suspend the Lindgren trap between two vertical objects (usually trees) by attaching the trap to a cord that spans the gap between the two vertical objects.
- Adjust the top wire of the trap to center the trap so that it remains in a stable upright position.
- Adjust the tension on the cord suspending the trap so that the trap is positioned about 1.5 m from ground level.

- Fill the collecting head with collecting fluid to about 1/4 full.
- Place an identification marker containing the site and trap number (in waterproof ink or pencil) and attach it to the trap.

Trap Servicing

Traps are serviced by emptying the entire contents of the collecting head (specimens and collecting fluid) into a whirl-pak bag. If needed, use a squirt bottle to wash any specimens on the sides of the collecting head into the whirl-pak bag. Refill the trap head and attach it to the trap. Each trap should be processed separately and a label containing the site, trap identifiers, and date of collection placed in the bag. If needed add collecting fluid to the bag to cover the specimens and double bag the sample.

Specimen Processing

Specimens collected in the field can remain in the trap solution until they can be processed back at the laboratory. Each Lindgren trap should be emptied into a sieve (or a fine mesh aquarium net) and rinsed thoroughly with tap water. Contents can then be transferred into whirl pak bags, to which 80% ethyl alcohol is added to completely immerse the specimens. Samples should be double bagged. Fresh water washing is essential because the propylene glycol and dirt will form a film on the insects if they are placed directly into alcohol. This film is difficult, or impossible to remove once specimens are in alcohol, thus rendering the specimens much less useful. Sample labels (immersible, written in pencil) must be placed inside the bag with the specimens. Change the alcohol in about 7 days, and if possible refrigerate or freeze the samples to await further processing. Remember that all samples to be stored must by double bagged.

Sampling Frequency

In order to fully sample any site or habitat, trapping should be continued throughout the active season, usually April to October in temperate latitudes. Such an extended trapping period is needed to accommodate the different phenologies exhibited by the various taxa. Traps should be serviced every two weeks. Refer to the section entitled "Recommended Pilot Studies and longer-term research" for the Lindgren trap sample program prescription.

Field Resource Costs: Equipment and Installation

An appropriate sample design must be implemented in order to gather meaningful data that can be used to monitor change and to provide flexibility in addressing biodiversity questions. Quantitative sampling requires a well-defined objective and utilizes the most appropriate collection technique for the habitat of choice (see Scudder 1996a). Additionally, sampling must take into consideration site variability, sampling frequency, and adequate replication (e.g., Marshall et al. 1994, Danks 1996, Finnamore et al. 1998).

In order to obtain reliable data for a monitoring project using the protocols listed, it will usually be necessary to replicate some of the sampling activity. This replication will compound the sorting, preparation and identification logistics, but if undertaken will provide greater reliability and resolution to detect change. Sampling replication (spatial and temporal) is directly related to arthropod activity (Southwood 1978). Because the phenologies of species are variable, even within a single taxon, sampling must be carried out for an extended period, usually over the whole of the active season. Compelling reasons for sampling over the entire active season (see, Kremen et al. 1993; Wiggins et al. 1991) include finer resolution for monitoring alpha and beta diversity, capturing the full complement of size classes (i.e., micro-, meso-, and macro-fauna) that exhibit a range of ecosystem requirements (highly specific to generalist) and a wide range of dispersal abilities, and coverage of different life cycles and development times (difficult to predict on single sample entries). Activity is directly related to climatic conditions, and geographic location. These must be factored into the monitoring program. Seasonal variation requires sampling over the entire period of activity and must take life cycle duration into account. The time will vary depending on latitude, longitude and climatic factors, and in Alberta these vary significantly across the province. However, sampling in Alberta should generally run from May to October inclusive.

Efforts should be directed at random sampling (e.g., trap placement) as this is an important prerequisite for robust statistical analyses. If done properly, meaningful comparisons can be made with other independent monitoring sites over extended time periods.

The key to an effective field sampling plan is to ensure that the required funds and personnel will be available over a time frame that is of adequate duration (Danks 1996). For any single site, estimated costs for trapping equipment, general field equipment, installation of a complete set of sampling methods, minimum costs for a single interval trap collection and for a complete field season are presented in Tables 14.1-14.3.

General equipment costs that allow for the installment of a replicated set of traps with servicing over a complete sampling season at one site are estimated to be $300 (Table 14.2). Restocking of certain equipment items will add to overall cost both in terms of number of sites chosen and the frequency with which traps are emptied. Restocking costs per site over a May to October field season are likely to be equivalent to the initial start up cost of $300. Therefore, maximum total general field equipment costs are estimated to be $600 per site. It should be noted that this cost can be reduced with sharing of general equipment and buying items such as liquid preservative in bulk.

Estimates of minimum equipment costs for individual traps and minimum replication per site are presented in Table 14.3. Total costs per site are $2,016.

Estimates of minimum installation of a complete set of sampling methods for one site are presented in Table 14.4. Based on a research crew of 2 trained people, working for $15 per hour, it is estimated that installation costs for all traps will be $180 (2*6*15). Costs to service the traps during a single sample collection period are estimated to be $90 (2*3*15). Costs to service the traps during 12 sample collection periods (bi-monthly for 6 months) are estimated to be $1080 per site (2*3*12*15). Total costs to complete sampling in a single site (all traps, serviced 12 times), excluding transportation to and from the site is estimated to be $1,260 ($180 + $1080).

The total cost for the equipment (Table 14.3), installation and servicing (Table 14.4) for a single site is estimated to be $3,276 and represents the costs for monitoring just the arthropod component of the project. Note that these costs will change with different combinations of sampling frequency, trap type and trap replicates used. Calculations for any combination of these variables can be obtained by using the information presented in Tables 15.3 and 15.4.

Sorting and Sample Preparation

Introduction

The recommendations for sampling presented in this report will generate a large number of samples and specimens. It is imperative that thought be given to archiving this information. If this component of the project is done properly it will enable future questions to be explored. Choice of target groups may change in the future as information, interests and management questions continue to evolve. This should not be an impediment to a broad-based arthropod sampling program, in fact it is a benefit because broad-based trapping programs, if done properly, will ensure a multitude of options remain open for selection of target groups to monitor change over time and space.

