# TERRESTRIAL ARTHROPOD BIODIVERSITY: PLANNING A STUDY AND RECOMMENDED SAMPLING TECHNIQUES

A Brief Prepared by the Biological Survey of Canada
(Terrestrial Arthropods)

1994

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## Abstract

Knowledge of biodiversity is important for wise management and use of the earth's resources. Terrestrial arthropods (insects and their relatives) are by far the most diverse groups of animals and important contributors to biodiversity. However, a synopsis of techniques suitable for assessing diversity for terrestrial arthropods is not readily available to many of those responsible for general assessments of biodiversity. This brief therefore offers general guidelines for planning a study of arthropod biodiversity, including attention to long-term planning, choice of taxonomic groups, and the resources required for sampling, sorting and identification. The brief recommends in some detail the specific sampling methods appropriate for this purpose. It proposes a standard sampling protocol for the assessment of regional biodiversity, suggesting that any such general inventory should include, at a minimum, Malaise, flight-intercept and pan traps, as well as behavioural extractors such as Berlese funnels, and it presents some estimates of the time required to process samples, for use in planning a budget. The major current impediment to properly planned and executed studies of arthropod diversity is the limited number of systematics experts available to identify species. Resources for systematics support therefore should be included in project budgets.

# BIODIVERSITÉ DES ARTHROPODES TERRESTRES: PLANIFICATION D'UNE ÉTUDE ET TECHNIQUES D'ÉCHANTILLONNAGE RECOMMANDÉES


## Résumé

La connaissance de la biodiversité est importante si l'on veut effectuer une gestion sage de l'utilisation des ressources de la terre. Les arthropodes terrestres (les insectes et leurs parents) constituent de loin les groupes d'animaux les plus diversifiés et contribuent grandement à la biodiversité. Bon nombre des personnes responsables des évaluations générales de la biodiversité ne disposent toutefois pas d'un sommaire des techniques convenables pour l'évaluation de la diversité des arthropodes terrestres. Le présent précis offre donc les grandes lignes pour la planification d'une étude de la biodiversité des arthropodes, dont une prise en considération de la planification à long terme, du choix des groupes taxinomiques et des ressources requises pour l'échantillonnage, le tri et l'identification des arthropodes. Le précis recommande et décrit, en détail, des méthodes d'échantillonnage particulières convenant à cette fin. Il propose un protocole d'échantillonnage standard pour l'évaluation de la biodiversité régionale, mentionnant que tout inventaire général de ce genre devrait comprendre, au moins, des pièges de Malaise, l'interception d'arthropodes en vol et des bacs jaunes, en plus d'extracteurs éthologiques tels que l'extracteur de Berlese. De plus, il donne une certaine évaluation du temps requis pour traiter les échantillonnages, ce qui peut faciliter la planification d'un budget. Le nombre limité d'experts systématiques pouvant identifier les espèces constitue présentement le principal obstacle à la planification et l'exécution d'études sur la diversité des arthropodes. Il faudrait donc inclure dans les budgets des projets des ressources pour l'appui systématique.

## INTRODUCTION

Biodiversity has received recent national and international recognition. The importance of biodiversity arises from the fact that the world depends on self-sustaining biological systems that include many kinds of organisms. Knowledge of biodiversity is required to understand the natural world and the natural and artificial changes it may undergo; and in turn, such knowledge permits the wise use and management of ecosystems, both as elements of natural heritage and as reservoirs of actual and potential resources.

Although biodiversity has been recognized as important in this general context, not all of the methods for the actual study of diversity in particular habitats are well known. In particular, standard methods are required to assess the overwhelming numbers of insects, mites, spiders, and their relatives, which form more than 75% of the world's known species, and present the greatest problems in arriving at estimates of regional biodiversity.

This brief[1] focusses on sampling methods appropriate to assess the taxonomic diversity of terrestrial arthropods. It also emphasizes the importance of proper long-term plans for any study of biodiversity. Carefully developed plans are especially critical for arthropod inventories because extensive and repeated sampling is required to capture the many species with widely different habits, very large amounts of material are generated, and it is difficult to identify many of the species.

## BIODIVERSITY

Biological diversity, or "biodiversity", has been used to refer to almost any measure (taxonomic, numerical, genetic, etc.) of the variety of organisms that live in a particular place. Although many different definitions of biodiversity have been developed for particular uses, the focus here is on taxonomic diversity or species richness–the total number of kinds of organisms within a given area, habitat, or community. Such assessments emphasize species, the functioning entities in nature and the categories by which all biological information is organized and retrieved. An emphasis on the methods required to obtain reliable measures of species richness recognizes sampling and identification of species as the essential baseline for understanding diversity: properly conducted inventories are the core of future endeavours.

> "Conservation of biodiversity is more than an aesthetic or moral issue; it is integral to our health and economy" (Standing Committee on Environment 1993: 22)

Several types of analysis that use additional information, notably species abundance as well as the number of species present, generate indexes of diversity of potential

[1]Prepared by a subcommittee of the Survey: S.A. Marshall, R.S. Anderson, R.E. Roughley, V. Behan-Pelletier, and H.V. Danks

value for understanding community structure (e.g. Pielou 1975; Kempton and Taylor 1976; Southwood 1978; Magurran 1988). Such analyses are beyond the scope of this brief. However, many of the techniques listed here are suitable for simultaneous assessments of taxonomic and community diversity, especially by standardizing sampling effort.

## PLANNING A STUDY

It is important to emphasize at the outset that it is not now possible, nor will it be possible in the foreseeable future, to inventory more than a carefully chosen subset of the arthropod fauna of a region or habitat, because as many as half of our insect species either are undescribed or cannot be identified at present. Consequently, any study of arthropod biodiversity must be planned very carefully (Table 1). Rosenberg et al. (1979) provide a general discussion of Canadian arthropod surveys and the elements of an ideal survey.

Table 1. Components of a properly planned biodiversity study

- Establish goals
- Select groups for study
- Decide how to deal with unnameable species
- Assemble financial resources for sampling and sorting
- Arrange for resources and systematics expertise for identification
- Define sampling methods
- Ensure follow-up (curation of voucher specimens, publication of results, etc.)

### *Goals*

The goals of a study dictate its plan. A general inventory requires substantially different resources and sampling protocols than a more specific study. What are the best means of trapping a diversity of predatory beetles? Will specimens be released following their capture, requiring live-traps to be used? Will a systematist be willing to identify moths taken from a fluid preservative? These and other questions must be anticipated and dealt with early in the planning process. Sampling protocols might also be selected to allow the results of a study to be compared in a standard way to previous studies or to other ongoing studies.

### *Groups*

It is not normally feasible to inventory all taxa of terrestrial arthropods. The choice of taxa to be inventoried is governed mainly by the goals of the study, available resources and systematics support. Several arthropod taxa can be identified as meeting criteria such as habitat specificity, diversity, vagility, or other biological attributes relevant to the goals of the study, but it will be necessary to choose practical candidates from that list of potential taxa. Probably the best way to narrow the list is by assessing the available systematics support (see below).

