

TERRESTRIAL INVERTEBRATES

Colin C. D. Tingle¹

Natural Resources Institute, University of Greenwich at Medway, Central Avenue,
Chatham Maritime, Kent ME4 4TB, UK

INTRODUCTION

Invertebrates occupy a large array of ecological niches within the terrestrial environment. They are a highly successful group of animals, which makes many important contributions to the functioning of the living world. Some are involved in the decomposition process leading to the recycling of nutrients; some with pollination of flowering plants; many are herbivorous and these have a major impact on plant biomass and survival; whilst others play important roles through the regulation of animal populations, as parasites or predators. In turn, invertebrates provide an important food source for many amphibians and reptiles, birds and some mammals (see chapters 11, 12 and 13). Some invertebrates (particularly insects) are highly mobile and only transient occupants of a particular habitat or area, whilst others may be sedentary, with small home ranges and key roles in the ecology of that range. Many invertebrates show strong seasonality in their occurrence and abundance and even great variation in their activity on a daily basis. Because of this ecological diversity, different techniques are needed to sample those living in different habitats and within different strata of the same habitat. No single method of sampling will be efficient at capturing the entire range of invertebrate fauna within a given area. Thus, methods must be chosen which target the taxa of interest or, if general collections are required, then the use of a number of different techniques simultaneously may be required.

Terrestrial invertebrates are often directly exposed to pesticides, as they live in a number of habitats which are deliberately sprayed to control insect pests, fungi or weeds or to protect human beings from disease vectors. Others are directly exposed by deposition of insecticide sprays which miss their target, e.g. soil-dwelling invertebrates in sprayed forests. In either case, exposure may be through contact or ingestion. Other invertebrates may be indirectly affected through the removal or reduction of food sources, be they vegetable, fungal or animal. Insecticides are designed specifically to kill insects and thus most invertebrates are sensitive to these chemicals. Sensitivity to other pesticides varies, but some herbicides and fungicides are also directly and highly toxic to this group of organisms.

The assessment of pesticide impact on terrestrial invertebrates generally relies on some quantification of population levels, relative abundance and/or species composition in sprayed areas and a statistical comparison with the same criteria in unsprayed areas. In some cases the collection of invertebrates for residue analysis can be helpful and direct mortality assessment (cadaver counts) can also be useful in determining the effects of some insecticides. To be done thoroughly, the scientific assessment requires a substantial input of time and resources. The reliability of the results of any study on pesticide impacts will be greater, the more that is known about the ecology of the area to be treated. Thus, where possible, trials should be sited in areas where data have already been accumulated on invertebrate abundance, and/or species composition, diversity and the role of invertebrates in the ecosystem and its functioning.² In practice, this will rarely occur as the biology and ecology of the invertebrate fauna of many tropical habitats are often poorly known or completely unstudied. Thus some study

¹Contact address: 9 Norman Avenue, Henley-on-Thames, Oxon. RG9 1SG UK. tc09@gn.apc.org/colin.tingle@thenrgroup.net

²Data on previous use of pesticides or other contaminants in the area should also be sought and documented so that results are correctly interpreted.

of the ecology of the site will usually need to be carried out in conjunction with the ecotoxicological assessment. Invertebrates are particularly prone to large natural variation in abundance both in time and space. This makes them one of the most difficult groups of animals to study quantitatively in the field. Long-term (3–5 years) or repeated short-term (minimum 2–3 months each year for 3–5 years) studies are preferable, because the results of a single, short-term study will be difficult to interpret.

The aim of this chapter is to describe some of those sampling methods for invertebrates which are useful in the assessment of pesticide impact work and to provide guidelines for the selection of appropriate techniques for use in different situations.

STUDY DESIGN

Pesticide impact on non-target invertebrates is best assessed using a replicated experimental design (see chapter 2). Details are given by Southerton *et al.* (1988) for designs used in agro-ecosystems. However, large-scale applications of pesticides often take place in non-crop situations, and pesticide impact studies then have to be done as monitoring exercises. In these situations, true replication is impossible. Drawing inferences about cause and effect is much more difficult in this type of situation (Eberhardt and Thomas, 1991) and real care is necessary to avoid collecting data which will prove impossible to analyse and interpret (see chapter 2).

However the study is structured, be it a standard, replicated experiment or a monitoring exercise, several basic features should be incorporated into the design.

- At least 1 month's worth of pre-spray data are required on invertebrate abundance in the trial site and a year's data would be preferable.
- Invertebrate abundance often follows seasonal patterns. Thus a comparison of relative abundance of a given invertebrate taxon between different months may be highly misleading. Similarly, year to year variation of some invertebrates can be very high. Account should be taken of this when designing the study to ensure meaningful results.
- Part (or parts) of the study area should be left unsprayed throughout the study, as a control.
- Plot size must be appropriate for the scale of pesticide applications and activity of invertebrate groups of interest. For example, darkling beetles (Tenebrionidae) may cover distances of 400 m during foraging excursions and thus require very large plot sizes; whereas some springtails (Collembola: Isotomidae) may cover only 5 m. **Note:** There are some situations in which discriminative spray applications are used, e.g. ground-spraying against tsetse fly or barrier treatment against locust, where reinvasion is an inevitable process and an important factor in judging the impact of spraying.
- Pesticide-treated and untreated sites or plots should be sufficiently distant to prevent unintended contamination of the untreated area and to prevent invasion of the treated area by fauna from the untreated area which could confound the results.
- Replication or (where unavoidable) pseudoreplication (see chapter 2) should be adequate for statistical tests to have the power to demonstrate effects above the level of natural variation for the study area. Wherever possible, the use of several unsprayed sites for comparison with the spray impacted site will improve the reliability of findings (Underwood, 1994).
- Selection of sample sites for a monitoring exercise must use a stratified system (see chapter 2), leading to 'matching' of sites across treatment areas, unless work is carried out in a homogeneous study area or unless heterogeneity is so widespread that different sample sites represent a similar diversity of habitat types.

- As much data about the environmental conditions of the study area and individual sample sites should be recorded as possible, e.g. temperature, humidity, soil type, vegetation, distance from field boundaries or other habitat features, etc.
- People living in or near areas selected as study sites for pesticide impact assessment should always be consulted at an early stage. If trials are within farmers fields, then this will inevitably happen, however, if study sites are natural or semi-natural savanna, woodland or forest areas, local people can still provide an enormous wealth of information and should be consulted and involved in the process of setting up the sampling programme. Equipment used for marking out study sites and for sampling invertebrates may be attractive to local people. If they have not been informed of trials, equipment may be moved, stolen or damaged. Consultation and involvement of people living in or using the study area will minimize these types of problems and lead to lower costs, less frustration and more reliable results.
- Design of the study must be in proportion to the resources available. Thus if staff numbers are limiting some methods of sampling for invertebrates (see 'Sampling Techniques' for details, page 165) may be inappropriate. Similarly, if transport is limiting, this will have to be taken into account in determining the practical feasibility of using certain sampling methods.

Design of the study and selection of sampling methods will be affected by the type of pesticide under investigation, the ecosystem in which it is to be applied, the application method and the taxonomic ability of the staff involved or outside expertise available. Table 8.1 is a generalized scheme which gives guidance in selecting sampling methods appropriate to particular situations. More detailed information to aid in the selection process is given below.

Which pesticide?

Different classes of pesticides have different modes of action and each individual pesticide affects different fauna to a differing extent or not at all. The characteristics of the individual pesticide will be important in determining the methods used in evaluating environmental impact. Wherever possible, specific information relating to the chemical should be used in deciding the scope of the environmental monitoring. The overview below provides a synopsis of the main features of the major pesticide groups, which may help in an initial selection of sampling methods for invertebrates appropriate to particular situations. Assuming recommended dose rates are followed, the broad groupings of pesticides give clues to the type of sampling which will be necessary.

Organochlorines	Measurement of residue levels in selected invertebrates (particularly those important as vertebrate prey) from sprayed and unsprayed areas. Species composition of non-target groups and similarity between sprayed and unsprayed sites (e.g. Sorensen's Index (QS)). Diversity and species richness (see page 00). Relative abundance of indicator groups, particularly predatory mites (Acari: Mesostigmata), springtails (Collembola: Isotomidae) and parasitic wasps (Hymenoptera).
Organophosphates	Acute effects on bees (Hymenoptera: Apoidea) and effects on production of new queens. Relative abundance of bees and wasps (Hymenoptera), certain beetles (Coleoptera: ground beetles [Carabidae], soldier beetles [Cantharidae] and ladybirds [Coccinellidae]), jumping spiders (Araneae: Salticidae), Collembola and predatory mites (Acari: Prostigmata, Mesostigmata). Species composition and diversity of faunal assemblages (particularly spiders [Araneae]).