The most time-consuming and costly aspect of any arthropod biological inventory or monitoring study is the conversion of trap samples into prepared, labeled and sorted lots to be identified (Marshall et al. 1994). As with sampling itself, strict control and standardization of field protocols by technicians is required to ensure data integrity and long-term maintenance of specimens (Danks 1996). Differences among technicians create additional problems in sub-sampling or other quantitative work (pers. obs.). Constant care and attention is required even during prolonged and relatively mundane activity (e.g., specimen labeling).

It is particularly important to plan sorting and preparation with identification in mind. Specimens of many groups are much more difficult or impossible to identify if carelessly or incorrectly prepared. Detailed instructions about preparation for identification must therefore be obtained from cooperating systematists or trained principal investigators as the project is developed.

A well designed monitoring project is likely to result in the collection of several thousand specimens, some of which will become valuable identified voucher specimens, and provide a solid foundation to monitor change in selected taxonomic groups gathered from forest habitats. It is important to consider the long-term integrity of this material (Marshall et al. 1994) and it cannot be overemphasized how important this material will be as new questions and analytical tools become available to address future questions. Unidentified material or unsorted trap catches should be properly archived for later incorporation into, or comparison with, the long-term goals of the monitoring project.

Long-term terrestrial arthropod projects need to establish appropriate databases to allow for maintenance and exchange of data files on specimens and habitats (Marshal et al. 1994, Danks 1996). Development of computer-based data management systems has created an extensive amount of literature concerned with tracking information on specimens collected during structured biodiversity studies. Aspects of standard reporting and tracking of arthropod specimens have been proposed by Noonan (1990) and Noonan and Thayer (1990). Monitoring programs for arthropods are feasible, given proper planning. Well-defined protocols, and the need to keep basic data associated with collected specimens are at the core of data management and. The following sections describe the components necessary to finish the 'back end' of a successful terrestrial forest arthropod monitoring project.

General Sample Processing

Specimens from the field should be stored in a cool dark place (refrigerate if possible), but not in direct sunlight or near a heat source. Replace alcohol with fresh 80% alcohol after 1 to 2 weeks. The sooner the catch is processed the better in terms of quality specimens and reduced sample backlog. For long-term storage (several months to years) it is essential to replace alcohol (to prevent ice crystal formation) and to keep the catch below 0°C, preferably in a deep freeze at -10°C or lower to prevent specimen deterioration. Archive samples in labeled 'Rubbermaid' containers to keep samples together, reduce evaporation, and increase ease of storage and efficient retrieval.

Sample processing consists of sorting, mounting, and labeling specimens in preparation for identification. Sample processing should be done in an area that is equipped with the basic laboratory essentials (e.g., water, bench space etc.). A typical sample sorting station(s) should be equipped with the following: stereo-microscopes and cold light sources, petri dishes, vials and rubber stoppers, 95-100% denatured ethyl alcohol (to be diluted to 75-80%), 750 ml squeeze bottles for alcohol and water, sieve series (coarse, medium and fine), fine point forceps, insect pins, insect points, white glue, paper for temporary labels, archival quality label paper (acid free, neutral pH card stock) for permanent labels, microscope slides, cover slips, mounting medium, glyptol, slide warmers and slide labels, soft lead pencils, fine and coarse scissors, counters, clear 'scotch' tape, and paper towels.

A typical sorting procedure for any of the trap samples that were collected into fluid is:

- Remove the sample from the specimen container (e.g., whirl-pak, Malaise collecting head). Gently rinse tap water into the specimen container and ensure that the contents are flushed into a series of sieves.
- Continue washing the sample (i.e., to rinse off the collecting fluid and any debris) so that the contents are separated into the 3 sieve fractions (coarse, medium and fine).
- Place the three sieve fractions into separate glass, high-sided petri dishes and cover the contents with 80% denatured ethyl alcohol.
- The fractions can now be sorted to target taxa. Generally, it is preferable for the first sort to be an ordinal level sort, where all specimens are sorted and assigned to orders. Once the majority of specimens have been sorted the remaining material (i.e., fluid, debris, small arthropods) should be viewed under the stereo-microscope to ensure that all specimens have been removed from the fraction.
- When sorting it is desirable to sort whole specimens, however there will be specimens that are damaged. As a general rule, only collect damaged specimens that still have a head (e.g., in some cases the head may be all that remains).
- It is important to record the sort information on a data sheet (hard copy). A typical data sheet contains the following information: data label, date sorted, name of sorter, time taken to sort, column of target taxa (i.e., first sort = list of orders), columns for numbers of individuals sorted and assigned to each order, number of vials containing the specimens (this is particularly important when more than one vial is used). At the bottom of the data sheet, there should be a space to write in totals for order, individuals and vials. An example of a typical data sheet used in the forest biodiversity surveys conducted by N. Winchester, can be obtained from the author.
- Ordinal sorts from the 3 fractions (i.e., a complete sample) can be combined, and stored in glass vials. Immerse the specimens in 80% ethyl alcohol, (for temporary storage, until further sorting, the alcohol should not reach the level of the rubber stopper), and add an immersible label (that matches the data sort sheet). Seal the vial with a rubber stopper and properly store the vials.

Sample Sorting Costs

Removal of specimens from bulk samples collected from a series of different traps is time consuming and expensive. Estimates of time vary depending on such factors as trap type, locality, seasonality, and care in collecting samples. Estimates of the time required for sorting specimens from given traps samples is difficult to arrive at, however Marshall et al. (1994) give estimates of the minimum time required to process selected samples from one site using their recommended sampling methods and protocols. Their minimum estimates apply only to experienced sorters, sorting specimens from a small number of selected families. For any single sample site, total time to process all trap material (collected from 8 Malaise, 8 flight interception, 16 pan, 16 pitfall and 10 extractors) was 504 hours. At a pay rate of $10.00/hr, the total cost was $5,040. Thus, for any single-site program carried out in most areas of Canada, over a seven-month period from April to October (active period) would cost $35,280. These processing costs do not reflect costs involved with species identifications.