*Unnameable species*

Because relatively few groups of terrestrial arthropods are well known, it will not be possible to name many of the species collected, especially in diverse groups of small body size. There are four ways to deal with these "unnameable" species:

- Taxa are sorted to morphospecies and assigned an identifying number (e.g. Genus A sp. 1). This method allows species diversity to be assessed, but requires a competent systematist to sort the taxa into morphospecies. It prevents extraction of information on species biology from the literature, and prevents comparisons with other studies unless the specimens from the other study have been similarly treated by the same systematist.

- Taxa are treated at some higher taxonomic level (e.g. Genus A spp.). This method is of limited value, because even though it requires accurate sorting, preferably but not necessarily by a competent systematist, it masks significant differences in natural history attributes of congeneric species, prevents detailed analysis and prohibits detailed comparisons with other studies.

- Only taxa that can readily be identified are included. If several taxa meet the criteria defined by the goals of the study, candidates can be selected from that list of potential groups. As already noted, one of the best ways to narrow the list of otherwise suitable potential candidate taxa is to look at the available systematics support. What can be identified to the desired or required level? A few groups, notably butterflies and large moths, dragonflies, and other groups covered by comprehensive handbooks such as those published in the Agriculture Canada "Insects and Arachnids of Canada" Handbook series, might be identifiable without direct involvement by professional systematists. Such readily identifiable arthropod taxa remain the exception and not the rule, and participation by professional systematists is nearly always required. Although such a procedure makes a study with limited support feasible, it does little to resolve the lack of knowledge about many insect groups.

- Resources are acquired to resolve systematics problems. Biodiversity proposals that aim to solve rather than avoid the problems created by arthropod biodiversity should include a budget item for support of professional or student systematists to study the taxonomy of several of the key groups being sampled. This is the optimum solution to the problem of unnameable species.

*Resources for sampling and sorting*

Because arthropod diversity is high and large numbers of specimens have to be processed, any sampling programme involving arthropods is time consuming and expensive. Removal of specimens from bulk samples, and the preparation of those specimens for identification, can take up to several hours per sample depending on the type of sample, the taxa removed, the fraction of the material prepared, preparation

techniques, and the experience of the person doing the processing. One worker found that removal of all beetles, Hymenoptera, and spiders from flight-intercept traps in Montana (without any attempt to separate families) took from 40 minutes to 5.7 hours per trap depending on the experience of the individual doing the work (M.A. Ivie, pers. comm.). When studying abundant groups, such as many families of Diptera, it is unlikely to be practical to prepare all the specimens in a sample, and subsampling is therefore necessary. A subsample of 8,000 Brachycera (higher Diptera) from one summer's pan-trap samples from an Ontario old-growth forest required about 800 hours of student assistance to sort, mount and label. Estimates of the time required for processing and sorting specimens from given trap samples therefore are very difficult to arrive at, but some minimum estimates are indicated in Table 2. Note the caveats given in the caption, and also the fact that identification during such processing would be to the family level only; in most instances, further identification to the generic or species level would be made by professional systematists.

Biodiversity proposals should include a budget item for support of systematists.

For any single site, estimated monthly processing time using the recommended sampling protocol for terrestrial arthropod inventory amounts to 504 hours. Using biology undergraduate students at a rate of $10 per hour, the cost of processing material collected at this one site per month would be $5,040. Thus for any single-site inventory carried out in most areas of Canada, processing for the seven-month period from April to October would cost $35,280. Compared to the processing costs other expenses (supplies, travel) are minimal, but note that these processing costs are incurred to reach only the stage at which species identification becomes possible.

### *Resources for identification*

At present, there are very few systematists both willing and able to identify large numbers of specimens in support of biodiversity studies, especially because the numbers of professional systematists in Canada and elsewhere, both in government and universities, and support for systematic biology in general, have declined greatly in recent years (Hunter 1991; Wiggins 1992; Heraty 1992). In other words, there is a shortage of highly trained specialists, and their willingness to participate in a study should not be taken for granted.

All systematists require that specimens submitted for identification be appropriately prepared and properly labelled, and this takes substantial time, effort and expertise. Good samples of well prepared material facilitate accurate identifications and may subsequently prove of value in the personal research programme of participating systematists. Adequate numbers of specimens always should be submitted from the various sampling

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Table 2. Estimates of minimum time required to process selected samples from one site using recommended sampling methods and protocol (see Table 3). These minimum estimates apply only to experienced sorters, sorting specimens from a small number of selected families (or subsamples when specimens are numerous) to the family level only. Beginning sorters take much longer, but steadily reduce the time as they gain experience. Based chiefly on information provided by Scudder (in press).

| Sampling Method | Sort | Preparation | Identification | Total time required (hrs) per sample | Traps or samples | Times | Total per month using recommended protocol |
|---|---|---|---|---|---|---|---|
| Malaise trap | 1.5 | 3.5 | 5 | 10 | 2 | 4 | 80 |
| Flight intercept trap | 3 | 5 | 8 | 16 | 2 | 4 | 128 |
| Pan trap | 1.5 | 3.5 | 5 | 10 | 4 | 4 | 160 |
| Pitfall trap | 0.5 | 1 | 2 | 3.5 | 4 | 4 | 56 |
| Behavioural extractors | 3 | 5 | 8 | 16 | 10 | 0.5 | 80 |
| Totals | 9.5 | 18 | 28 | 55.5 | — | — | 504 |

sites and times of year to permit the recognition of rare species. For some taxa, submission of subsamples or selected representatives only should be avoided.

Listings of systematists are available in Arnett and Arnett (1993), Ananthakrishnan (1991) and, for Coleoptera, Noonan *et al.* (1993). Other specialized listings are available. Coddington *et al.* (1991) have proposed the establishment of a global network of systematists under the auspices of the IUBS-SCOPE-UNESCO Program on biodiversity. Listings of arthropod systematists willing to receive material appear periodically in the *Newsletter of the Biological Survey of Canada (Terrestrial Arthropods)*. However, the fact that a systematist exists does not indicate ability or willingness to become involved in a study. Any prospective participants should be consulted well in advance of the initiation of sampling and made aware of what is expected of them as well as of the logistics and ultimate goals of the survey. Such systematists can provide constructive suggestions as to the sampling protocol.

A few systematists are able to do identifications simply because it is part of their job, but more often some kind of incentive is required in the form of coauthorship, specimens of use in the systematist's research, reciprocal identifications, or financial compensation for the time and resources involved. In particular, as emphasized above

for the treatment of unnameable species, specific support for systematics resources should be included in plans for biodiversity studies, because it will often be necessary to fund professionals, post-doctoral associates, or students directly to ensure that identifications will be made, rather than relying on mainly volunteer effort.

### *Sampling Protocol*

Once target groups have been decided, sampling methods can be chosen. The second part of this brief provides information relevant to this choice, and also indicates the sorts of techniques necessary for an appropriate general inventory.

### *Time frame and Follow-up*

Results from a given study of biodiversity are useful only if they reach a stage at which the information can be communicated and used to add to the store of knowledge about the taxa and sites being studied. Therefore, a realistic time frame to complete the study, and appropriate reference materials, must be established.