Table 8.1 Matrix for determination of suitable sampling techniques

Sampling method	Non-target fauna of interest										Habitat										Pesticide										Application method						Technical knowledge	
	Bees	FI	VD	AI	EI	SI	IR	FC	P/S	W/F	O/P	WL	OC	OP	Ca	Py	IGR	Bio	H	F	PP	Kn	Tr	ULV	Fog	Aer	G/Sd	B	D/PO	Ent	Non Tax							
Pitfall traps					●	○		●	●	●	●		●	●	●	●	●	●	●	●	●	●	●	○	○	○	○	○	○	○	○	○						
Quadrats			○		●	○		●	●	●	●	●	●	●	●	●	●	●	●	●	●	●	●	●	○	○	○	○	○	○	○	○						
Cryptozoa boards					●	○		●	●	●	●	○	●	●	●	●	●	●	●	●	●	●	●	○	○	○	○	○	○	○	○	○						
Food baits	○	○		●	●	○		●	●	●	●	●	●	●	●	●	●	●	●	●	●	●	○	○	○	○	○	○	○	○	○	○						
D-Vac/ suction sampler	○	○		○	●			●	●	●	●	●	●	●	●	●	●	●	●	●	●	○	○	○	○	○	○	○	○	○	○	○						
Sweep net	○	○		○	●			●	●	●	●	●	●	●	●	●	●	●	●	●	●	○	○	○	○	○	○	○	○	○	○	○						
Malaise trap	○	○		○	●			●	●	●	●	●	●	●	●	●	●	●	●	●	●	○	○	○	○	○	○	○	○	○	○	○						
Yellow water trap	○	○		○	●			●	●	●	●	●	●	●	●	●	●	●	●	●	●	○	○	○	○	○	○	○	○	○	○	○						
Light trap					●			●	●	●	●	●	●	●	●	●	●	●	●	●	●	○	○	○	○	○	○	○	○	○	○	○						
Hive counts/observation	●							●	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○						
Lure/pheromone traps				●	●			○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○					
Beating				○	○			○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○					
Funnel traps	○	○		○	●			○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○					
Sheet traps				●	●			○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○					
Trunk traps				●	○			○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○					
Soil cores						●		○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○					
Litter bags					●			○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○					
Monolith samples					●			○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○					
Formalin drench					●			○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○					
Direct collection				○	○			○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○					
Termite colony health					●			○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○	○				

FI = Flying insects
 VD = Vegetation dwellers
 AI = Arboreal invertebrates
 EI = Epigeal invertebrates
 SI = Soil invertebrates
 IR = Insecticide residues

FC = Field crops
 P/S = Pasture/savanna
 W/F = Woodland/forest
 O/P = Orchards/plantations
 WL = Wetlands

OC = Organochlorine
 OP = Organophosphate
 Ca = Carbamate
 Py = Pyrethroid
 IGR = Insect growth regulator
 Bio = Biologicals (e.g. Bt)
 H = Herbicide
 F = Fungicide
 PP = Phenyl pyrazole

Kn = Knapsack sprayer
 Tr = Tractor/vehicle-mounted sprayer
 ULV = Ultra-low-volume
 Fog = Fogging
 Aer = Aerial application
 G/Sd = Granules/seed dressing
 B = Baits
 D/PO = Dips/pour-ons

Ent = Entomologist
 Non = Non-specialist
 (with some biological training)
 Tax = Taxonomic assistance required

● Appropriate method
 ○ Less suitable but can still give useful results
 □ Not applicable

Carbamates	Acute toxicity to bees. Relative abundance of ants (Formicidae) and other Hymenoptera, predatory mites (Acari: Prostigmata) and ground beetles (Carabidae). Species composition and diversity.
Pyrethroids	Relative abundance of spiders (Araneae) (particularly money spiders [Linyphiidae]), parasitic Hymenoptera, silverfish (Thysanura), leaf beetles (Chrysomelidae) and Formicidae. Species composition and diversity.
Phenyl pyrazoles	Termites (Isoptera) via assessment of colony health and/or termite activity. Acute toxicity to bees. Relative abundance of Acari, Araneae, earwigs (Dermaptera), certain grasshoppers, crickets and relatives (Orthoptera), Coleoptera (certain Carabidae, certain weevils [Curculionidae], certain Tenebrionidae), robber flies, big headed and other flies (Diptera [Asilidae, Pipunculidae, Muscidae]), Hymenoptera (Apoidea, Chalcidoidea, Scelionidae, Sphecidae, Tiphiidae, Braconidae, Formicidae). Diversity and species composition.
Insect growth regulators	Relative abundance of orb web, lynx and jumping spiders (Araneidae, certain Oxyopidae, certain Salticidae) and predatory mites (Acari), Orthoptera, lacewings, ant lions and their relatives (Neuroptera), Coleoptera (Tenebrionidae, Curculionidae, Chrysomelidae, Coccinellidae), butterflies and moths (Lepidoptera) and Hymenoptera (Braconidae). Species composition, faunal similarity (QS), diversity (particularly mandibulate herbivores) and species richness (Araneae).
Biologicals	Relative abundance, diversity and species composition (particularly macrolepidoptera, Orthoptera and parasitic Hymenoptera).
Herbicides	Relative abundance of Collembola, Acari, earthworms (Annelida: Oligochaeta), nematodes (Nematoda), honeybees (Apidae) and Carabidae. Secondary effects on soil fauna (see below) and vegetation-dwelling fauna (see below) caused by reductions in vegetation.
Fungicides	Relative abundance of parasitic Hymenoptera (particularly Chalcidoidea), predatory bugs (Hemiptera), mesostigmatic and other Acari. Also Annelida (earthworms and pot worms [Enchytraeidae]).

Where is it used?

The composition of invertebrate faunal assemblages varies with biome, thus different non-target invertebrates may be affected by the application of the same pesticide in different habitat types. As a broad guideline, the following key invertebrate groups are likely to be at risk when the following types of habitat are subjected to pesticide application.

Agro-ecosystem	Beneficial invertebrates – bees, parasitic Hymenoptera and Diptera, predatory Coleoptera, predatory Diptera and Neuroptera, predatory Acari and Araneae. Detritivores and recyclers – Annelida, millipedes (Diplopoda), Acari, Coleoptera and Diptera.
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Woodland/forest	Invertebrate faunal diversity. Detritivores and recyclers – Annelida, woodlice (Isopoda), Diplopoda, Coleoptera, cockroaches (Blattodea), Isoptera and Formicidae. Pollinators – Diptera, Hymenoptera. Invertebrates important as food for higher animals – Lepidoptera, Formicidae and Isoptera.
Pasture/savanna	Diversity. Primary consumers – Orthoptera, Coleoptera, Lepidoptera. Detritivores – Isoptera, dung beetles (Scarabaeidae) and Formicidae.
Orchards/plantations	Pollinators – Diptera, Hymenoptera (bees in particular). Beneficial invertebrates – parasitic Hymenoptera and Diptera, predatory Coleoptera, predatory Diptera and Neuroptera, predatory Acari and Araneae.

Application method

The method of pesticide application can also have a major influence on the fauna affected (due to differences in formulation, drop size, drift and pesticide fate) and thus on the sampling methods needed to appraise effects.