Based on the forest biodiversity surveys conducted on Vancouver Island, British Columbia (N. Winchester, unpublished data) the costs associated with sorting samples are presented in Tables 15.5-

15.9. Factors that influence sort time are trap type, number of specimens, amount of debris in samples, and time of collection. When using the tables, the following points should be noted:

- Estimates are means and standard deviations for 30 samples collected from a single forest site on Vancouver Island.
- Samples spanned the entire sample season from April to October, inclusive.
- Sorting was done by a trained para-taxonomist with several years of experience.
- All data presented are for ordinal sorts, except for the Malaise trap where data are given to examine the costs involved with sorting target families from one order.
- Labor costs for extraction of soil/litter samples are minor and are not included in this section.

For any single site the average sort cost using the recommended sampling protocols contained in this report are presented in Tables 15.5-15.9. Costs range from $8,712 for 22 Pan traps from one sample site serviced 12 times, to $388 for 12 Lindgren traps from one sample site serviced 12 times. These tables can be used to calculate individual trap costs (any combination of number of traps) for any servicing period over the active season (1-12). The total cost for any single site using the full complement of traps sampled over the entire sample season is $15,022.

Specimen Mounting and Identification

Mounting of specimens is important, and, if properly done, assists greatly in identification of specimens. Securing reliable identifications to species is the greatest single difficulty in work on large insect projects. Except in the few best know groups, expert knowledge is required to ensure that identifications are accurate, and such expertise has to be planned for from the earliest stages, because it is both extremely limited and in great demand for these and many other activities (Danks 1996). Because a good systematist is a scientist, not a technician, specialists are best able to help if they receive quality specimens that have been prepared correctly for identification. Different arthropod groups require different mounting techniques and it is recommended that this aspect of the project should be coordinated by a scientist that has the knowledge, experience and contacts with taxonomists in different groups. For example, the most logical choice for a project in Alberta would be to involve Dr. A.T. Finnamore and the Provincial Museum of Alberta to take a lead in the tasks involved with specimen mounting, labeling, identification, storage and archiving of the large arthropod specimen collection that will be generated if the protocols for sampling terrestrial arthropods are implemented.

Different arthropod groups require different mounting techniques. Large insects can be pinned, most smaller insects can be pointed, but some are left in alcohol vials, while soil microarthropods such as mites are slide mounted. Martin (1977) and Borrer et al. (1992) should be consulted for basic techniques in specimen mounting, however several modifications to these techniques exist and specific directions need to given by an experienced entomologist.

Using selected guidelines presented by Danks (1996) under requirements to optimize identification by specialists, the information presented in the following tables will give an idea of the costs involved in specimen preparation for identification to species.

1. Sort all material from substrates (traps). Refer to Sample Sorting Costs, Tables 15.5-15.9.
2. Organize material into higher taxa. Refer to Sample Sorting Costs, Table 14.4. Target groups 17 families from the Diptera. Generally sorting to family is level required to then proceed with the next steps in specimen preparation for species identification.
3. Prepare (preserve, mount, etc.) appropriately. Based on data collected in an arthropod biodiversity project on Vancouver Island, the total costs (equipment, specimen preparation) for one family of beetles (Staphylinidae) are presented in Table 14.8. Total costs for processing 3,000 specimens from one target family of beetles was $1,653.53.

4. If necessary, pack and ship material using proper methods to safeguard it. These costs vary and depend on shipping institution (e.g., University, Museum) and mode of shipping (e.g., air freight, courier, final destination). Due to the variable nature of these costs, a cost estimate for shipping is not included in this report.
5. Specimen storage and costs are difficult to estimate and depend on such factors as place of storage and equipment needed (e.g., cabinets, specimen drawers).

Special Considerations

Marshall et al. (1994) clearly pointed out the need for a long-term plan to organize information collected from a well-designed arthropod project that is likely to result in the collection of tens of thousands of specimens. "Some specimens will become valuable identified voucher specimens, and some specimens will be unidentifiable at the present time, yet worthy of retention". It is important to consider the long-term integrity of the material. Voucher specimens and samples residues are best placed in the care of museums with legislated mandates for long-term care and maintenance of natural history specimens.

To organize the information from the arthropod monitoring program it is suggested that the following points be considered to facilitate an efficient retrievable baseline that best meets the needs outlined by Shank and Farr (in prep.). Cost considerations will be variable depending on the infrastructure and agreements that are set up to handle specimens. Therefore it is beyond the scope of this report to deal with these costs, however the following considerations are relevant:

- The Provincial Museum of Alberta should be explicitly mandated to coordinate specimen processing, identification, long-term curation, and management of the associated data, as well as archiving of the sample residues. This step will require a great deal of thought and planning to address the resources needed to ensure success (e.g., physical space, equipment, and personnel).
- Securing reliable identifications to species is an essential component to monitoring terrestrial arthropods. Costs vary and depend on a number of factors (e.g., collaboration agreements, contracts). Since this is a real cost to the overall project details should be worked out in advance. The responsibility for this important task will rest with the Alberta Provincial Museum in conjunction with consultation from taxonomic/biodiversity specialists.
- Data management is an important component of biodiversity monitoring. Data, like specimens, must be curated and archived. Standards must be established for data entry and data must be curated to ensure its long-term integrity and accessibility (e.g., this step should allow for monitoring past and present effects of habitat change on species in the mixed boreal forests under study. For additional discussion see, Mickevich (1999), Oliver (1999).

Recommended Pilot Studies and Longer-Term Research

Introduction

Recent work on forest insects indicates that multiple species, many of which are new to science, exist in habitats associated with forests and that they play an essential role in ecosystem functions (see Watt et al. 1997). The arthropod communities associated with Alberta's forested natural regions have not been well documented, although extensive work on some groups in specified regions has been completed (see Hammond 1992, Spence et al. 1997). Conservation of biological diversity is a major environmental issue and the development of a comprehensive program for monitoring forest biodiversity in Alberta has a high priority in terms of silvicultural practices, land-use planning and biodiversity research (Shank and Farr, in prep.). The reasons for maintaining biodiversity have been clearly identified and results from intensive arthropod studies in British Columbia support the generalization that northern temperate forests across Canada contain a unique set of species that have evolved to form forest communities that are specific to microhabitats within the ecosystem. Since natural resource management has unknown consequences on the arthropod component in the forests of Alberta, information is needed to monitor the effects of harvesting (traditional and alternative) and silvicultural systems being field tested in order to ensure optimal management of the resource.