Planning a realistic time frame is especially important because if resources to complete all aspects of a biodiversity study are not budgeted, much of the initial effort may be wasted; it is easier to sample than sort, easier to sort than identify, and so on. Therefore, the most difficult stages of a project to complete and to fund are the later ones, comprising identification, publication of results, and curation of specimens; yet these three elements are especially critical. Identification to species not only provides names that allow information to be recorded for future use, but also gives access to existing information for comparison. Publication of results forces unfinished analyses to be completed and anecdotal ideas to be validated, and makes information available in a standardized format that is much more reliable than some sort of informal report. Deposition and curation of voucher specimens in museums – which have a long-term commitment to specimen maintenance – provides reference material for both ecological information (species associated with particular habitats, for example) and taxonomic information (Danks *et al.* 1987). Future taxonomic study may improve the understanding of particular taxa; if voucher specimens exist, the specimens can be re-examined to ensure that the information associated with them is attached to valid species names.

A realistic time frame, and appropriate reference materials, must be established.

## CHOOSING SAMPLING METHODS

### *Sampling arthropods: General guidelines*

Either active or passive methods can be used to sample arthropods. Active methods require collection by an individual using various kinds of equipment. Passive methods establish specialized types of traps at sampling stations in the field, which are serviced at given intervals. Passive traps collect large numbers of specimens, and generally remove the bias introduced by the different abilities of individuals to collect specimens

by active methods. Passive sampling is less labour intensive, and also captures many species not commonly taken by active collection (although active collecting is very valuable too in some groups). For these reasons, passive sampling methods are most frequently used in sampling arthropod biodiversity.

Most passive traps left in the field for a period of time require the use of a preservative, generally a fluid. A solution in water of common salt and detergent (wetting agent) is most commonly used for frequently serviced traps, and ethylene or propylene glycol is commonly used for traps that must be left for long periods or traps in desiccating environments. Photographic soap is a powerful, non-scented wetting agent, and automobile antifreeze ["Prestone"] mixed 1:1 with water provides a satisfactory alternative to expensive laboratory grade ethylene glycol. Different brands of antifreeze have different properties, so standardization is important. Unfortunately, ethylene glycol is highly toxic and attractive to vertebrates, a potential problem which can be solved by the addition of a bitter substance such as quinine sulfate or by the use of propylene glycol. If possible, it is preferable to sample more frequently so that toxic preservatives are not required. For most temperate environments, salt solutions give adequate protection from decay for about one week. Obviously, metal containers should not be used with salt solutions.

All passive trapping methods require the removal of specimens from the trap and their placement in a container in preservative until the samples can be processed. Most trap types can be serviced with minimal habitat disturbance by removing the specimens using an aquarium net with a fine mesh (to ensure that all sizes of specimens are retained), then rinsing the specimens very gently with water prior to storing the sample in 80% ethanol. The ethanol should be replaced 1 to 2 days later to avoid excessive dilution by the water added in the cleansing process. Addition of 5% acetic acid to the ethanol will prevent specimens from becoming excessively brittle and will facilitate dissections. We do not recommend the use of methanol or, especially, formalin as preservatives. "Whirl Pac", small "Zip-loc" plastic bags, or plastic "margarine" containers are ideal for the initial or field phase of specimen storage. Transfer to glass or polypropylene jars can be made later in the laboratory, ideally at the time fresh ethanol is being added.

### *Choosing methods for use in a study*

The scope of any study is limited by the sampling methods chosen. Southwood (1978) provides descriptions and an extensive and useful discussion of most techniques for assessing insect populations. Other useful overviews of arthropod sampling techniques can be found in Martin (1977), Disney *et al.* (1982), Canaday (1987), Steyskal *et al.* (1986), Gadagkar *et al.* (1990), and Górny and Grüm (1993). Some of the most commonly and

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widely used sampling methods, their limitations and their relevance to biodiversity studies are discussed below. The taxa most frequently collected by each technique are noted, and standardized sampling protocols are proposed for most techniques.

Table 3. Recommended sampling methods for use in general inventory of arthropod biodiversity.

| Sampling Method | Principle | Format | Variables | Benefits |
|---|---|---|---|---|
| Malaise trap | Flying organisms encounter mesh panel and fly up into trap head | Erect mesh panel guyed and supported by 2 poles at ends of panel; panel assymmetrical with one end higher than the other and collecting head at higher end | -Type of mesh<br>-Method of guying<br>-Dimensions<br>-Fluid preservative<br>-Orientation<br>-Possible combination with flight-intercept and pan traps | -Large numbers of specimens<br>-Not labour intensive<br>-Widely used and easily standardized<br>-Combinations with other methods |
| Flight-intercept trap | Flying organisms encounter mesh panel and drop to ground | Erect mesh panel guyed and supported by 2 poles at ends of panel; panel symmetrical with pan traps placed underneath panel | -Type of mesh<br>-Method of guying<br>-Dimensions<br>-Fluid preservative<br>-Orientation<br>-Possible combination with flight-intercept and pan traps<br>-Use of roof | -Large numbers and high diversity of rare /cryptic taxa<br>-Not labour intensive<br>-Easily standardized<br>-Simple construction<br>-Combinations with other methods |

*Conducting an inventory*

Recommended methods for conducting an inventory of a good cross section of the arthropod biodiversity of a site are listed in Table 3. These methods include Malaise, flight-intercept, and pan traps, and the use of behavioural extractors. All methods are easily standardized, cost- and labour-effective, and provide a diversity of high quality material (if serviced as suggested). An accurate picture of diversity requires multiple-year sampling, because time of occurrence and abundance of many species differs from year to year (compare Owen 1991).

| Limitations | Sampling Period | Recommended Standard | Taxa Collected |
|---|---|---|---|
| -Complex construction<br>-Expensive<br>-Samples only selected portion of aerial fauna<br>-Tourists possible<br>-Visible; subject to damage /vandalism<br>-Immatures unlikely | -1 to 7 days | -Townes (1972) style with black mesh<br>-Dimensions (see Townes 1972)<br>-Head preservative 80% ethanol with 5% acetic acid<br>-7-day intervals<br>-2 traps per site, oriented 90°; spaced 25 m apart | -Strongly flying, positively phototactic taxa<br>-Hymenoptera (large)<br>-Diptera<br>-Some Coleoptera, Lepidoptera, other orders |
| -Subject to flooding<br>-Tourists possible<br>-Visible; subject to damage/vandalism<br>-Immatures unlikely | -1 to 7 days | -1.25 m (4′) x 1.85 m (6′) fine black mesh panel<br>-Roof used (see text)<br>-6 pans (see Pan trap; Standard) under central panel; pans sunk flush with ground<br>-Use salt, soap, water<br>-7-day intervals<br>-2 traps per site, oriented 90°; spaced 25 m apart | -Weakly flying, negatively phototactic taxa<br>-Coleoptera (small)<br>-Hymenoptera (small)<br>-Diptera (small) |

Table 3 (*continued*) Recommended sampling methods.