High-volume from knapsack or tractor	Fauna on ground cover vegetation (Araneae, Acari, praying mantids [Mantodea], Orthoptera, book and bark lice [Psocoptera], Hemiptera, Thrips [Thysanoptera], Neuroptera, Coleoptera, Diptera, Lepidoptera [larvae] and Hymenoptera); soil surface (Diplopoda, centipedes [Chilopoda], Pauropoda, Araneae, Acari, harvestmen [Opiliones], pseudoscorpions [Chelonethi]; sun spiders [Solifugae]; scorpions [Scorpiones], false scorpions [Amblypygi], Thysanura, Collembola, Blattodea; Dermaptera; Hemiptera; Isoptera; web spinners [Embiidina]; Orthoptera, Coleoptera and Hymenoptera); and within the soil (Annelida, Nematoda, Isopoda, Diplopoda, Chilopoda, Symphyla, Acari, Chelonethi, Collembola, Hemiptera, Isoptera, Embiidina, Coleoptera, Diptera and Hymenoptera).
Ultra-low-volume (ULV)	Fauna associated with low growing but upright vegetation (see above) or, if aerially applied, with the vegetation canopy (those listed above as vegetation-dwelling plus stick insects [Phasmatodea]), arboreal invertebrates (particularly Araneae, Acari, Chelonethi, Collembola, Psocoptera, Blattodea, Mantodea, Orthoptera, Hemiptera, Thysanoptera, Coleoptera, Diptera, Lepidoptera and Hymenoptera), vegetation-dwelling invertebrates and flying insects (Orthoptera, Thysanoptera, Neuroptera, Coleoptera, Diptera, Lepidoptera and Hymenoptera). Epigeal fauna only if little or no vegetation cover.
Fogging	Canopy invertebrates, flying insects, arboreal invertebrates and (to a lesser extent) epigeal fauna.
Aerial	See fogging/ULV.
Granules/seed dressing	Epigeal and soil-dwelling invertebrates.
Baits	Epigeal invertebrates and scavengers (e.g. Formicidae).
Pour-ons	Biting flies (Diptera [Tabanidae, Hypoboscidae, etc.]); detritivorous Coleoptera (Scarabaeoidea, Tenebrionidae) and Diptera (Muscidae); Isoptera (particularly Termitidae); and dung-dwelling Coleoptera (Histeridae) and Diptera (particularly larvae).
Dips	Diptera (Tabanidae, Hypoboscidae, etc.).

Technical expertise

Many of the sampling techniques described below will catch a wide variety of insects or other invertebrates. The taxonomic expertise of the staff involved in the work will govern how much information can be gained from the samples. Almost any work on pesticide impacts on invertebrates will involve some basic taxonomy. Generally, individual species will be affected differently by a given pesticide and thus adverse impacts will often only be detectable if fauna are identified to species. Wherever possible an entomologist or invertebrate zoologist should be involved in the work. For the non-entomological biologist, many methods will allow assessments to be made of biomass, overall numbers and, possibly with the aid of a key, separation of the catch into orders. If further division is required, then fauna which look identical may be grouped as a 'morphospecies', given a number or a letter to distinguish them and counted separately. A reference collection should be established during sorting of samples, so that different groups are not confused and standard records are kept. Quick sketches and notes on major features will aid separation of different taxa found. Reference specimens may then be sent to specialist taxonomists for further identification. Such taxonomists can be contacted through your local or national natural history museum, biology department of your local university, via local or national wildlife groups, via the government's Wildlife and National Parks Department, Environment Department or Conservation Department. If no assistance can be found through any of these routes, then contact the Natural History Museum, London or, for southern African fauna, the Transvaal Museum, Department of Invertebrates (see <http://www.nfi.co.za/coleoptera/identfees.html>).

SAMPLING TECHNIQUES

EPIGEAL INVERTEBRATES

Pitfall trap

Pitfall traps provide a good technique for collecting data on the presence and absence and/or relative abundance of a wide range of surface active invertebrates. Animals fall into a container, set flush with the soil surface. With careful sorting and appropriate taxonomic evaluation, data can be collected on fauna ranging from microscopic mites to large scorpions and beetles. Pitfall traps are widely used, but do have limitations which must be taken into account when interpreting results (Adis, 1979).

Pitfall trapping is suitable for fieldwork in isolated areas, as a variety of containers can be adapted for use as traps (see below), provided the same size and type of container is used throughout a given study. Ideally, a standard pitfall trap should be used (Adis, 1979), but none has yet been agreed. The container should be placed in a sleeve, set permanently in the soil (see method sheet). This will minimize disturbance when emptying and resetting traps. At least 30 traps per treatment area (e.g. 30 in the sprayed area and 30 in the unsprayed area) will be necessary and their arrangement will depend on where they are used and the type of spray operation under study. However, they are generally best placed in a line or grid, with not less than 2 m between traps. The same preservative *must* be used throughout a given study; formalin is probably most readily available, but picric acid solution is the favoured choice.

Limitations Many factors influence the catch, e.g. climatic conditions, vegetation, ground-surface irregularities, trap diameter, shape and form of the trap, killing or preserving agents, whether the trap is covered or not, species selectivity, number and arrangement of traps, material from which the trap is made, time after traps are set, trampling around traps, etc., and great care is required in standardization of these factors in a given study. Pitfall trap catches actually measure 'activity abundance' and provide no absolute measure of population.

Processing Trap contents should be strained from the formalin or other preservative and poured into a petri dish (or similar). Invertebrates should be sorted from debris using forceps, a paintbrush, pipettes, etc. Use a magnifying glass or binocular microscope to sort smaller invertebrates, if available.

Resulting data Numbers of individuals can be sorted into species or morphospecies and counted to give data on relative abundance and faunal composition and/or diversity. Catches can be weighed³ or measured (Rogers *et al.*, 1977) to calculate biomass.

Fauna sampled Most epigeal invertebrates, but certain carabid beetles are particularly susceptible, whilst other species avoid these traps or easily escape. They are also not suitable for trapping certain types of spiders.

Sampling period Traps may be emptied daily, weekly or monthly.

Equipment Traps can easily be made using locally available materials, e.g. jam jars, yoghurt pots, plastic cups or plastic milk bottles. Where possible, traps should be glass or plastic, 6 cm diameter and not less than 12 cm deep. Traps, marker flags and trap covers should all be made in advance.

Staff required 1 (preferably 2). More staff may be required to sort catches, depending on trap numbers; 3–5 staff ideal.

Food baits

Food baits can be used to collect data on relative abundance or activity of a number of invertebrate groups. Suitable foodstuffs or other attractants are left out in appropriate places and monitored regularly to count and identify the fauna attracted (Southwood, 1966). Depending on the objectives of the study, a large range of data can be collected, e.g. time taken to find baits, rate of bait removal, numbers of individuals attending baits, number of species at baits, etc. Baits and traps containing them tend to be highly species-specific and details of a wide variety are given in Southwood (1966). Here, just two examples are given, for termites and for ants.

It is notoriously difficult to estimate populations of ants and termites, but some measure of their foraging activity (and thus colony health) can be made using food baits.

For termites, a variety of wooden or card baits may be used, depending on the termite species of interest and the duration of the trial (French and Robinson, 1981). The baits should be placed on the ground in a grid of at least 10 baits per site. There may be 5–10 sites per treatment area.

Limitations Baits provide a relative measure of activity abundance *only* and are influenced by many other factors, such as temperature, time of day, season, rainfall, soil type, vegetation, presence of other food sources, proximity to termite mounds or nests, etc.

Processing Baits should be examined *in situ*, lifted and replaced as originally set. Any termites feeding on baits should be counted and collected for identification.

Resulting data Several criteria can be noted: any termite runs coming into contact with baits can be taken as evidence of termite activity in the vicinity; attack on baits (i.e. evidence of feeding on the bait); damage to bait (e.g. proportion of area of bait eaten). Baits should be taken back to the laboratory and weighed at the end of the sampling period. Loss of weight can then be used as quantitative data.

Fauna sampled Termites.

Sampling period Baits should be left for several weeks before monitoring and then visited weekly (wet season) or monthly (dry season).

Equipment Locally available materials can be used as baits, e.g. toilet rolls, cardboard or soft-wood boards or pegs. These should be cut to size and weighed individually before they are needed. Marker flags should also be made up in advance.

Staff required 1 (preferably 2).

For ants, a variety of food baits can be used either singly or together, depending on the species of interest (Murphy and Croft, 1990; Tingle, 1993). Peanut butter, fish paste, breakfast cereal, grain, honey, and/or moribund insects can all be used. As with termite baits, grids of at least 10 baits per site should be used, preferably with five or more replicates or pseudoreplicates per treatment area. Baits should be covered with coarse mesh wire to prevent squirrels, birds and other animals from robbing them. If possible, records of numbers of ant nests and their distance from baits would aid interpretation of results.

³Dry weight is usually required for comparison with other biomass data, thus animals should be oven-dried to constant weight

Limitations The numbers and species of ant attracted will depend on the baits used, time of day, temperature, rainfall, season, vegetation, availability of other food and proximity of nests. The results provide an estimate of foraging activity and abundance of some ant species only.

Processing Count ants at baits and estimate quantity of remaining bait *in situ*.

Resulting data The number of species on baits, number of individuals of each species, percentage of bait remaining, time taken to find bait dishes.

Fauna sampled Ants.

Sampling period Baits are best visited on a regular basis, beginning 1 h after setting and continuing at regular intervals (e.g. 3 h, 6 h, 9 h, 24 h) for at least 1 day.