This short-term study focuses on documenting of costs and identification of efficiencies associated with monitoring arthropod communities. Concerns over number of sites and ecozones sampled will have to include consideration of statistical power and cost projections. Arthropod sampling could be conducted in a maximum of 3 randomly chosen sites in each of 6 ecozones (i.e., 18 sample sites). If sites need to be reduced then it is suggested that a minimum of 3 replicates be retained and the number of ecozones be reduced to a minimum of 3 (i.e., 9 sample sites). Each site will be sampled using the arthropod sampling protocols recommended in this report (see section entitled "Data analyses"). To sample arthropods, Malaise flight-intercept traps, pan traps, pitfall traps, Lindgren traps, and soil/litter extractions will be used to collect arthropods. If further reductions in costs are needed then two logical possibilities exist: (1) components of the trapping program need to be deleted, (2) frequency of trap cleaning needs to be reduced. It should, however, be noted that any reductions in number of sites and/or reduction in sample frequency will have an impact on the quality of data in terms of level of sensitivity to detect changes due to finer-scale disturbances.

Intensive sampling using active and passive techniques (e.g., baited Lindgren trap, extractors, flight interception traps) will be conducted in replicate sites to assess the overall diversity of arthropods from which selected arthropod taxa (e.g., Coleoptera) can be used for finer grained analyses (e.g., refer to data analysis in section entitled "Integration and data analyses"). To ensure that the proposed pilot project sampling program provides a representative sample of the target taxa potentially present in the forest sites, this intensive sampling should be conducted in part, under the direction of specialists in all the details needed to ensure a successful program (e.g., field sampling, final specimen preparation, identification, deposition of material). Sampling should be carried out bimonthly throughout the active season (May until October, inclusive). If sample frequency needs to be reduced then it is suggested that a minimum of 3 intervals be retained (early, mid, late) and the number of trap collections be reduced from 12 to 3.

Sorting, mounting and classification of forest arthropods from the trap systems need to be conducted at an adequate facility and in consultation with entomologists. Selection of the initial target taxa will be based on advice forwarded by entomologists from the technical advisory group. For example, research on the Staphylinidae (rove beetles) in temperate forest (e.g., Spence et al. 1997, Winchester 1997b, Hammond et al. 1997) indicates that this group is a target group for a monitoring program. Staphylinids have many of the characteristics of valuable bioindicators for conservation management, rehabilitation and monitoring (Noss 1990, Kreman et al. 1993). Higher taxa are broadly distributed geographically and have specific habitat requirements. Populations are relatively stable under natural environmental conditions. Diversity is high, they represent a trophically heterogeneous group, and most species tend to be numerous. Staphylinids are easily sampled, they can be sampled throughout the entire sample season, morphospecies are comparatively easily recognized, and specialists for identification are available. They are one of the most easily assessed beetle groups, and as such can serve as a proxy for other ground-dwelling forest arthropods.

Field Methodology

NOTE: As previously noted in this chapter the final decision on type of traps, replicates, site selection and placement etc. will be decided by the AFBMP and will include factors such as costs, logistics and accessibility.

1. Selection of field study sites. A minimum of 3 sites per ecozone. Within each site traps are randomly placed (i.e., randomized plot design).
2. The sample design and trap type used should follow the recommendations outlined in the section entitled "Monitoring protocols and logistics".
3. Table 14.10 gives the cost breakdown of the optimal pilot study design. When viewing the table, note the following:

- Design: 18 sites: 3 sites per ecozone (6).
- Design is based on minimum replication needed for adequate analysis for all ecozones using the data analyses package recommended in the section entitled "Selection of taxa for monitoring" of this report.
- Trap costs are based on costs generated for each trap type based on tables contained in the section entitled "Monitoring protocols and logistics" of this report.
- Specimen sorting means that all samples are processed, and sorted to order. From these samples costs are given to pull out one group (Staphylinidae), and properly mount and label specimens in that family in preparation for shipment to a systematic authority for species determination.
- Costs do not include: lab space, storage and general lab supplies. In addition, identification to species and archiving sorted samples and residues are not included.
- Total cost $399,463.00

Table 14.11 gives the cost breakdown for the first example of a variant on the original sample program. When viewing the table, note the following:

- Design: 9 sites: 3 sites per ecozone (3)
- Design change is a reduction of ecozones, from 6 to 3.
- Design change is a deletion of the light trap program.
- The remaining points listed under Table 14.2 apply to this table.
- Total cost $181,961.00

Table 14.12 gives the cost breakdown for the second example of a reduced sample program.

- Design: 9 sites: 3 sites per ecozone (3) (as per Table 14.2).
- Design change is a deletion of the light trap program and a reduction of the pan trap program. All pan traps (12 per site) associated with the Malaise traps have been deleted.
- The remaining points listed under Table 14.2 apply to this table.
- Total cost $129,055.00

Table 14.13 gives the cost breakdown for the third example of a reduced sample program.

- Design: 9 sites: 3 sites per ecozone (3) (as per Tables 15.2 and 15.3).
- Design change as per Table 14.3 and sampling frequency (i.e., trap servicing) has been reduced from 12 to 3 sample times.
- The remaining points listed under Table 14.2 apply to this table.
- Total cost $46,191.00

Other scenarios exist to change the total costs, however it should be stressed that the minimum design considerations must take into account the tradeoff between trap replication, and sample site replication. Sample frequency and subsequent processing of samples is where the bulk of the costs lie, therefore it is suggested that any changes in the suggested pilot program should revolve around sample frequency (maximum of 12 bi-monthly trap servicing; minimum of 3 seasonal trap servicing (early, mid and late. Traps should still be set to gather a 2-week catch for each of the 3 sample intervals).

Future Research

Preface

This section on suggested areas for future research is a module that provides a standardized format to sample canopy arthropods in northern temperate forests. Although these protocols were developed in coniferous forests, the methodologies can be used in other Canadian forests (e.g., Aspen forests). In this module, the term 'canopy' is used in its broader sense, and means the crown of trees. This synopsis of canopy arthropods addresses appropriate sampling methodologies and survey design considerations for two sampling techniques, Lindgren traps and Branch clippings.