| Sampling Method | Principle | Format | Variables | Benefits |
|---|---|---|---|---|
| Pan trap | Organisms fall or fly into shallow pan placed on substrate | Shallow pans of fluid set into/on substrate | -Position of pan lip with respect to substrate surface<br>-Pan shape, size, depth<br>-Colour of pan<br>-Pan material<br>-Fluid preservative<br>-Use of roof | -Inexpensive<br>-Supplies readily available<br>-Not labour intensive<br>-Large numbers of small cryptic flying taxa |
| Pitfall trap | Organisms fall into deep container placed in substrate | Cylindrical container sunk flush with ground substrate | -Baited/unbaited<br>-Size, depth of container<br>-Container material<br>-Fluid preservative<br>-Use of cover | -Inexpensive<br>-Supplies readily available<br>-Frequently used in previous studies<br>-Limitations extensively studied<br>-Live-trapping possible<br>-Baiting possible<br>-Immatures possible |
| Behavioural extractors | Organisms of limited vagility resident in a substrate type are extracted through the use of heat and desiccation | Tapered cone-shaped container with sunken hardware cloth platform on which samples are placed; cover and source of heat from above; sample collection container below | -Size of sample<br>-Type of litter collected<br>-Use of sifter<br>-Length of time for extraction<br>-Energy source<br>-Collect live or into preservative<br>-Amount of litter processed | -Collection of numerous cryptic organisms not otherwise collected<br>-Association of adults and immature stages<br>-Live-collecting possible<br>-Residents only<br>-Can be standardized |

| Limitations | Sampling Period | Recommended Standard | Taxa Collected |
|---|---|---|---|
| -Live-trapping not possible<br>-Evaporation rates high<br>-Flooding possible<br>-Baiting not possible<br>-Tourists possible<br>-Immatures unlikely | -1 day to 1 month | -Use "Showcase" trays, 500 ml, 15 x 17 cm (see text)<br>-Exterior of trays painted yellow<br>-Trap lip flush with substrate surface<br>-Use salt, soap, water<br>-7-day intervals<br>-Use with plastic roofs<br>-4 traps per site; spaced 10 m apart | -Small taxa living on/near ground substrate<br>-Hymenoptera (small), "Parasitica"<br>-Diptera (small)<br>-Coleoptera (small)<br>-Arachnida |
| -Possible biased sampling<br>-Flooding possible<br>-Tourists possible | -1 day to 1 month | -Use plastic 450 ml (16 oz.) beer cups<br>-Use salt, soap, water<br>-7-day intervals<br>-Use with covers (see text)<br>-4 traps per site; spaced 10 m apart | -Ground-dwelling, active taxa<br>-Coleoptera<br>-Arachnida<br>-Collembola |
| -Very labour intensive<br>-Specialized equipment needed<br>-Complex construction<br>-Energy source needed | -4 times per year (in season) per site; 10 samples per period | - 30 cm (12") tapered metal cones (Martin 1977: 48) using electricity; 60W bulbs, 10 cm (4") from litter, process for 8 hrs; 1 litre litter per funnel, 3 funnels per sample<br>-Uniform field sampling effort per sample; vary microhabitat selection | -Small, cryptic taxa largely of limited vagility<br>-Coleoptera<br>-Hymenoptera (ants, some Parasitica)<br>-Hemiptera<br>-Arachnida (Araneae, Acari)<br>-Diplopoda<br>-Chilopoda |

## SAMPLING METHODS: PASSIVE COLLECTION TECHNIQUES

### •Malaise Traps

A diverse sample of winged insects can be taken using traps which collect flying insects. Various designs of Malaise traps, which essentially are open-sided tents that intercept flying insects and direct them to some sort of trap head, are readily available, many based on Townes' (1972) design. All take large numbers of insects, either into fluid or a dry container. Malaise traps are an efficient way to collect most active, flying groups such as Tachinidae, aculeate Hymenoptera, and Ichneumonidae and are an excellent complement to substrate sampling methods. They tend to take large numbers of non-habitat-associated species ("tourists" or vagile species breeding in nearby habitats), and require frequent servicing if some of the specimens (especially large Diptera) are to be kept in adequate condition for specific identification.

Malaise trap samples vary widely depending on trap design (Disney *et al.* 1982), mesh size (Darling and Packer 1988), and colour (Roberts 1972). These factors can be corrected for by using standard designs available from several commercial sources. However, Malaise trap efficiency also varies with aspects of installation. Factors such as direction of prevailing winds, likely flight paths, Malaise head position, and even how tightly the trap is guyed markedly effect both numerical and taxonomic composition of the catch. Nonetheless, Malaise traps are arguably the most effective way to trap large numbers of insect species at a given site, but because they sample only the flying stages effectively, they should be used together with other trapping techniques in attempts at exhaustive inventory.

Malaise traps in combination with pan traps can take overwhelming numbers of specimens. Finnamore (1994) estimated that one season's catch in 39 pan traps plus one Malaise trap placed in a fen complex near Edmonton, Alberta, totalled approximately 1.5 million specimens.

Malaise traps with dry trap heads, usually using dichlorvos or pyrethroids as a killing agent, must be emptied daily, and the samples either processed immediately or stored in a freezer. Most extensive Malaise trap surveys use trap heads that funnel the catch into alcohol or some other preservative. Unfortunately, this renders the Lepidoptera and larger Diptera difficult to identify, but it is satisfactory for most other taxa. If traps cannot be serviced frequently, foil can be wrapped around the outside of the Malaise head container to limit evaporation of the preservative.

We recommend the use of 2 Townes-style Malaise traps (Townes 1972) made of fine black polyester mosquito netting (sold for use in tent fly screens) per site. Trap dimensions should be those of Townes (1972). Trap heads should contain 80% ethanol with 5% acetic acid and should be serviced at weekly intervals.

### •Flight-Intercept Traps

Many flying insects, especially small or weak flying species, drop down when they encounter a Malaise trap baffle rather than moving up into the trap head. These species

can be collected by putting pan traps under the centre panel of a Malaise trap. Pan traps set under a Malaise take a substantially different insect assemblage than do Malaise trap heads (Marshall 1979; O'Hara 1988). Alternatively, it is possible to simply stretch an interception panel, usually made of black mesh, above either a series of pan traps arranged in a linear fashion, an elongate trough, or even heavy-gauge plastic (preferably yellow in colour) lining a shallow excavation. This kind of trap, frequently termed a flight-intercept trap (FIT), is especially efficient in dense forest for sampling small, flying insects such as small beetles that are otherwise difficult to collect (Peck and Davies 1980). Although similar traps with a pane of glass as the central panel have been used (termed window traps: Chapman and Kinghorn 1955), we suggest the use of a porous mesh as this does not divert or otherwise affect the wind currents on which small, weakly flying insects may float.

Window or intercept traps may also be used above ground level, and modified intercept traps have been used for sampling insects in forest canopies (Basset 1988). Spill-proof pan traps for use as suspended traps in forest insect sampling can be made by cutting an opening on the broad side of a plastic container such as an antifreeze container (Canaday 1987). Intercept-trap efficiency can be increased significantly by treating the interception panel with a contact pyrethroid insecticide such as permethrin ["Ambush"]. Flooding of the traps can be a problem and should be dealt with, as for pan traps, through the use of roofs, drainage holes, or frequent (at least once per week) servicing.