Equipment A marker flag and wire mesh covers for bait dishes should be made up in advance. Baits can be made up from locally available sources. Any dish may be used (but all treatment area dishes *must* be the same). Take transparent polythene and clothes pegs to the field to cover traps if raining.

Staff required 1 (2 preferable).

Other methods

Quadrats (Critchley *et al.*, 1980); Cryptozoa boards (Sutton, 1972); food baiting for flies (Stubbs and Chandler, 1978), cockroaches, crickets and beetles (Southwood, 1966); direct counts (Ausden, 1996). See also tethered litterbags below. See also termite colony health assessment below.

VEGETATION-DWELLERS

Sweep netting

Sweep netting requires little equipment and always catches a wide range of vegetation-dwelling and visiting fauna. Samples are taken along fixed transects and should be carried out at the same time of day in a given study. Night sweeping is beneficial for trapping some groups, e.g. grasshoppers. The position of the transect should be selected randomly or using stratification (depending on the habitat). During sampling the operator walks at a constant, steady speed, repeatedly sweeping the net from side to side (to cover an area of ± 1 m on either side) over a fixed distance, e.g. 50 m. This distance can be varied, depending on the vegetation and the invertebrate species of interest. A minimum of 10 transects per treatment area should be sampled.

Sweep netting can also be used on bushes and trees. In this case, either the time spent sweeping or number of sweeps made should be standardized. Up to 3 min is an appropriate length of time to sweep per sample, or 70–100 sweeps.

Limitations The fauna caught in sweep nets is influenced by vegetation type, height of sweep, vigour of sweep, number of sweeps, speed of walk, temperature, rainfall, wind speed, light intensity, time of day, and season. All these must be standardized within a given study. This method produces data on relative abundance only.

Processing Anaesthetize or kill fauna and sort from debris on a white tray, count and identify. Weigh if biomass estimates required (preferably oven-dried).

Resulting data Numbers of invertebrates, biomass and species composition.

Fauna sampled A wide range, particularly Araneae, Orthoptera, Mantodea, Hemiptera, Lepidoptera (particularly larvae), Diptera, Hymenoptera and certain Coleoptera.

Sampling period Preferably weekly, at set time of day (e.g. 09.00–12.00 h).

Equipment Sweep nets can be bought or made from locally available materials. Marker flags should be made up in advance. Pyrethroid insecticide spray may be used to knock-down invertebrates.

Staff required 1. Additional staff will be needed to sort, identify and count the catch; 3–5 staff ideal.

Other methods

Suction samplers, e.g. Dvac (Southwood, 1966) and other suction devices (Stewart and Wright, 1995); vegetation beating (Southwood, 1966; Ausden, 1996); photoeclector (Törmälä, 1982); and sugaring for moths (Ausden, 1996). See also transect counts below.

FLYING INSECTS

Malaise trap

Malaise traps are useful for making general collections of flying insects. Hymenoptera and Diptera are particularly vulnerable to this technique, Hemiptera, Lepidoptera, Orthoptera and Coleoptera to a lesser extent. Standardization of Malaise trap catches is extremely difficult (Grant, 1989) and initial setting of the trap is a question of trial and error to attain highest possible numbers of insects in a given time period. Traps are best set close to trees or bushes if possible, with the tallest end pointing towards the sun. Care should be taken to make sure that the surfaces are as taut as possible and that the material, particularly the middle wall, reaches the ground. The collection bottle should be angled to allow easy access for insects from the top of the trap into the jar. The jar should be charged with 70% alcohol. Traps may be left for 1 day or up to 1 month before emptying, though if left for longer periods it is advisable to check the traps regularly to ensure collection jars have not dried out, traps have not been damaged or collapsed, spiders have not built webs over the entrance to the collection jar, etc. Several traps will be necessary for each treatment and as many should be used as can be emptied and catches sorted within the time scale of the study.

Limitations Catch is dependent on vegetation type of habitat, temperature, wind direction and speed, light intensity, rainfall, season, and colour and size of the trap. Malaise traps are large and conspicuous and may be damaged or removed by animals or people. Sample processing is very time consuming. They measure activity abundance.

Processing Collection bottles containing trapped fauna should be removed from the trap and replaced with fresh ones. Back at the laboratory, fauna should be strained from the alcohol, transferred to a Petri dish (or similar) and sorted, counted and identified as appropriate.

Resulting data Species composition, relative abundance and biomass.

Fauna sampled Mainly flies (Diptera), Lepidoptera and Hymenoptera, with some Coleoptera, some Orthoptera and some Hemiptera.

Sampling period Empty traps every 5–10 days. The period between trap emptying should be kept constant to allow comparable catches. Traps should be emptied at the same time of day on each occasion. Avoid use during heavy rains in the wet season.

Equipment Traps can be made from locally available material (cotton mosquito netting or 'Nitet') of an appropriate colour. The roof of the trap should be of white material. The collection unit can usually also be constructed from locally available plastic jars (see method sheet). Marker flags should be made in advance. Alcohol (or other preservative) should be available locally. A GPS is useful to mark (and hence map) the position of individual traps.

Staff required 2. More staff may be necessary to sort and identify the catch; 3–5 staff ideal.

Water trap

Water traps consist of a coloured dish containing water, to which many flying insects are attracted. The dish is usually placed on the ground or, where vegetation is high, may be placed on a stand. Insects enter the trap and drown. Different coloured traps attract different groups of insects (e.g. yellow for dipteran flies, aphids, some beetles and chalcidoid wasps). Red, blue and green traps are less effective, particularly for Diptera. At least 10 traps should be used for each treatment area, but the more that can be processed then the more reliable will be the result. The same size, colour and type of trap should be used for the different treatment areas and they should be set at a standard height above the vegetation; highest numbers will be caught if the trap is set just

above the level of the surrounding vegetation. They should be filled to 1 cm from the top with water and a few drops of detergent should be added to reduce surface tension. Traps should be checked regularly and frequently.

Limitations Catch is dependent on vegetation type, temperature, wind speed, light intensity, rainfall, season, colour of trap and height of the trap above vegetation. They measure activity abundance.

Processing Trapped fauna can be removed with a nylon mesh sieve on a handle, or by pouring the contents through a small piece of muslin or nylon mesh (Noyes, 1982). They may then be transferred to a white tray (or similar) for sorting, counting and identification.

Resulting data Species composition, relative abundance and biomass.

Fauna sampled Mainly aphids (Homoptera: Aphididae), flies (Diptera), Hymenoptera, some Coleoptera, and some Lepidoptera.

Sampling period Empty traps daily. Avoid use during heavy rains in the wet season.

Equipment Traps can be adapted from locally available bowls or dishes, painted the appropriate colour. Marker flags should be made in advance. Detergent should be available locally.

Staff required 1. More staff may be needed to sort, identify and count the catch; 2–3 staff ideal.

Transect counts

A set route is walked and all fauna of interest are identified and counted within a set distance (or area) to either side and in front of the recorder. Transect counts can be used for Lepidoptera, Odonata, Orthoptera, Araneae (spider web counts) and less commonly Hymenoptera, Diptera and Coleoptera. Transects may be quite long depending on fauna (up to 2 km) and are divided into sections representing different microhabitats. Data for each sub-section should be recorded separately. Transects should be walked regularly, at least once a week. The procedure varies depending on the invertebrates of interest, and the following is suitable for butterflies only (Pollard, 1977).

The transect should be walked at a steady, slow pace and all insects seen within an imaginary ‘box’ extending 5 m in front, 5 m high and 2.5 m to either side should be recorded. Any insects which cannot be positively identified on sight, should be caught and identified. If they cannot be caught, the record should be ignored. Temperature, wind speed and sunshine should be recorded at the beginning and end of the transect and more frequently if possible. The length of the transect (and any sub-sections) should also be noted.

Limitations This method is subject to many variables and provides only a relative estimate of abundance. Transects must be carefully matched in terms of habitat, vegetation type, microclimate, length, etc. The transects must always be walked at the same time of day (see ‘Sampling Period’).

Processing A prior knowledge of the fauna to be sampled is necessary, so that identification can be made speedily and accurately *in situ*.

Resulting data Counts of numbers of invertebrate of interest seen.

Fauna sampled Butterflies (similar but slightly different methods for grasshoppers, dragonflies, etc.).