Users of these protocols are encouraged to consult the following reference material:

1. Stork, N. E., J. Adis, and R.K. Didham (eds.) 1997. Canopy Arthropods. London: Chapman and Hall. ISBN 0 412 74900 9.
2. Marshall, S. A., R. S. Anderson, R. E., Roughley, V. M. Behan-Pelletier, and H. V. Danks. 1994. Terrestrial arthropod biodiversity: planning a study and recommended sampling techniques. A brief prepared by the Biological Survey of Canada (Terrestrial Arthropods).
3. Winchester, N. N. and G. G. E. Scudder 1993. Methodology for sampling terrestrial arthropods in British Columbia. A brief prepared for the Resources Inventory Committee (RIC), Ministry Environment, Lands and Parks, British Columbia.
4. International Canopy Network (ICAN). Newsletter and server. Contact--email address: canopy@elwha.evergreen.edu. Homepage: http://esnet.edu/ican
5. University of Victoria, Biology Department, Canopy web page. Address: http://web.uvic.ca/~canopy/

Introduction

Canopy Arthropods

The study of forest canopy arthropod assemblages and the systematics of canopy arthropods has developed rapidly during the last 20 years (Stork and Best 1994; Stork et al. 1997). In general, canopies of rainforests contain a large percentage of the species present in these forest systems (see Stork et al. 1997). Canopies of natural forests in temperate (Schowalter 1989; Winchester and Ring 1996a,b; Behan-Pelletier and Winchester 1998; Winchester 1997a,b) and tropical (Erwin and Scott 1980; Erwin 1983; Stork 1988; Basset and Kitching 1991; Basset 1994, 1997, Didham 1997, Davies et al. 1997, Hammond et al. 1997, Kitching et al. 1997) regions contain largely undescribed and little understood assemblages of arthropods that have greatly expanded estimates of the total number of arthropod species on Earth. Estimates of total species numbers range as high as 100 million species, concordant with the assumption that tropical forest canopies provide the habitat template for this incredible diversity (Wilson 1988; Erwin 1988, 1991; Stork 1988, 1993; Gaston 1991; Kitching et al. 1997; Stork et al. 1997).

Structured Inventory

The study of canopy arthropods involves the recording and collecting of ecological groups, in a series of stations throughout a defined area. Improved canopy access techniques and the development of new sampling methods have enabled researchers to incorporate taxonomic inventories into broader-based ecological programs. The Biological Survey of Canada has approved the principle that meaningful inventories require detailed knowledge at the species level and therefore protocols should ensure: (1) identification of arthropod groups that characterize the diversity associated with aerial habitats, at the macro- and microscale levels; (2) avoidance of single-species approaches, and initial concentration on arthropod groups that trained systematists can identify (e.g., Arachnida, Coleoptera). The absence of an adequate systematic framework makes this step of crucial importance; and (3) all representative subhabitats (e.g., suspended soils, epiphytes) are sampled using a quantitative approach. Quantitative sampling requires a well-defined objective(s) that includes the most appropriate collection technique for the habitat of choice.

With limited resources available for a pilot study a full arboreal sample program is not feasible. Therefore it is suggested that the following sample programs could be implemented to supplement and significantly enhance the arthropod elements and ground-based monitoring protocols.

Aerial Beetle Traps (Lindgren Funnel Traps)

Lindgren funnel traps are designed to catch a wide spectrum of beetle taxa. These traps can be baited with a pheromone/host volatile, although to maximize canopy species richness, alcohol lures should be used. Traps are placed in the canopy by completing the following steps:

- Select a canopy gap (area between adjacent trees where there is a clear and open space between the canopy and ground. Lindgren traps can also be attached onto tree branches, using the same pulley system and ensuring that there is a clear drop to the ground.
- Ascend (using single rope techniques) a single tree.
- Install a line (using shock cord) across the canopy gap. The line fixed at a set height in the first tree now spans across to the second tree where the line runs to the ground.
- Install a simple closed pulley that is fixed onto the line that is between the two trees. The distance between the two trees that the pulley is fixed at, can be adjusted by shortening or lengthening the line that runs to the ground from the second tree.
- Install a trap line (shock cord) that runs from the ground through the pulley and back to the ground. This line is situated in the gap (generally the middle) and must be placed so that the trap can be raised and lowered without getting caught in tree branches.
- Attach the Lindgren funnel trap to one end of the line and pull the trap into the canopy gap, to the height of the pulley. The distance of the trap from the ground to the canopy is set by the researchers and can be adjusted by changing the pulley height.
- If metal spacers are used then 2-3 Lindgren traps can be fixed onto one line.
- Trap solution is a mixture of Propylene Glycol and water.
- Traps can be serviced at two-week intervals, although shorter intervals can be used.
- Cleaning of traps does not involve access into the canopy. The trap can be raised, lowered and cleaned from the ground.

Lindgren Trap Samples

Aerial Lindgren trap samples are processed using the same procedures as those outlined for ground Lindgrens. All costs associated with this program are identical to the ground Lindgren trap program, except for the initial installation which requires the services of two qualified climbers

Branch Clipping

Branch clipping involves enclosing a branch in a large plastic bag and removing the branch from the tree (Ohmart et al. 1983; Majer and Recher 1988; Schowalter 1989; Costa and Crossley 1991; Basset et al. 1992; Winchester 1997a, 1998). The number of samples to be collected varies with tree species and sample design. Individual bags may need to be pooled to be sufficiently representative (Blanton 1990). For example, Winchester (1997a) sampled each of five Sitka spruce trees by collecting bagged branches where three samples were taken at each of three heights (33 m, 45 m, and 54 m) over 6 sample times.

Branches are collected in the canopy by completing the following steps:

- Ascend (using single rope techniques) a single tree.
- Select single branches from each height to be sampled. Replicate branches for any combination of tree and height should be taken (e.g., a minimum of 3 branches).
- Using a heavy weight (e.g., 70 lbs.) polyethylene bag, put the bag around the branch to be clipped. Note that this means that the end of the branch is often the sample being collected. Branches could be sectioned and bagged all the way to the trunk, although on large trees this may not be feasible. The branch being clipped should not be disturbed before the bag is in place, since this will result in a loss of mobile arthropods. Once bagged, the branch can then be clipped (e.g., usually a small saw is used to cut the branch).
- Collected branches can then be lowered to the ground or brought down with the climber/researcher.