We recommend the use of fine black polyester mosquito netting (the same as that suggested for Malaise trap construction), installed with a row of standard yellow pan traps (see "pan traps") sunk flush with the ground under the centre panel. Suggested dimensions for a standard panel are 1.25 m (4') high by 1.85 m (6') long. We suggest the use of a clear plastic roof stretched tent-like over the central panel. The edges of the roof should not extend below the top of the center panel when viewed from the side. Because the preservative used in the pans under an intercept trap usually has a large surface area, a preservative that evaporates slowly is preferred. Two traps should be installed per site and servicing should be at weekly intervals.

An excellent combination of techniques for long-term survey sites would be a Townes-style Malaise trap (Townes 1972) made of fine black polyester mosquito netting (sold for use in tent fly screens), installed with a row of pan traps under the center panel. This combines the advantages of pan trapping, intercept trapping, and Malaise trapping in a replicable sample station.

**•Pan Traps**

Pan traps provide a very simple, inexpensive sampling technique using shallow pans of fluid, usually set into the substrate such that the lip of each pan is flush with the substrate surface. Pan traps rely on the organism falling or flying into the fluid preservative; collection may be accidental or the insects may be attracted to the colour of the pan or, apparently, to the fluid surface. Several commercially available

containers, such as microwave trays, organizer trays, and food distribution containers make satisfactory pan traps. If studies are to be comparable with one another then pan shape, surface area, colour, material, edge characteristics, repellent or attractive properties of trap fluid, and depth of installation must be standardized.

Traps installed flush with the soil surface differ from traps installed flush with the litter surface (Greenslade 1964), and traps sitting on the substrate surface (sometimes called water traps) collect a different fauna than those set into the substrate (Disney *et al.* 1982). Traps sunk into the substrate take a wider variety of taxa and larger numbers of specimens. Different taxa are attracted to different trap colours (see for example, Finch 1991). Yellow seems to be the most widely used colour at the present time and is especially attractive to various groups of Homoptera and Hymenoptera, although white is more attractive to some Diptera (Disney *et al.* 1982). The material from which the trap is made can affect trap efficiency (Luff 1975); smooth surfaces such as plastic are more efficient than surfaces that tend to become pitted, such as metal. Toxicity, surface tension, and other attributes of trap fluid also influence trap efficiency. Because different taxa are differentially sampled by pan and pitfall traps (Topping 1993), samples of this sort do not necessarily show a close correlation with density.

Although flooding is not often a problem in frequently serviced traps, the addition of a roof prevents traps from being flooded by rain. Such flooding will lead to the loss and degradation of specimens. The addition of roofs must be carefully considered as they add an extra variable to a sampling programme. Roofs attract some organisms seeking shelter, and exclude other organisms that may be unwilling to enter a confined area. Drainage holes provide an alternative method of avoiding flooding. Small, screened, holes near the lip of a trap generally will prevent fluid overflow. Pan traps can be serviced with minimal habitat disturbance by removing the specimens using a fine-mesh aquarium net, then rinsing the specimens very gently with water prior to storing the sample in alcohol.

We recommend rectangular plastic food packaging trays, such as the 500 ml, 15 x 17 cm, "Showcase" trays distributed by Edmeads Packaging, Kitchener, Ontario. This type of readily available product provides a low cost, highly effective pan trap. The outside of these translucent trays should be spray-painted yellow. Traps should be sunk flush with the substrate surface. Current inventory programmes use 4-6 such traps per site; ecological studies will use 10 or more traps, laid out on a grid, per habitat type sampled. Preservative should be salt solution with soap and servicing should be at weekly intervals. Little is known about the effect of trap spacing, a factor which probably will be governed by microhabitat type and variability. Snider and Snider (1986) varied trap spacing from 0.5-4 m in a northern Michigan forest, but found little effect on catches of ground beetles or springtails. Nevertheless, it is probably better to keep traps 5 to 10 m apart.

Pan traps collect a large variety of terrestrial arthropods including large numbers of small arthropods restricted to or living near the substrate surface. Recent pan-trap

surveys of peatlands in Canada have yielded more than 2,000 species of insects in a single year's sampling (Finnamore 1994; Blades and Marshall 1994).

Taxa efficiently sampled using pan traps include spiders, springtails, ground beetles, sphaerocerid flies, mycetophilid flies, alate aphids, leafhoppers, seed bugs, and several microhymenopteran families.

**•Pitfall Traps**

Although the term pitfall trap is sometimes used to refer to any trap sunk in the ground, we here restrict the term to traps of relatively small diameter which are deeper than pan traps and generally placed so that the lip of the trap is flush with the ground surface. Pitfall traps can be simple containers sunk into the substrate or they can be more elaborate devices with funnels directing specimens into the trap (Clark 1992; Rivard 1962). Pitfall traps may have fluid preservative in the bottom of the container, or they may be kept dry or lined with damp paper if living specimens are desired. Traps may be baited, such as with dung or carrion, but this may render them of limited use for quantitative studies (Southwood 1978). Baited pitfall traps do, however, offer an efficient technique for qualitative comparisons of particular communities among sites (Anderson 1982). Unbaited pitfall traps have been extensively used in sampling ground-dwelling beetles, particularly carabids (Dennison and Hodkinson 1984; Luff 1975; Baars 1979). A partial listing of the extensive literature on pitfall traps can be found in Dunn (1989).

We recommend the use of plastic 450 ml (16 oz.) "beer glasses". Four to 10 traps should be installed per site with covers (15-30 cm (6 to 12")-square pieces of plywood raised at the corners and weighted with a stone). The preservative should be salt solution with soap and servicing should be at weekly intervals.

Taxa collected by unbaited traps include many ground-dwelling beetles, spiders, and some springtails and mites. Taxa collected by baited traps depend on the type of bait used (Greenslade and Greenslade 1971; Newton and Peck 1975). Some types of baited traps will attract large vertebrates and precautions must be taken to exclude these scavengers. Unbaited pitfall traps offer an advantage over simple pan traps in that they permit the collection of living specimens which can be counted, identified and released or used in mark/recapture studies.

**•Light Traps**

Several authors have argued that light traps are the best way to undertake insect surveys (Holloway 1980; Gadagkar *et al.* 1990), because they collect well known taxa efficiently, they collect large numbers of insects, and they have been in wide use for a longer period than other mass-trapping devices. Light traps certainly are one of the most powerful collecting tools available, but a large number of variables affects the size and taxonomic composition of light-trap catches (Bowden 1982). Trap size, height, design, surroundings, and bulb type are some of the variables that should be considered. Diptera, for example, are taken in larger numbers in incandescent traps and Lepidoptera

are more abundant in ultra-violet (UV) traps (Southwood 1978). Mercury-vapour light traps will attract a somewhat different fauna than ultra-violet lights. Most light traps use a killing agent such as cyanide, Vapona or ethyl acetate; however, an electrified trap designed by Mizutani *et al.* (1982) bypasses the need for highly toxic killing agents. Some light-trap designs collect insects directly into a fluid preservative, which is suitable for groups such as Hymenoptera, Coleoptera and Psocoptera but undesirable for Lepidoptera and Diptera. A combination of electricity and a safe insecticide, such as a pyrethroid, might be the best combination. Because of its use in one of the most important insect surveys done to date (the Rothamsted Insect Survey: Taylor and French 1974) the Rothamsted light trap (Williams 1948) is recommended as an appropriate design for arthropod biodiversity studies.