Sampling period Walk transects at least once a week. Wherever possible, transects in sprayed and unsprayed areas should be walked on the same morning or afternoon and in similar climatic conditions. The time of day at which transects are walked should be kept as consistent as possible. Pre-spray monitoring should be for at least 4 weeks and post-spray for at least the first, third and sixth months (more frequently where possible).

Equipment Record sheets should be made up or photocopied in advance (see example after method sheet). A butterfly net can be made from locally available materials or purchased. Sample vials and a thermometer will have to be purchased. Marker flags should be made up in advance.

Staff required 1 or 2 (but do not change the number of people used once sampling has begun). If 2 people, one calls out sitings while the other records data and must not contribute to sitings.

Honeybee activity at hives

Impacts of pesticides on honeybees can best be measured by quantification of activity and/or swarm size and comb production. Artificial hives may be constructed or natural hives monitored. The numbers of bees entering and

leaving hives should be recorded during a set period (e.g. 3 min). Such worker bee activity should be monitored each hour over a standardized period (e.g. 09.00–12.00 h), over a number of days before and after spraying. Bee deaths can be monitored by collecting bees falling from hive entrances for several days before and after spraying. Hive desertion should also be monitored. No method sheet is provided for this technique and an apiculturist or other bee specialist are best consulted. The swarm can be weighed after smoking and comb production estimated by direct observation (Douthwaite *et al.*, 1988). As many hives should be monitored as possible, but a minimum of 10 per treatment area is required.

Other methods

Suction traps, lure traps (Southwood, 1966); light traps (Butler and Kondo, 1991); sugaring for moths (Ausden, 1996); window traps (Chapman and Kinghorn, 1955); wind-orientated traps (Vogt *et al.*, 1985); transect methods for grasshoppers (FAO, 1994); and dragonflies (Brooks, 1993).

ARBOREAL INVERTEBRATES

Funnel or sheet trap

Funnels or sheets can be laid out on the woodland floor to catch invertebrates which fall from the canopy, 'knocked-down' by aerially applied insecticide (Grant, 1989). The traps should be reasonably large ($\pm 2 \text{ m} \times 2 \text{ m}$). Smaller sheets or funnels can be adapted to measure knock-down around sprayed tree trunks (Lambert *et al.*, 1991). Sample trees should be matched by species, girth and surrounding woodland type. This gives a good guide to fauna suffering acute effects of insecticide applications. If paired sheets, one of which is impregnated with insecticide, are used, estimates of recovery of fauna suffering knock-down can be made. Standardization is achieved by recording fauna in collectors at set times, preferably at first light to avoid predation of catch.

Limitations Catch is dependent on habitat, temperature, rainfall, wind speed, predation from unattended traps, and recovery rate from knock-down. This is **not** a quantitative method. Sheets are prone to inversion by wind and rocks or other heavy objects should be used to anchor the sheet down.

Processing Remove fauna from sheet or funnel *in situ*, using forceps, pooter, paintbrush, etc.

Resulting data Species composition of susceptible fauna.

Fauna sampled A wide variety of invertebrates, depending on habitat sampled.

Sampling period Twice daily (or more frequently, if possible), starting as soon after spraying as possible. Pre-spray sampling at set time daily.

Equipment Traps can be easily made from locally available materials, e.g. cotton, linen or nylon bed sheets. Support poles can be fashioned on site (provided there are trees nearby).

Staff required 2 (minimum).

Trunk trap

These traps are useful in assessing faunal composition and relative abundance of invertebrates which inhabit or regularly move up and down tree trunks. They work best on trunks which are relatively smooth, but can be adapted to any surface. Design for a simple trap is given by Moeed and Meads (1983), which can be adapted for use with different materials readily available in developing countries, if necessary. Sample trees should be matched by species, girth and surrounding woodland type. Traps should be set at a standard height above the ground (e.g. 1 m). Collecting vessels should be charged with 70% alcohol or formalin and may be left for up to a week before emptying. Moeed and Meads' trap has a removable collecting tray, but if such traps are not available a simple hand pump can be adapted to empty the trap (see method sheet).

Limitations Dependent on tree species, girth, bark type, season, woodland type. Gives relative abundance only. Dependent on activity.

Processing Extract fauna from trap using forceps and vacuum pump. Sort, count and identify fauna in white tray or Petri dishes.

Resulting data Species composition, numbers and biomass.

Fauna sampled A variety of invertebrates including Acari, Araneae, Chelonethi, Collembola, Thysanura, Psocoptera, Thysanoptera, Coleoptera, Orthoptera, Hemiptera, Hymenoptera, Mantodea and Blattodea.

Sampling period Empty traps weekly.

Equipment Marker flags or paint, traps can be made from locally available plastic boxes and heavy gauge plastic sheets. Sample pots have to be purchased. A suction pump will also have to be adapted using locally available materials, but will require the purchase of a basic pump of some type.

Staff required 2. Additional staff may be needed to sort, identify and count catch; 3 or more staff ideal.

Direct collection

Direct removal of invertebrates using an aspirator (pooter) or forceps can also be used in sampling trunk-dwellers (Ausden, 1996). Collection should be carried out for a set period of time (e.g. 5–20 min, depending on the fauna of interest and the tree and habitat type).

Other methods

Beating (Ausden, 1996); adapted Dvac (Lambert *et al.*, 1991); canopy sampling (Basset *et al.*, 1997). **Note:** Canopy fogging techniques are **not** recommended for pesticide impact studies.

SOIL INVERTEBRATES

Soil cores

Absolute population measures can be made by extracting invertebrates from soil cores. To make comparisons of numbers, diversity or biomass of fauna from different areas, soil type should be the same as should the volume of soil taken and the depth to which the core is taken. Cores can be taken using a spade, trowel, auger, steel tube or other digging implement. In general, pesticides will not normally penetrate far down into the soil horizon, so it is rarely useful to take cores down to deeper than 10 cm and usually 5 cm will be adequate to assess any impact of a pesticide on soil fauna. Small cores (up to 10 cm diameter) can be subjected to Tulgren funnel extraction or flotation extraction, whilst larger cores can be hand-sorted to extract macro-invertebrates. Flotation and sieving can yield information on virtually all fauna present in a core (including immobile stages, e.g. eggs and pupae). Tulgrens provide a more limited yield, requiring movement by fauna and generally do not provide useful data on nematodes, annelids and many of the more delicate Acarina and insects.

Limitations Fauna sampled and numbers obtained are dependent on soil type, season, time of day, weather conditions, volume of core, depth to which core is taken, and surface vegetation.

Processing Cores may either be broken up in a white tray and sorted by hand (to extract macro-fauna) or fauna extracted from cores using Tulgren funnels (see method sheet) or flotation techniques (see method sheet). Sort, count and identify fauna in Petri dishes. Mount micro-invertebrates on microscope slides for examination and identification.

Resulting data Species composition, numbers of individual species or taxa and biomass.

Fauna sampled A variety of invertebrates including Annelida, Nematoda, Myriapoda (Diplopoda, Chilopoda, Pauropoda and Symphyla), Isopoda, (Chelonethi), Acari, Collembola, Thysanura, Araneae, Isoptera, Psocoptera, Thysanoptera, Coleoptera, Orthoptera, Hemiptera, Hymenoptera and Blattodea.

Sampling period Take cores weekly (for 3–4 weeks pre-spray) and weekly for the first month post-spray, then monthly for the following 3–6 months. Where Tulgren extraction is used, funnels should be left in full sun for at least 5 days.

Equipment A soil corer can be made from locally available metal tubing. Alternatively, a spade or trowel can be used to take a fixed volume of soil. All the equipment necessary for flotation extraction of invertebrates can be adapted from locally available materials. Tulgren funnels can be made using metal or enamelled funnels (providing these can be bought locally), but the size *must* be standardized. A rack to hold the Tulgrens can be made from

wood or scrap metal and where no laboratory or electricity is available, the rack of Tulgrens can be left in full sunshine. In the dry season in the tropics, the high temperature and low humidity will generally allow reasonable extraction of invertebrates.

Staff required 1 (preferably 2). Additional staff will be needed to sort, identify and count the catch; 3–5 staff ideal.