Additional notes:

- A collapsible-bag sampler can be used (see Blanton, 1990).
- Macro-arthropods can be anaesthetized and immediately collected from the bag by dropping in a small ball of cotton-wool, saturated with ethyl acetate.
- Branch clipping northern temperate coniferous forests can be done at three time intervals: early (vegetative bud growth/burst), mid (full cone development) and late (end of growing season). Sample times will vary with geographic location and research objectives.

Branch Clipping Samples

- At all times collected branches should be kept out of direct sunlight and as soon as possible samples should be stored in a walk-in cooler at 5°C. Samples can remain in cold storage for a maximum of 1 week after which the quality of the samples significantly declines.
- Depending on the design, basic measurements of the branch can be taken. At the very least, fresh weight, dry weight, maximum length, width and age of the branch should be recorded. For example, pre-calculated regressions of dry weight against leaf area can be used to evaluate effective sample size (Basset et al. 1997).
- To remove arthropods from the branch, remove the branch from the bag and place it on a white sheet in a container with sufficient height to contain the branch. Mobile arthropods can be collected using a wet Q-tip or an aspirator. Large arthropods can be manually collected. Extreme care must be taken to collect all mobile arthropods, especially fast moving individuals (e.g., most arachnids) and those individuals capable of flight (e.g., parasitic wasps). The sample bag must be examined for arthropods. Once the desired branch measurements have been recorded the branch can be sectioned into smaller pieces and each piece should be examined for the presence of arthropods. The sorted branch pieces should be returned to the original collecting bag, which is then sealed with a twist tie and this bag is then suspended below a light source (brightness, not heat is required). Contents of the bag should be checked periodically over the next 48 hrs for any arthropods that were missed during the original sorting, most mobile arthropods will be at the top of the bag.
- Macro-arthropods can be removed and immediately mounted.
- Arthropods from the branch samples may be obtained by rearing larvae (e.g., Lepidoptera), pupae (e.g., Lepidoptera, Diptera) and parasitoids (e.g., Hymenoptera). Rearing of parasitoids is particularly useful since adults are often the only stage that can be identified to species and valuable information associated with hosts can be obtained.
- Branches must now be processed for microarthropods (mainly Acari). All branch parts should be placed in a 5-gallon bucket and submerged in water, to which 10 NaOH tablets are added. This 'mixture' should be stirred occasionally and left for 48 hours. After 48 hrs, branches should be removed from the solution and rinsed with tap water over a sieve (75 micrometer). Contents of the

bucket should be poured into the same sieve and all material should be transferred to a whirl-pak bag, labeled and preserved in 75% alcohol. Samples can now be stored in the freezer until they are ready for sorting. The clean branches should now be oven-dried for 72 hours (or until completely dry) and total dry weight recorded.

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Table 14.1 Abiotic attributes that can be positively associated with insect diversity and distribution.

Spatial/temporal Referencing	Global positioning reference (preferably differentially corrected) of latitude and longitude, expressed as decimal degrees, and altitude
	Slope, aspect
	Height of trap above surface or depth extracted below surface
	Date of sample collection
Climate Monitoring (ITEX protocols)	Daily, preferably hourly, maximum, minimum temperature
	Daily soil temperature (max. - min.) at surface, 5 cm and 10 cm depths
	Millimeters of precipitation
	Depth of snow
	Date of nearest lake ice breakup
	Date of nearest lake ice formation
Soil Parameters	Soil organic carbon
	Bulk density
	pH
	Soil structure
	Rooting depth
	Soil carbon levels
	Carbon biomass
Soil Fauna Distribution Studies	Soil moisture
	Particle size distribution
	Electrical conductivity
	Soil temperature (max., min.) at surface, 5 cm and 10 cm depths
	Exchangeable Ca status
Associated Biotic Protocols	For example: Complete Vegetation plots
	Refer to: //www.cciw.ca/eman/ 1. Canadian Biodiversity Monitoring: Terrestrial forest plots and links with UNESCO/SI forest monitoring plots. 2. Complete set of bioclimate monitoring and configuration (EMAN)
	Co-dominant species
	Shrub/forb density, distribution, and identity
	Amount, size and decomposition state of coarse woody debris
	Characterization of stand structure

Table 14.2. List of general field equipment required to set up and service all traps in a single sample site. Equipment shared between traps is represented by = sign. The preservation of traps is 50:50 Propylene glycol (Glycol-P) with distilled water mixed with a teaspoon of Bitrix (20 L per site, for initial installation and for each subsequent trap cleaning). Maximum cost for equipment required for each field installation is $300.00.

Field Installation						
Equipment	**Malasie (3)**	**Pitfall (10)**	**Pan (10)**	**Light (5)**	**Soil/Litter**	**Lindgren (12)**
Cord*	90 m	N/A	N/A	5 m	N/A	24 m
Wooden poles*	6	N/A	N/A	N/A	N/A	N/A
Stakes*	42	N/A	N/A	N/A	N/A	N/A
Hammer	1	=	=	=	=	=
Trowel	2	=	=	=	=	=
Shovel	2	=	=	N/A	=	=
Axe	2	=	=	N/A	=	=
Knife	1	=	=	=	=	
Garden hand rake	N/A	N/A	N/A	N/A	2	N/A
Garden bulb corer	N/A	N/A	N/A	N/A	2	N/A
Carboy (water)+*	1	1	=	N/A	N/A	N/A
Carboy (pres.)+*	N/A	1	=	N/A	N/A	=
Spare 'bucket'	2	=	=	N/A	N/A	N/A
Markers*	3	10	10	5	N/A	12
Aquarium net	N/A	2	=	N/A	N/A	=
Whirl pak bags +*	24 (pans)	20	20	N/A	N/A	24
Squeeze bottle	3	=	=	N/A	N/A	=
Soft lead pencil	4	=	=	=	=	=
Label paper+*	1 sheet	1 sheet	1 sheet	1 sheet	1 sheet	1 sheet
Sifters	N/A	N/A	N/A	N/A	2	N/A
Pillowcases+	N/A	N/A	N/A	N/A	4	N/A
Cooler (storage)	1	1	1	1	1	1
Galvanized nails*	N/A	40	N/A	N/A	N/A	N/A
Flagging tape (roll)+*	2	=	=	=	=	=
Brown bags+*	N/A	N/A	N/A	N/A	20	N/A
+ Equipment that needs to be restocked after each trap cleaning at each study site. * Equipment that needs to be restocked for each additional study site that is used.						