Light traps, although widely used, collect only certain, vagile taxa (many of which may be "tourists") which are active at night. Often they require nightly setup and servicing. Taxa collected include primarily Lepidoptera, Coleoptera, some Diptera, Neuroptera, Hymenoptera, and Hemiptera.

**•Emergence Traps and Tent Traps**

Traps that surround or cover a unit of habitat, and subsequently collect arthropods that move upwards towards the light, can collect a wide variety of insects directly associated with a specific habitat unit. While most studies using this kind of trap have dealt with specific taxa such as pest leafhoppers (Cherry *et al.* 1977), such devices are of potential value for biodiversity studies. Rosenberg *et al.* (1987) used a modified LeSage and Harrison aquatic emergence trap (LeSage and Harrison 1979) to sample arthropods in an Ontario peatland. This proved efficient for the collection of Chironomidae, but, for instance, took only 12 species of Sphaeroceridae. Pan traps in similar peatlands would be expected to take 40-50 species of Sphaeroceridae (Marshall 1994). Emergence or tent traps collect only positively phototactic arthropods present on or in the habitat at the time of installation of the trap, which can be an advantage or disadvantage depending on the goals of the project. These methods collect only resident species and can take large numbers of specimens. Adis and Schubart (1985) used tent traps ("ground photo-eclectors") to collect between 1,000 and 7,000 specimens (mostly Diptera) per square metre in a central Amazonian forest, compared to between 30 and 60 specimens per square metre collected in the forest canopy.

We recommend these types of traps for specialized studies where the fauna emerging from a particular type of habitat or vegetation is being analyzed. Emergence traps can be placed over a portion of a tree, for instance, to survey bark beetles emerging from the tree trunk or limbs.

**•Sticky Traps**

Traps that take insects which come into contact with an adhesive surface are widely used for sampling pest taxa such as stable flies (Williams 1973), mosquitoes (Dow and Morris 1972), and aphids (A'Brook 1973). Their efficiency is affected by colour, size,

orientation, shape, environmental conditions, and type of adhesive. Grease-based adhesives are more easily dissolved than resin-based adhesives, but only weak insects are trapped effectively by grease (Southwood 1978). Even with the easily dissolved grease-based adhesives, specimens are difficult to handle and the quality of the specimens collected often is well below the level required for specific identification of many taxa. We feel that flight-intercept traps are better alternatives to sticky traps when identification of a variety of taxa is an important component of a project. We do not recommend the general use of sticky traps in biodiversity studies.

## SAMPLING METHODS: ACTIVE COLLECTION TECHNIQUES

### •Substrate Sampling

Extraction of invertebrates from substrate samples can yield large numbers of wingless arthropods. Of special interest and value is the frequent capture and thus association of conspecific adults and immature stages. Despite this attractive feature, identification of immature stages to the species level is difficult and in many instances it is not possible. Substrate sampling is mainly of importance for surveying wingless taxa such as Collembola, mites, some groups of spiders and some ground-substrate-dwelling groups of beetles which generally will not be collected through other means. Substrate sampling is an excellent complement to Malaise/flight intercept/pan trapping. It should be noted that substrate sampling effectively samples resident species and very few "tourists" are represented. The main methods of substrate sampling are collecting by hand, behavioural extractors, and soil washing and flotation (see Górny and Grün 1993). Behavioural extractors and soil washing and flotation require active field collection of samples followed by passive processing to separate the arthropods from the substrate.

#### *Collecting by Hand*

The simplest method for sampling the biodiversity of large litter and soil arthropods such as millipedes, centipedes and some larger active beetles, is to mark a quadrat of 0.5-1.0 $m^2$ (or some other area) in the field and remove the litter and soil progressively, usually to a depth of 10 cm. The litter and soil can be processed onto a sheet or board through a range of sifters which are used to break up the debris and separate the arthropods from it. The arthropods present are simply collected as they move. Organisms which are not active, even if large, may be incompletely sampled using this method. This process depends on specimen movement and visual observation by the collector of that movement and results may be highly subjective and dependent on the observer. An alternative would be to employ field quadrats to collect the sample but use a behavioural extractor to remove the organisms.

Although offering a reliable means of standardization by sampling a defined area of substrate, the method is labour-intensive and experience shows that randomly selected quadrats yield low numbers of specimens and low species diversity per unit

of sampling effort regardless of how specimens are extracted. We do not recommend its use for general biodiversity surveys but we acknowledge that it may be useful for confirmation of microhabitat use of various taxa.

*Behavioural Extractors (Berlese and Tullgren Funnels)*

Berlese and Tullgren extractors are methods of separating arthropods from soil and litter, which generally involve using heat and desiccation to stimulate the animals to leave the samples on their own (Martin 1977). As a result only active, free-living stages are extracted. These extractors and their many modifications are the most practical and widely used methods of assessing the diversity and abundance of smaller, less mobile, cryptic arthropods in soil and litter. They can also be used successfully to collect arthropods from loose bark, rotting wood, bracket fungi, mosses, flowers, manure and nests (Martin 1977). The sample unit can be a core or cube, ranging from 2.5 cm to 1 metre in diameter, or can be a specific volume of substrate (Wright and Coleman 1988). In a survey of 20 recent publications on soil arthropods the most common core size used was 5 cm in diameter, to a depth of 15 cm in soil. However, the core size used will depend on the size of arthropods being collected and the habitat (Edwards 1991). In peatland soils, for example, Borcard's (1991) sample size was 15 cm x 15 cm to a depth of 10 cm. Samples may be collected randomly, or they may be collected using field quadrats.

Berlese and Tullgren funnels and their many modifications are the most practical and widely used methods of assessing the diversity and abundance of smaller, less mobile, cryptic arthropods in soil and litter.

For inventory purposes a quantifiable volume of substrate is not essential and the size of the sample unit may depend on the type of microhabitat being assessed. In these instances, the microhabitat can be concentrated by passing the litter through a sifter (Norton and Kethley 1988). As these authors note, this eliminates bulky, larger pieces of substrate and enhances uniform drying of the siftings.