Litter bags

Leaf litter bags can be used to sample soil invertebrates (particularly micro-fauna), but provide a relative measure of abundance only. Bags of different mesh size can be used to allow access to, or exclude a particular size range of invertebrates. Mesh sizes of around 4 mm will generally allow access to any soil-dwelling invertebrates; mesh of around 600 μm will allow access to nematodes, Collembola and mites but exclude macro-invertebrates, whilst mesh sizes below 60 μm will exclude all but the smallest micro-invertebrates. If bags are filled with a known weight and/or area of dry leaf material, then removal and decomposition can be assessed quantitatively as well. Leaf material used must be the same for each bag and each treatment area. If macro-invertebrates are of interest, large numbers of bags are needed in each treatment area, with a minimum of 50 of each mesh size. Bags may be tethered on the soil surface or buried in soil. Any depth can be used (provided it is standard for all treatment areas), but between 10 cm and 15 cm is recommended (maximum 30 cm). Different fauna are likely to occur in different seasons and thus time of burial and retrieval are important. Invertebrate density is likely to be higher during the rainy season, but bags should be left buried or tethered for a minimum of 1 month regardless of season. On retrieval, each bag should be immediately transferred to a separate plastic bag or plastic box. All soil and debris should be knocked from the bags and collected. This material may then be subjected to flotation extraction (Murphy, 1962) (see also method sheet) and sieving to remove and collect invertebrates present.

Limitations The fauna sampled is dependent on soil type, leaf litter presence and depth, season and woodland type. This method gives relative abundance only. It is dependent on the activity of organisms. The fauna extracted will depend on sample processing (which should thus be standardized).

Processing Extract fauna from litter in bags using hand sorting/flotation. Sort, count and identify fauna in a white tray or Petri dishes (mount micro-fauna on glass microscope slides and view under a high power microscope).

Resulting data Species composition, numbers and biomass.

Fauna sampled A variety of invertebrates including Annelida, Nematoda, Myriapoda, Isopoda, Cheloni, Acari, Collembola, Thysanura, Araneae, Isoptera, Psocoptera, Thysanoptera, Mantodea, Blattodea, Orthoptera, Hemiptera, Coleoptera and Hymenoptera.

Sampling period Length of time before collection depends on whether sampling invertebrates is combined with observing litter decomposition. If so, see chapter 7 on soil processes for timings. If only used for invertebrate samples then: tethered bags: wet season – leave in place for a minimum of 4 weeks pre-spray; replace bags at time of spraying and leave in place for a minimum of 4 weeks (maximum 2 months); dry season – leave in place for a minimum of 4 weeks pre-spray; replace bags at spray time and leave in place for a minimum of 4 weeks, maximum 3 months. Buried bags: wet season – leave in place for a minimum of 4 weeks pre-spray; replace bags at time of spraying and leave in place for a minimum of 4 weeks (maximum 3 months); dry season – leave in place for a minimum of 4 weeks pre-spray (maximum 6 months).

Equipment Litter bags can be made if nylon or plastic mesh material is available locally. Leaf litter can be collected locally, so long as leaf type is kept standard. Maps to mark positions of bags or GPS to note position of bags and markers.

Staff required 1 (preferably 2). Additional staff will be needed to sort, identify and count the catch; 3–5 staff ideal.

Termite colony health assessment

Measuring the relative abundance of social insects like termites and ants is notoriously problematic. However, the only way in which a pesticide can have a significant effect on these insects is if the health of the colony is affected. The death of individual workers is largely irrelevant, unless catastrophically large in extent. The colony health of mound-building termites can be assessed by monitoring the length of time taken for workers to repair any damage to the termitarium. The mound and its structure are crucial to maintaining a healthy colony (through

temperature regulation and protecting the colony from predators or other enemies, and from rain, wind, etc.), and thus any damage to the mound will be a priority for repair by the colony's workers. A healthy colony will rapidly mobilize workers to go to the scene of any damage and instigate repair.

Inflicting deliberate damage on a pre-determined number of termite mounds and recording the length of time taken for the colony to repair the damage is thus a useful tool in assessing the relative colony health. The technique is simple, quick and does not require taxonomic expertise. The larger the number of mounds that can be included, the better (depending on the size of the study area and of the mounds). In general, between 50 and 100 mounds is ideal (but for the larger Macrotermitinae, where mound density will generally be lower, 30 mounds may be adequate). The methodology can be adapted to the time available within the study and/or the persistence of the active ingredient (a.i.) (see method sheet).

Limitations To date, this method has only been tested on mound-building termites. It is dependent on season and species. It gives a relative measure only.

Processing Specimens should be taken from each mound of differing appearance, for identification.

Resulting data Termite species (following identification by termite specialist); time taken to repair damage inflicted on the mound.

Fauna sampled Any mound-building termite species.

Sampling period Either daily for first 10 days, followed by weekly for next 3 months, followed by monthly for next 6 months; or weekly for first 8 weeks, followed by monthly for next 9 months.

Equipment All necessary equipment can be purchased locally or made from local materials.

Staff required 1–2.

Other methods

Monolith sampling (Anderson and Ingram, 1989); formalin drenching for earthworms (Southwood, 1996).

COLLECTING TERRESTRIAL INVERTEBRATES FOR RESIDUE ANALYSIS

Any of the methods described above which result in a dry sample can be used to collect invertebrates for residue analysis or the animals can be collected directly using an aspirator or forceps. Residue analysis is expensive and great care should be taken with sampling to prevent cross-contamination and other errors outlined in chapter 6 which will lead to spurious results. Attempts to relate residue levels to population effect is often disappointing because of the variability of residues and it is important to follow methods in chapter 6 meticulously to avoid this. Collecting implements should be **absolutely** clean before sampling begins and different ones used for samples from different treatment areas, or they should be washed in acetone between samples. Fauna should be collected into aluminium canisters or vials and *frozen* or kept in *10% formalin* solution until residue analysis can be undertaken. Vials should have aluminium lids *without* plastic inserts, and should be clearly labelled (see chapter 6). Advice from an experienced pesticide chemist should be sought.

DATA PROCESSING

Most of the data collected using the methods described in this chapter will be in the form of counts and will be estimates of relative abundance, e.g. numbers of ladybird beetle sp. g caught in a sweep net sample, numbers of the spider *Habrocestum risicalli* caught in pitfall trap S4, etc. Processing data of this kind for analysis is covered in several places within the handbook (see chapter 1, Worked Example; chapter 2).

However, several of the methods can also provide data on the species present at a particular site, in a particular trap or sample, i.e. species composition. From this, other data can be compiled on species richness, species diversity and species similarity between traps, samples or sites. Comparisons of these data can be made if the

data are processed in a suitable way. It is often then possible to test differences in species richness or diversity statistically.

Similarity

There are a number of indices, which describe the similarity in species composition between samples or sites. Sørensen's Quotient (QS) is given by the formula:

$$QS = \frac{.2j}{(a + b)} \times 100$$

where j = the number of species common to both samples
 a = the number of species recorded in sample A
 b = the number of species recorded in sample B

Sørensen's Quotient increases as the number of species common to both samples increases and reaches 100% similarity when all species are common to both samples. Sørensen's Quotient is dependent on sample size and thus of limited value.

Mountford's Index overcomes this problem and is given by:

$$e^{a!} + e^{b!} = I = e^{(a + b - j)!}$$

where a , b and j are as above.

I is obtained by interpolating within the table of exponentials, using the following expression as an approximation to I :

$$\frac{2j}{2ab - (a + b)j}$$

From here, it is possible to classify sites based on a hierarchical comparison of their indices of similarity. The sites are first compiled into a coincidence table (see Table 8.2) which has sites as both column and row headings and then the value of the similarity index between each pairing placed in the appropriate box in the table.

These two sites are then grouped together and indices of similarity calculated between this group and each of the remaining sites. A reduced coincidence table is then compiled. The pair with the next highest index of similarity selected and the procedure of combining sites and re-evaluating indices repeated.

The sites are then plotted on a graph of similarity producing a cluster arrangement (see Figure 8.1).

Diversity

There are a number of indices used to measure (or, strictly speaking, summarize) the diversity of fauna found at any given site in any particular habitat. All have drawbacks and none are perfect descriptions of species diversity. One of the most common and widely used is the Shannon-Wiener function (H'). The method of calculating this index is given in chapter 11, page 224.