Table 14.3. Estimates of minimum equipment costs for individual traps and trap replicates. Total costs are summarized for each trap/replicate and for total trap/replicates. These projected costs are for one sample site.

Trap Type						
	Malasie	***Pitfall**	**+Pan**	**Lindgren**	**Extraction**	**Total Cost**
Single Trap	$300.00	$2.50	$3.00	$47.00	$82.00	$434.50
# of Traps/Site	3	10	10 (12)	12	6	
Trap Replicates	$900.00	$25.00	$30.00	$569.00	$492.00	$2016.00
Grand Total/Site $2016.00						
*Pitfall traps include cups and covers +(12) pans are the pans placed under the Malaise traps						

Table 14.4. Estimates of minimum costs for installation of a complete set of arthropod sampling traps for one site. Minimum costs are included for complete trap servicing at one site, at one time (collection of trapped contents). Costs are given at 12 times, which span the entire active season (collections are bi-monthly for 6 months). Further details are outlined in the section on Monitoring Protocols and Logistics.

Traps	**Malasie (3)**	**Pitfall (25)**	**Pan (10)**	**Lindgren (12)**	**Soil (6) - Litter (3)**
Installation (all traps) Cost $180.00	12 hours --				
*Single trap servicing (all traps) Cost $ 90.00	6 hours--				
**Service frequency (all traps) Cost $1080.00	12 visits--				
Total cost $1350.00					

*Calculated: 2 persons, 3 hours each, at $15/hour
**Details of service frequency (bi-monthly, 6 months) are presented in the section on Monitoring Protocols and Logistics.
Note: Total cost is based on an experienced field crew of 2 people, working for $15.00/hour and does not include travel time or benefits.

Table 14.5. Summary statistics of the costs associated with sorting 30 Malaise trap samples collected from a single study site on Vancouver Island (N. Winchester, unpubl. data). Total cost is the product of the mean sort time, recommended number of traps per site (3) and recommended trap servicing for the entire sample season (12), for one site. Costs are presented for ordinal level sorting and for a selected set of families (17) from the order Diptera.

Malaise Traps (n = 30)					
	Mean	Std. Dev.	Min.	Max	Range
# orders	14.9	2.5	8	19	11
# individuals	6482	3687	1214	14088	12874
Sort time (hrs.)	9.3	5.9	2.5	2.6	23.5
Family Sort (Order Diptera) (Target = 17 families)					
# families	5	3	1	15	14
# individuals	5342	3321	852	12183	11331
Sort time (hrs.)	2.8	4.5	.25	18.5	18.25
Cost: Order level sort @ $15.00/hour: 9.3 x 15 = $139.50. Total = $139.50 x 3 x 12 = $5022.00 Family level sort @ $15.00/hour: 2.8 x 15 = $42.00. Total = $42.00 x 3 x 12 = $1512.00					

Table 14.6. Summary statistics of the costs associated with sorting 30 samples collected from a single study site over the sample interval June-October inclusive, for the recommended protocols using Pitfall traps to monitor terrestrial arthropods. Total cost is the product of the mean sort time, recommended number of traps per site (10) and recommended trap servicing for the entire sample season (12).

Pitfall Traps (n = 30)					
	Mean	Std. Dev.	Min.	Max	Range
# orders	3.47	2.57	2	4	2
# individuals	72	46.54	12	177	165
Sort time (hrs.)	0.5	0.14	0.25	0.75	0.5
Cost: Order level sort @ $15.00/hour: 0.5 x 15 = $7.50. Total = $7.50 x 10 x 12 = $900.00					

Table 14.7. Summary statistics of the costs associated with sorting 30 samples collected from a single study site over the sample interval June-October inclusive, for the recommended protocols using Pan traps to monitor terrestrial arthropods. Total cost is the product of the mean sort time, recommended number of traps per site (22) and recommended trap servicing for the entire sample season (12).

Pan Traps (n = 30)					
	Mean	Std. Dev.	Min.	Max	Range
# orders	10.3	2.6	7	16	9
# individuals	426	383	40	2211	2171
Sort time (hrs.)	2.2	1.2	.5	6	5.5
Cost: Order level sort @ $15.00/hour: 2. x 15 = $33. Total = $33 x 22 x 12 = $8712					

Table 14.8. Summary statistics of the costs associated with sorting 30 samples collected from a single study site over the sample interval June-October inclusive, for the recommended protocols using Lindgren traps to monitor terrestrial arthropods. Total cost is the product of the mean sort time, recommended number of traps per site (12) and recommended trap servicing for the entire sample season (12).

Lindgren Traps (n = 30)					
	Mean	Std. Dev.	Min.	Max	Range
# orders	10.3	2.6	7	16	9
# individuals	426	383	40	2211	2171
Sort time (hrs.)	2.2	1.2	.5	6	5.5
Cost: Order level sort @ $15.00/hour: 0.18 x 15 = $2.70. Total = $2.70 x 12 x 12 = $388					

Table 14.9. Specimen preparation costs involved with the processing of 3000 staphilinid beetles from 180 Malaise traps. All specimens were collected and processed by N. Winchester. Specimens were collected in 1992, over a 6 month period, June-November, inclusive. Costs do not include basic laboratory costs such as scopes, computer, basic lab supplies (e.g. forceps, paper towels).