The essential components of these extractors (see Martin 1977; Steyskal *et al.* 1986) are 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 70-80% ethanol with 5% acetic acid. A source of heat and desiccation (light bulb, electric resistance wire, or if necessary, sunlight) is placed above the sample. The objective is to create a steep gradient of temperature and moisture throughout the sample (Edwards 1991). The arthropods react to the heat and desiccation by moving downward (away from the heat) and eventually fall through the screen at the bottom into the preservative (Martin 1977). Normally, soil cores should not be deeper than 5 cm and should be inverted when placed in the sample containers. Chemicals (e.g. napthalene) may be used in place of the heat source, but generally these are not very effective. Cheesecloth below the sample and/or a baffle in the funnel can be used to reduce debris falling out of the

sample as it dries or is agitated by the movement of larger organisms. The wattage of the light bulb used depends on the size of the funnel (Martin 1977). The length of time of extraction varies from 6 hours to 1-2 weeks and depends on the moisture content of the sample, the intensity of the heat source, the depth and uniformity of the sample and the types of arthropods desired. Mites, for example, and especially immatures, may take a relatively long period of time (up to 14 days) to exit the sample. When funnels must be operated outdoors a tight-fitting hood is recommended to avoid contamination of samples by insects attracted to the light (Martin 1977). Although such species can often be identified as contaminants because their natural history is known, contamination can also result from specimens hanging on to, or becoming entangled in, the cheesecloth. To avoid this possibility, the cheesecloth and wire mesh should be removed after the processing of each sample and such specimens removed.

Commonly used modifications of the Berlese-Tullgren funnel include the Macfadyen high-gradient funnel, the Kempson apparatus (Kempson *et al.* 1963), and the Merchant-Crossley extractor (Merchant and Crossley 1970; Norton 1986). Norton and Kethley (1988) describe a light-weight, collapsible and easily transportable Berlese funnel made of rip-stop nylon, which is both simple to construct and efficient. Another design is presented by Wheeler and McHugh (1987). Edwards (1991) reviews most modifications of behavioural extractors and provides details of preferred methods for sampling different soil types.

We recommend the use of a sifter to concentrate the substrate. Five-litre samples of sifted litter, up to 10 per site per sampling period, should be collected. In inventory studies, a uniform effort should be made to sift in various microhabitats (e.g. under fallen logs, under fungi, under fruit- or seed-fall, etc.). For litter layer arthropods, such as beetles, a defined area of litter can be sifted for a defined time period.

For a given site, samples should be taken 4 times per year to ensure that active stages of species quiescent during some parts of the year are represented. All storage and transport of samples should be in containers which do not permit the buildup of excessively high temperatures or high humidity. We suggest that each sample be placed in a lightweight cotton or ripstop nylon bag; pillow cases are ideal.

We recommend that funnels be constructed of lightweight sheet metal or aluminum flashing. Funnels should be 50 cm (20") high and 35 cm (14") in diameter at the top and tapered to 2.5-5 cm (1-2") in diameter at the bottom. Hardware cloth of 1.25 cm (1/2") mesh should be placed in the funnel 15 cm (6") from the top and covered with single-ply cheesecloth. Samples should be processed for between 8 hours and 14 days using 60W bulbs suspended 10 cm (4") above the top of the sample.

### *Soil Washing and Flotation*

Behavioural extraction methods extract only the active stages of arthropods, and in arid soils, deep soils, and mineral soils with high clay content are inefficient for certain groups, such as endeostigmatic mites and podurid and onychiurid Collembola (Walter *et al.* 1987). For any inventory in these habitats, and for these groups, soil washing

and flotation (Kethley 1991), or direct heptane flotation of soil cores (Walter *et al.* 1987) is recommended. Soil washing often is a difficult technique to use because it requires large quantities of water (about a 4:1 ratio of water to soil), but as Kethley (1991) notes this technique provides complete life-history data for many microarthropods and is the only effective way to assess diversity in deep soils where arthropod numbers are very low. The recent development of heptane flotation (Walter *et al.* 1987; Kethley 1991) is based on the affinity of the arthropod cuticle for petroleum derivatives such as heptane. This method is useful in some circumstances as it allows large numbers of soil samples to be stored for some time and processed when convenient. In contrast, processing using behavioural extractors should be carried out as soon as possible after field collection of the samples (Edwards 1991).

**•Vacuum Sampling**

Projects that attempt to approximate complete inventories of a given habitat or locality, or projects that require an estimate of the degree to which a trapping programme is sampling the total fauna, require techniques that collect virtually all the arthropods in a given unit of habitat. Suction devices such as the widely used "D-vac" (Dietrick 1961) are often considered to obtain total faunal samples, but differ in efficiency for different taxa and for different substrates. Theoretically, a very high power suction sampler such as the motor-vehicle-sized "McCoy Insect Collector" (McCoy and Lloyd 1975) can take almost 100% of the fauna, unfortunately along with a great deal of substrate. Vacuum samples which include litter, leaves, and other debris must be laboriously sorted by hand unless some kind of behavioural extractor is used. Behavioural extraction necessitates that the specimens be kept alive, and even so only a sample of the arthropods in the collection would be extracted. Vacuum sampling might take some species missed by passive trapping programmes, but should not be considered as a means of measuring absolute species richness.

**•Chemical Knockdown**

Another approach to total inventory is to make collections by using chemicals to kill or stun everything in a unit area of habitat. This method has been used to sample fruit-tree insects (Collyer 1951) and is now used for biodiversity studies in the tropical forest canopy. The recent (and controversial) estimates of total global biodiversity (Erwin 1982; Stork 1988) are based largely on chemical knockdown of insects from tropical trees. The target area is sprayed or fogged, usually with a pyrethroid insecticide, and the affected organisms fall onto sheets, trays, or funnels placed below. A method for canopy spraying is described by Martin (1966), the merits of canopy fogging are discussed by Paarmann and Stork (1987) and Adis *et al.* (1984) and a summary of the protocol and methodology underlying insecticide sampling in trees is given by Stork (1988).

Knockdown methods are selective for larger insects that do not stick to the foliage, and can undercollect smaller insects by as much as 50% (Muir and Gambrill 1960).

Much of the fauna living under bark or in leaves is not sampled. Despite these concerns, this technique remains the most popular and effective approach to the study of canopy arthropod biodiversity. Chemical methods do have the advantage of being relatively independent of insect activity and climatic conditions, and can be applied to specific microhabitats such as individual trees, specific parts of trees, or a specific volume of canopy. Canopy fogging in a tropical forest yields thousands of specimens in a matter of a few hours. Stork (1988) reports 24,000 individuals representing over 2,800 species from only 10 trees in Borneo. In order to take seasonality into account fogging samples should be taken at various intervals throughout the year. Immature stages are rarely collected using this technique unless they feed externally on foliage.

Canopy fogging was originally developed for study of temperate forests; however, it is most effective in tropical forests where the forest canopy is complex. Most foliage in a tropical forest is in the canopy and there is high plant-species diversity and an abundance of epiphytes. This technique is somewhat expensive and complex to set up and to use effectively. It would be useful for studying the insects of the forest canopy in particular, but we do not recommend the use of this method in most studies of biodiversity, particularly at temperate latitudes.

### •Suction and Rotary Traps

In contrast to Malaise traps, which passively sample flying insects, suction and rotary traps actively sample the insects in a given volume of air. They do this either by pumping a volume of air through a filter (Johnson 1950) or by using a mechanically rotated net that continuously samples aerial fauna (Chamberlin 1940). Taylor (1962) suggests that these traps sample 85% of the flying population. Suction traps collect slowly and weakly flying insects which form a kind of aerial plankton. Certain elements of the fauna are easily collected in this manner. Most studies using these types of traps deal only with specific pest species (Taylor 1962), or list general collections identified only to order (Nichols 1960).