Table 8.2 Coincidence table giving species similarity for 10 DDT sprayed and 10 unsprayed sites in savanna woodland based on Sørensen's Quotient of similarity applied to pitfall trap catches of invertebrates

	U1	U2	U3	U4	U5	U6	U7	U8	U9	U10	S1	S2	S3	S4	S5	S6	S7	S8	S9	S10
U1																				
U2	50.7																			
U3	45	44.9																		
U4	41.1	39.6	37.3																	
U5	44.2	44.8	50.3	36.9																
U6	39.6	41.3	37.2	37	38.8															
U7	35.1	33.9	32.9	35	33.3	31.9														
U8	40.6	40.2	37.9	52.8	38.9	39.9	34.2													
U9	36.1	41	33.3	37.2	42.1	38.1	33.7	40												
U10	40.5	40.2	38.7	39.4	39.4	52	32.8	41.1	38.4											
S1	38.9	40.7	40.2	43.1	39.3	38.3	32.5	38.4	37.8	40										
S2	39.6	40	38.8	41.9	37.9	37.9	33	40.1	39.2	37.8	46.2									
S3	31.9	32.7	29.8	34.2	32	31.9	32.8	33.2	26.4	30.5	33.2	31.6								
S4	30.5	33.1	33.7	36.2	33.8	34.3	28.2	31.8	34.7	32.6	35.2	33.2	42.6							
S5	34.6	36	32.2	35.3	36.8	30.7	41.7	33.3	36.3	35.8	31	34	36.2	34.2						
S6	31.1	35.5	31	31.2	34.2	32.2	42.1	34.8	32.9	36.7	35.9	31.6	35.8	35.9	44.9					
S7	33.7	36.1	35.4	33.5	29.7	32.4	23.2	36.9	30.9	33	33.9	32.7	41.5	44.6	33.9	34.1				
S8	32.3	30.6	32.1	33.9	31.7	35.1	22	32.8	35.7	32.1	36.5	34.9	40.8	43.9	36.6	35.7	56.5			
S9	31.3	31.7	36.6	34.4	36	32.5	27.7	36.4	37	33.4	31.4	32	41.9	44.2	34.2	33.9	47.9	48.6		
S10	34.5	33.6	30.8	36.7	35.4	35.6	41.3	34.1	31.9	35.1	35	31.5	36.3	35.4	49	44.2	35.8	36.4	37	

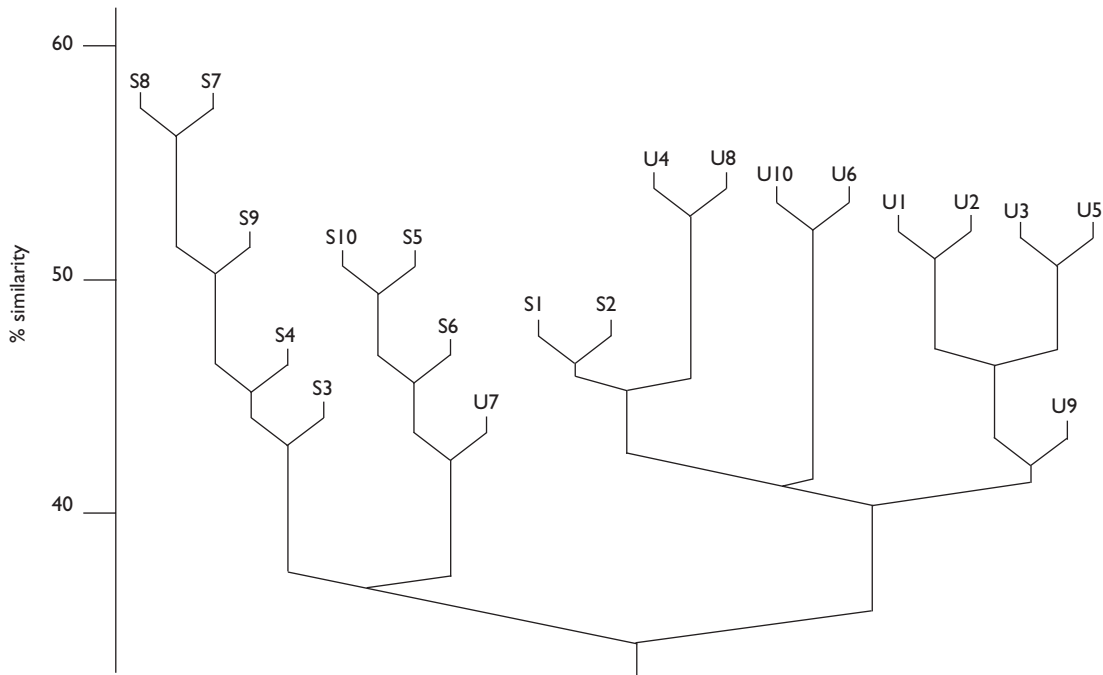


Figure 8.1: Dendrogram of species similarity of invertebrates caught in pitfall traps in two different treatment areas in Zimbabwe. Hierarchical classification of sample sites according to Sørensen's Quotient of similarity. U1–U10 unsprayed; S1– S10 sprayed with DDT.

MOUNTING TECHNIQUES FOR STORAGE AND IDENTIFICATION OF INVERTEBRATES

The majority of invertebrates caught using the methods outlined above may be stored in 70% alcohol in stoppered, labelled, glass or plastic vials. However, some groups need mounting dry to display adequately the taxonomic features needed for their identification.

Butterflies and moths, for example, should be pinned on setting boards, with the wings spread for display. Larger beetles (>1 cm) should be pinned through the right elytron, with legs and antennae displayed, where possible. Smaller beetles should be mounted on card points, using Coleoptera gum, again displaying legs and antennae if possible. Orthoptera should be pinned, with legs and one or both pairs of wings displayed. Larger Diptera and Hymenoptera should be pinned through the thorax with wings on one or both sides of the body displayed (Figure 8.2). In some cases, side pinning is necessary, for Diptera in particular (see Figure 8.3). Small Diptera and Hymenoptera (e.g. Chalcidoidea) may be micro-pinned (see Figure 8.4), or gummed on to card points (Figure 8.5) or cards. The smallest Diptera and Hymenoptera may need to be mounted on microscope slides (Figure 8.6). Mites and Collembola should generally be mounted in lactophenol on microscope slides.

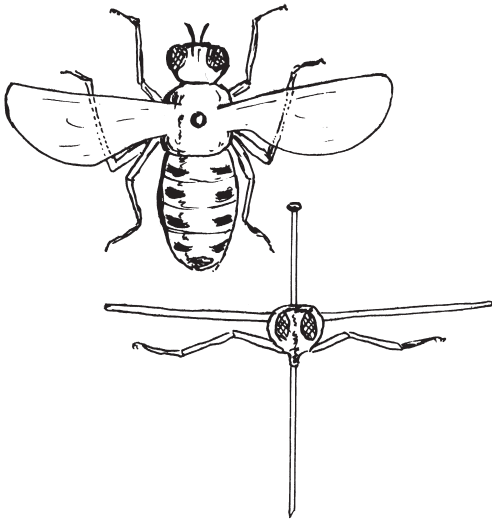


Figure 8.2: Setting of large Lepidoptera, Diptera and Hymenoptera, etc.

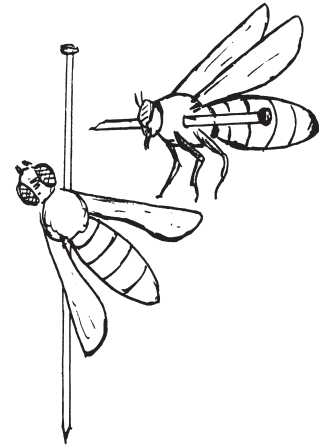


Figure 8.3: Side pinning

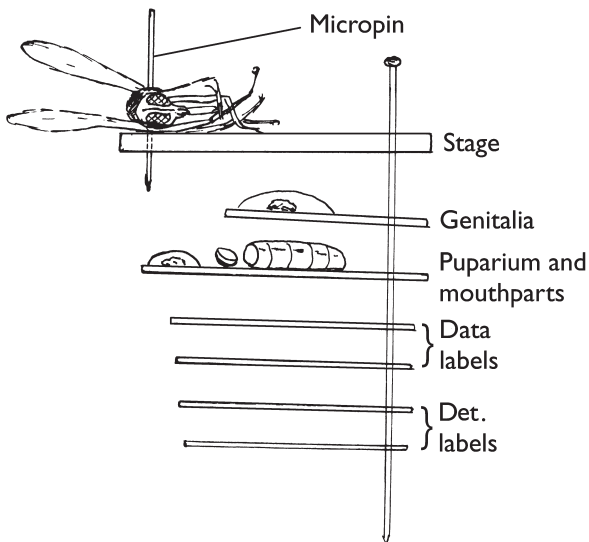


Figure 8.4: Staging for microlepidoptera, small Diptera, Coleoptera and Hymenoptera

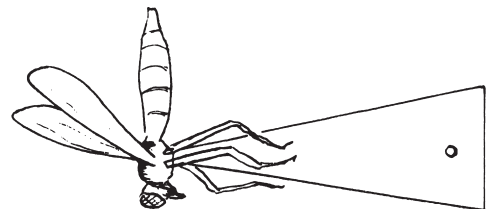
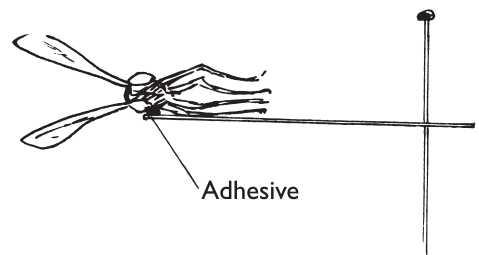


Figure 8.5: Card pointing for small Coleoptera, Diptera, etc.