Equipment costs		
Initial storage	+Glass vials (12 ml, allergy) (180	$48.05
	+Neoprine stoppers	$29.34
	*Ethyl alcohol (20L)	$72.48
Mounting	+Petri dishes (40, 100 x 15mm)	$65.00
	Filter paper (100, 100 x 15mm)	$8.81
	Insect pins (e.g., 3000 #2, Anticorro)	$436.50
	Points (Cost to make, 2500 @ $15/hr.)	$225.00
	*Insect glue (1 bottle, white)	$5.20
	*Labels (500 sheets, 8.5 x 11 Navajo Test, White, 80#)	$63.15
	Completed labels (3000, computer generated @ $15/hr)	$60.00
	*Schmid boxes (12)	$480.00
Mounting costs	@ $20.00/hr., trained, experienced technician. Total time 8 hours.	$160.00
TOTAL COST		$1653.53
* Bulk supplies that can be used in processing other groups. + Supplies that can be reused after specimen identification is complete		

Table 14.10. Pilot project field design costs for a short-term study (one-year) of ground arthropods. Costs for equipment, installation and servicing are detailed in the section entitled “Field Resource Costs: Equipment and Installation”.

Optimal Design: 18 sites (3 sites per 6 ecozones) and 12 sample times						
	Traps					
	Malaise	Pitfall	Pan	Light	Lindgren	Extraction
Equipment	$16,200	$450	$540	$40,500	$10,243	*N/A
Cost: $67,933						
Installation and complete servicing of all traps @ $15.00 for one site: $3120.00						
Cost: $56,160.00 (3120.00 x 3 sites x 6 ecozones)						
Specimen sorting (ordinal level)	$90,396	$16,200	$156,816	N/A	$6,998	*N/A
Cost: $270,410						
Specimen Preparation Example target group: O. Coleoptera, F. Staphylinidae						
Cost based on 9,000 specimens: $4,960						
Total costs: $399,463 (costs for a single site are $399,463/18 = $22,192); costs for a single site entry are $22,192/12 = $1,849).						
* N/A: Costs are related to in-lab costs of extractors. The price is variable; therefore, no estimate is given. Note: These costs do not include identification and archiving sorted samples and residues.						

Table 14.11. Reduced costs scenario 1 for the pilot study field design for a short-term study (one year) of ground arthropods. . Costs for equipment, installation and servicing are detailed in the section entitled "Field Resource Costs: Equipment and Installation".

Reduced Design: 9 sites (3 sites per 3 ecozones) and 12 sample times						
	Traps					
	Malaise	Pitfall	Pan	Light	Lindgren	Extraction
Equipment	$8,100	$225	$270	Delete	$5121	*N/A
Cost: $13,716						
Installation and complete servicing of all traps @ $15.00 for one site: $3120.00						
Cost: $28,080 (3120.00 x 3 sites x 3 ecozones)						
Specimen sorting (ordinal level)	$45,198	$8,100	$78,408	N/A	$3499	*N/A
Cost: $135,205						
Specimen Preparation Example target group: O. Coleoptera, F. Staphylinidae						
Cost based on 9,000 specimens: $4,960						
Total costs: $181,962						
* N/A: Costs are related to in-lab costs of extractors. The price is variable; therefore, no estimate is given. Note: These costs do not include identification and archiving sorted samples and residues. Light traps are dropped from the sampling program.						

Table 14.12. Reduced costs scenario 2 for the pilot study field design for a short-term study (one year) of ground arthropods. . Costs for equipment, installation and servicing are detailed in the section entitled "Field Resource Costs: Equipment and Installation".

Reduced Design: 9 sites (3 sites per 3 ecozones) and 12 sample times						
	Traps					
	Malaise	Pitfall	Pan	Light	Lindgren	Extraction
Equipment	$8,100	$225	$135	Delete	$5121	*N/A
Cost: $13,581						
Installation and complete servicing of all traps @ $15.00 for one site: $3120.00						
Cost: $28,080 (3120.00 x 3 sites x 3 ecozones)						
Specimen sorting (ordinal level)	$45,198	$8,100	$39,204	N/A	$3499	*N/A
Cost: $96,001						
Specimen Preparation Example target group: O. Coleoptera, F. Staphylinidae						
Cost based on 9,000 specimens: $4,960						
Total costs: $129,055						
* N/A: Costs are related to in-lab costs of extractors. The price is variable; therefore, no estimate is given. Note: These costs do not include identification and archiving sorted samples and residues. Light traps and Malaise pan traps are dropped from the sampling program.						

Table 14.13. Reduced costs scenario 3 for the pilot study field design for a short-term study (one year) of ground arthropods. Costs for equipment, installation and servicing are detailed in the section entitled "Field Resource Costs: Equipment and Installation".

<table>
<tr><td colspan="7">Reduced Design: 9 sites (3 sites per 3 ecozones) and 3 sample periods</td></tr>
<tr><td></td><td colspan="6">Traps</td></tr>
<tr><td></td><td>Malaise</td><td>Pitfall</td><td>Pan</td><td>Light</td><td>Lindgren</td><td>Extraction</td></tr>
<tr><td>Equipment</td><td>$8,100</td><td>$225</td><td>$135</td><td>Delete</td><td>$5121</td><td>*N/A</td></tr>
<tr><td colspan="7">Cost: $13,581</td></tr>
<tr><td colspan="7">Installation and complete servicing of all traps
@ $15.00 for one site: $780.00</td></tr>
<tr><td colspan="7">Cost: $7020 (780.00 x 3 sites x 3 ecozones)</td></tr>
<tr><td>Specimen sorting (ordinal level)</td><td>$11,300</td><td>$2,025</td><td>$8910</td><td>N/A</td><td>$875</td><td>*N/A</td></tr>
<tr><td colspan="7">Cost: $23,110</td></tr>
<tr><td colspan="7">Specimen Preparation Example target group: O. Coleoptera, F. Staphylinidae</td></tr>
<tr><td colspan="7">Cost based on 4,500 specimens: $2,480</td></tr>
<tr><td colspan="7">Total costs: $46,190</td></tr>
<tr><td colspan="7">* N/A: Costs are related to in-lab costs of extractors. The price is variable; therefore, no estimate is given.
Note: These costs do not include identification and archiving sorted samples and residues.
Light traps and Malaise pan traps are dropped from the sampling program.
Sampling frequency has been reduced to 3 times (early, mid, late season) per site (9 sites).</td></tr>
</table>