We consider that suction traps constitute an effective alternative method for selected faunal inventory projects. Suction traps collect a number of small, fragile, winged insects which are otherwise not sampled. Among the groups taken in suction traps are micro-Coleoptera, micro-Hymenoptera, Coniopterygidae (Neuroptera), and alate aphids (Homoptera). One drawback of both suction and rotary traps is that an electrical power outlet must be near the habitat. We do not recommend the general use of rotary traps.

### •Sweeping and Beating Vegetation

The use of an insect net is the most commonly and widely known technique for collecting insects. As a routine sampling method, the use of a net is appropriate in some habitats, but only under uniform conditions. Weather, vegetation type and age, weight of net, type of mesh, and handler skill are some of the factors affecting net collections. Those relatively few taxa which sit high on the vegetation and do not fall off when

approached might be efficiently sampled, and some of them may include species rarely collected in Malaise or pan traps. A measure of numbers of sweeps can be used for standardization, but because of high user bias this method is recommended only as an auxiliary sampling technique in an inventory project. If sweep netting is used we suggest that the standard dimensions of the net be 38 cm and that 20 sweeps of 180° constitute a sample.

Another commonly used method of general collecting is to place a sheet under a plant and to strike the vegetation so that the arthropods on the plant are dislodged and fall onto the sheet. Such beating sheets generally are supported by a framework of interlocking lightweight poles and are held under the plant with one hand while the plant is struck with a stick held in the other hand. As with the use of nets, standardization is possible but subject to bias; beating sheets are recommended only as an auxiliary sampling technique in an inventory project. If beating is used we suggest that the standard dimensions of the sheet be 1 $m^2$ and that 20 beats per tree or sample be made.

**•Specialized Collecting Techniques**

Specialists in every group of arthropods prefer particular, often specialized, methods of collecting. Specialized techniques can be used to supplement baseline inventory data gathered using the sampling methods discussed above, or they can be used for biodiversity measures using limited collections of particular taxa. Coddington *et al.* (1992) argue that "hit and run" sampling trips, or single-visit collecting trips, are the only practical way to sample tropical biodiversity. Given this premise, they offer an assessment of "looking up", "looking down", beating, and sifting as quantifiable methods of sampling spiders. This kind of spot assessment of selected taxa using selected techniques will probably remain the only practical approach to biodiversity assessment for much of the tropics, and offers many efficiencies over exhaustive inventories. For example, dealing with large numbers of common species is a major cost of processing mass samples. Specialized collecting usually allows for the rejection of common species after a certain number have been collected, thus lowering the cost of processing the resultant collections.

The main role of specialized collecting in broader biodiversity studies is to supplement, and assess the efficiency of, mass sampling devices such as pan traps and behavioural extractors. Specialists on phytophagous taxa are almost certain to find additional species by beating specific plants, searching appropriate hosts, and by rearing from hosts or plant parts. Similarly, specialists are able to recognise microhabitats from which they can net, aspirate or otherwise hand collect rare species which might be missed by other techniques. Members of the acarine suborder Oribatida, for example, are commonly associated with soil and litter, but any inventory of this group should include specialized collecting such as twig washing, leaf washing, observation of leaf surfaces, and collections from the axils of twigs and branches. It is therefore important for any inventory project to allow for site visits by specialists in the taxa under

consideration. Such visits can generate additional taxon records, and cast light on trapping efficiency and habitat heterogeneity from different perspectives.

## SAMPLE PREPARATION

The most time-consuming aspect of any typical biological inventory or biodiversity study is the conversion of "raw" samples into prepared, labelled and sorted lots to be identified in house or sent off for specific identification. It is essential that instructions for how to prepare specimens be solicited from cooperating systematists and that these be strictly adhered to. Specimens trapped in fluid usually must be dried in a critical-point drier (Gordh and Hall 1979), carefully mounted to the specifications of the cooperating systematist, labelled using appropriate data and paper (Darling and Plowright 1990), then sorted to "sendable units", usually family, and directed to the appropriate cooperating systematist. In some cases, material will have to be mounted on slides or sorted into alcohol. The importance of proper preparation cannot be overemphasized. For example, specific identification of 500 properly point-mounted, critical-point-dried flies might take a specialist about one week, a good investment of time if the data on the specimens rendered them useful. If the specimens were air dried, identification time might increase to four weeks and might render the investment of time impractical. If the submitted flies were not glued to points firmly enough to allow dissection without remounting, it could again double the time required for identification. Such specialized demands must be taken into account when preparing a budget for any inventory project. One hour a week spent emptying pan traps can easily keep a full-time technician busy preparing and sorting material. The ratio between sampling costs and processing costs can be as high as 1:40.

The ratio between sampling costs and processing costs can be as high as 1:40.

A well designed project is likely to result in the collection of tens of thousands of specimens, some of which will become valuable identified voucher specimens, and some of which will be unidentifiable at the present time, yet worthy of retention. It is important to consider the long term integrity of this material. As emphasized for the initial planning of a study (above), voucher specimens must be placed in museums with a long-term commitment to specimen maintenance. Unidentified material should be maintained with appropriate information for later incorporation into, or comparison with, the project database. Consideration of the cost of storage materials such as pins, unit trays, drawers, insect cabinets, and subsequent curatorial activity therefore should be an integral part of any inventory budget.

## DATA MANAGEMENT

It is important to establish appropriate databases to allow for maintenance and exchange of data files on habitats and specimens. Computerization of data, including unique specimen codes, can greatly facilitate database usage and future additions to the database. There is a need for systems that allow specimens to be readily tracked and

related back to a particular site and date. Janzen (1991) has pioneered the use of bar-coded specimen labels for this purpose. The use of bar codes can be costly and may be best suited for inventories in which species diversity is high and the number of individuals of many of the species collected is low. Standard use of precise latitude and longitude data for sample sites, notably by using Geographical Positioning System (GPS) capabilities, is highly desirable.

An extensive and developing literature considers requirements for specimen databases. For example, standards for fields and terms for collection data for insects have been proposed by Noonan (1990) and Noonan and Thayer (1990). These aspects of reporting and tracking information on specimens collected during biodiversity inventories are important for the long-term value of any study, but details of such requirements are beyond the scope of this brief.

## CONCLUDING REMARKS

It has been estimated that at present only about half of the insect fauna of Canada is known. Regional inventories, done properly and supported by an adequate systematics infrastructure, can work towards resolving this lack of knowledge while at the same time providing the baseline data needed for proper and efficient management of biodiversity. Such inventories are expensive and time consuming to carry out, because the numbers of arthropod species and specimens collected greatly exceed the numbers resulting from surveys of other taxa.

Nevertheless, surveys of arthropod biodiversity are feasible, given proper planning and suitable sampling protocols, as explained in this brief. We re-emphasize that the limiting factor in any inventory of arthropod diversity is the strength of the systematics resources available to support, and in many cases carry out, this kind of work. Consequently, much of the biodiversity in our own backyard will remain unknown unless the numbers of systematists and the support for this science increase.

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Published by
The Entomological Society of Canada

Supplement to the
*Bulletin of the Entomological Society of Canada,* Vol. 26(1)
March, 1994