USEFUL CONTACTS

The Curator, Invertebrate Collections, Natural History Museum, Cromwell Road, London SW7 5BD, UK.

Watkins and Docaster, Entomological equipment and supplies, P O Box 5, Cranbrook, Kent TN18 5EZ, UK.

REFERENCES

- ADIS, J. (1979) Problems of interpreting arthropod sampling with pitfall traps. *Zoologische Anzeiger, Jena*, **202**(3/4): 177–184.
- ANDERSON, J.M. and INGRAM, J.S.I. (eds) (1989) *Tropical Soil Biology and Fertility: A Handbook of Methods*. Wallingford, UK: CAB International.
- AUSDEN, M. (1996) Invertebrates. pp. 139–177. In: *Ecological Census Techniques. A Handbook*. Sutherland, W. J. (ed.). Cambridge: Cambridge University Press.
- BASSET, Y., SPRINGATE, N.D., ABERLENC, H.P. and DELVARE, G. (1997) A review of methods for sampling arthropods in tree canopies. In: *Canopy Arthropods*. Stork, N.E., Adis, J. and Didham, R.K. (eds). London: Chapman and Hall.
- BROOKS, S. J. (1993) Review of a method to monitor adult dragonfly populations. *Journal of the British Dragonfly Society*, **9**(1): 1–4.
- BUTLER, L. and KONDO, V. (1991) Macrolepidopterous moths collected by blacklight trap at Cooper's Rock State Forest, West Virginia: A baseline study. *Bulletin*, No. 705. West Virginia University, Agricultural and Forestry Experiment Station.
- CHAPMAN, J.A. and KINGHORN, J. M. (1955) Window flight trap for insects. *Canadian Entomologist*, **87**: 46–47.
- CRITCHLEY, B.R., COOK, A.G., CRITCHLEY, U., PERFECT, T.J. and RUSSELL-SMITH, A. (1980) The effects of crop protection with DDT on some elements of the subterranean and surface active arthropod fauna of a cultivated forest soil in the humid tropics. *Pedobiology*, **20**: 31–38.
- DOUTHWAITE, R. J., MAHMOUD, D.A. and ABDISALAM, S.T. (1988) Effects of drift sprays of endosulfan applied for tsetse fly control on honeybees (*Apis mellifera* L.) in Somalia. *Journal of Agricultural Research*, **27** (1): 40–48.
- EBERHARDT, L.L. and THOMAS, J.M. (1991) Designing environmental field studies. *Ecological Monographs*, **61** (1): 53–73.
- FAO (1994) *The Desert Locust Guidelines II. Survey*. D1/T3375/E/1/4.94/500. Rome: Food and Agriculture Organization of the United Nations.
- FRENCH, J.R.J. and ROBINSON, P.J. (1981) Baits for aggregating large numbers of subterranean termites. *Journal of the Australian Entomological Society*, **20**: 75–76.
- GRANT, I.F. (1989) Monitoring insecticide side-effects in large-scale treatment programmes: tsetse spraying in Africa. pp 43–69. In: *Pesticides and Non-target Invertebrates*. Jepson, P. C. (ed.). Andover, UK: Intercept.

- LAMBERT, M.R.K., GRANT, I.F., SMITH, C.L., TINGLE, C.C.D. and DOUTHWAITE, R.J. (1991) Effects of deltamethrin ground-spraying on non-target wildlife. Environmental impact assessment of ground-spraying operations against Tsetse flies in Zimbabwe. *Technical Report*, No. 1. Chatham, UK: Natural Resources Institute.
- MOEED, A. and MEADS, M.J. (1983) Invertebrate fauna of 4 tree species in the Orongorongo Valley, New Zealand, as revealed by trunk traps. *New Zealand Journal of Ecology*, **6**: 39–53.
- MURPHY, P.W. (ed.) (1962) *Progress in Soil Zoology*. London: Butterworths.
- MURPHY, C. F. and CROFT, B.A. (1990) Forest ant composition and foraging following aerial spraying of carbaryl to suppress Western Spruce Budworm. *Canadian Entomologist*, **122**: 595–606.
- NOYES, J.S. (1982) Collecting and preserving chalcid wasps (Hymenoptera: Chalcidoidea). *Journal of Natural History*, **16**: 315–334.
- POLLARD, E. (1977) A method for assessing changes in the abundance of butterflies. *Biological Conservation*, **12**: 115–134.
- ROGERS, L.E., BUSCHBOM, R.L. and WATSON, C.R. (1977) Length-weight relationships of shrub-steppe invertebrates. *Annals of the Entomological Society of America*, **70**: 51–53.
- SOUTHERTON, N.W., JEPSON, P.C. and PULLEN, A.J. (1988) Criteria for the design, execution and analysis of terrestrial non-target invertebrate field tests. pp. 183–190. In: *Field Methods for the Study of Environmental Effects of Pesticides*. Greaves, M. P., Smith, B. D. and Greig-Smith, P.W. (eds). *BCPC Monograph*, No. 40. Thornton Heath: British Crop Protection Council.
- SOUTHWOOD, T.R.E. (1966) *Ecological Methods*. London: Chapman and Hall.
- STEWART, A.J.A. and WRIGHT, A.F. (1995) A new inexpensive suction apparatus for sampling arthropods in grassland. *Ecological Entomology*, **20**: 98–102.
- STUBBS, A. and CHANDLER, P. (1978) *A Dipterists Handbook*. *The Amateur Entomologist*, Volume 15. Hanworth, UK: The Amateur Entomologist Society.
- SUTTON, S.L. (1972) *Woodlice. Invertebrate Types Series*. Oxford: Pergamon Press.
- TÖRMÄLÄ, T. (1982) Evaluation of five methods of sampling field layer arthropods, particularly the leafhopper community, in grassland. *Annales Entomologici Fennici*, **84**: 1–16.
- TINGLE, C.C.D. (1993) Bait location by ground-foraging ants (Hymenoptera: Formicidae) in mopane woodland selectively sprayed to control tsetse fly (Diptera: Glossinidae) in Zimbabwe. *Bulletin of Entomological Research*, **83** (2): 259–265.
- UNDERWOOD, A.J. (1994) On beyond BACI: Sampling designs that might reliably detect environmental disturbances. *Ecological Applications*, **4** (1): 3–15.
- VOGT, W. G., RUNKO, S. and STARICK, N.T. (1985) A wind orientated fly trap for quantitative sampling of adult *Musca vestustissima* Walker. *Journal of the Australian Entomological Society*, **24**: 223–227.

FURTHER READING

BETTS, C. (ed.) (1986) *The Hymenopterist's Handbook. The Amateur Entomologist*, Volume 7. Hanworth, UK: The Amateur Entomologist Society.

BROWN, R.A., JEPSON, P.C. and SOTHERTON, N.W. (1992) *Aspects of Applied Biology 31, Interpretation of Pesticide Effects on Beneficial Arthropods*. Wellesbourne, UK: Association of Applied Biologists, Horticultural Research Institute.

DOUTHWAITE, R.J. and TINGLE, C.C.D. (eds) (1994) *DDT in the Tropics: The Impact on Wildlife in Zimbabwe of Ground-spraying for Tsetse Fly Control*. Chatham, UK: Natural Resources Institute.

GREAVES, M.P., SMITH, B.D. and GREIG-SMITH, P.W. (eds) (1988) *Field Methods for the Study of Environmental Effects of Pesticides. BCPC Monograph*, No. 40. Thornton Heath: British Crop Protection Council.

JEPSON, P.C. (ed.) (1989) *Pesticides and Non-target Invertebrates*. Andover, UK: Intercept.

WALLWORK, J.A. (1976) *The Distribution and Diversity of Soil Fauna*. London: Academic Press.