Invertebrates: Malaise trapping

Version 1.0

This specification was prepared by Alison Evans in 2016.

Contents

Inventory and monitoring toolbox: invertebrates

DOCCM-599792

Department of Conservation
Te Papa Atawhai

Synopsis

The Malaise trap (Fig. 1) was invented by the Swedish entomologist René Malaise after finding that more insects were captured in his tent than he collected by netting. Malaise traps have wide applications and, when used in a standardised way, can contribute biological and ecological information to a variety of other fields, including taxonomy and systematics, biocontrol and biosecurity such as in forest health monitoring.

Figure 1. A standard Malaise trap set-up in forest. Note that the guy ropes are attached to trees but tent poles can be used if no suitable trees are available, and that the vertical panels are in contact with the ground. Malaise traps should be set up so the highest point faces north, the brightest direction.

Malaise traps are ‘tent-like’ passive intercept traps that primarily catch flying insects. They are usually set in contact with the ground to collect low flying species (they also catch some insects that emerge from the ground below the trap or that climb up from the ground). They can also be set in the forest canopy to sample flying insects.

The numbers of flying insects caught relate to both their activity levels (temperature and weather conditions) and their abundance. They are particularly good for sampling beetles. Huge numbers can be caught even over relatively short periods such as a week and, as only about 25% of New Zealand insect species have been described, the catch will often include species that are not able

to be identified or are new to science. However, the large numbers of insects caught even when a Malaise trap is set for only a week means that only species or groups of interest are usually sorted out. If the entire catch is of interest then subsamples may be taken for analysis.

Assumptions

There is a direct proportional relationship between the numbers of insects caught in a Malaise trap and the abundance of active insects of the same species in the environment.

Advantages

- Malaise traps are simple, easily transported, erected and serviced. They will work continuously once set.
- They can be used to monitor changes in seasonal abundance (provided the trap catch is collected weekly).
- They sample a wide diversity of predominantly flying insects.
- They do not require a power source.

Disadvantages

- Catch rates are highly susceptible to small changes in location and are highly site dependent.
- They only collect invertebrates that are able to fly and have a tendency to collect ‘vagrant’ or ‘transient’ species. They are expensive (particularly if you need several traps to get a representative sample).
- They catch and kill huge numbers of insects.
- They are not suitable for single insect species work or for studying insects of conservation value.
- They are affected by strong wind in exposed sites.
- If using 70% ethanol as a collecting fluid then it may evaporate (especially in exposed positions), or may deter some species from entering the trap.
- Not suitable for collecting moths because their diagnostic wing patterns are lost.

Safety issues

Ethyl alcohol (ethanol):

- Poison: Treat ethyl alcohol as a mild poison—it is not suitable for human consumption because it is made by distillation with benzene, which causes cancer.
- Fire: Be aware that ethyl alcohol is highly flammable (both liquid and as vapour).
- Do not use iso-propyl alcohol. It is less volatile but may impose health risks (e.g. heart fibrillation) in sensitised people (Cresswell 1995).

Propylene glycol antifreeze **should** be used because it is:

- Non-poisonous, but not suitable for human consumption.
- Environmentally friendly. It can be safely disposed of on soil or in water.

Ethylene glycol antifreeze **should not** be used because it is:

- Poisonous if ingested and can cause skin rashes on contact with skin.
- Not environmentally friendly. It must be disposed of at a special council disposal site.

Suitability for inventory

Malaise traps are particularly well suited for inventory because they catch a wide variety of flying insects and some ground active insects that climb. They are particularly suitable for flying beetles and flies, but they are unsuitable for moths (unless captured live) because the scales fall off destroying the diagnostic wing patterns.

Suitability for monitoring

Malaise traps are useful for monitoring whether a species is present or absent, and its relative abundance. This is because they are simple, work passively, and are easily operated. They are unsuitable for estimating relative abundance because multiple traps are necessary to counter the variability that occurs between traps and because it is impracticable to set and sort the large samples from multiple traps (replicates). As with other trapping methods Malaise traps do not sample a defined area (i.e. the insects fly in from a variety of distances from the trap).

Skills

The only requirement for setting up a Malaise trap and collecting the samples is attention to detail when following the instructions. Anyone who can pitch a tent can set a Malaise trap.

Sorting insect samples requires specialised training in entomology. This work may be outsourced to a suitable contractor. Sorting the sample by placing the invertebrates into major obvious groups such as beetles (Coleoptera), flies (Diptera), moths (Lepidoptera), ants and wasps (Hymenoptera), etc., and to collect everything you are not sure of into an ‘unknown’ group can speed up the process and reduce costs. Identification resources can be found on the Landcare Research website (http://www.landcareresearch.co.nz/resources/identification/animals). Other resources include ‘Invertebrate identification aids’ (doccm-388198) and ‘Invertebrates: advice and diagnostic support’ (doccm-2686377).[1]

[1] www.doc.govt.nz/documents/science-and-technical/inventory-monitoring/im-toolbox-invertebrates-advice-diagnostic-support.pdf

Resources

- Personnel—only one person is required to set up and service the Malaise trap.
- Notebook and pencil to record date, position and data associated with the position of the Malaise trap.
- Accurate map of area to identify the geographical boundaries of land and habitats.
- Malaise trap, poles and 'guy ropes' to passively catch insects.
- GPS to record position of the Malaise trap.
- Flagging tape to mark the position of the trap.
- Sample storage equipment—including an empty plastic jar suitable to fit on the Malaise trap, spare vials, a chilly bin, zip-lock bags, marker pens, 99% ethanol, pencil, labels, Chux cloth, white high-sided tray, sieve and funnel—to collect and store invertebrates for identification.
- Camera to record information on vegetation or habitat type and to take photos for presentations or reports.
- Safety equipment—such as cell phone, VHF radio and first aid kit—to contact emergency services if necessary or administer basic first aid.

To calculate the time required, take into account return travelling time to the site and add 1 hour for setting up the Malaise Trap. The time required to collect samples from the trap is about 5 minutes (perhaps 10 minutes if the trap needs adjustment or resetting) not including travel time. In most situations, sample sorting will be done in a laboratory rather than in the field.

Minimum attributes

Consistent measurement and recording of these attributes is critical for the implementation of the method. Other attributes may be optional depending on your objective. For more information refer to 'Full details of technique and best practice'.

DOC staff must complete a 'Standard inventory and monitoring project plan' (doccm-146272).[2]

Minimum attributes to record on the collection label:

- Name of location
- GPS position
- Type of sample (i.e. Malaise trap)
- Dates when sample was started and collected
- Full name of collector

[2] http://www.doc.govt.nz/Documents/science-and-technical/inventory-monitoring/im-toolbox-standard-inventory-and-monitoring-project-plan.doc

If information on the vegetation is required then this should be recorded using the standard RECCE plot method (see 'Vegetation: RECCE plots'—doccm-359575)[3] (Hurst & Allen 2007).

Data storage

Forward copies of completed survey sheets to the survey administrator, or enter data into an appropriate spreadsheet as soon as possible. Collate, consolidate and store survey information securely, also as soon as possible, and preferably immediately on return from the field. The key steps here are data entry, storage and maintenance for later analysis, followed by copying and data backup for security.

Summarise the results in a spreadsheet or equivalent. Arrange data as 'column variables', i.e. arrange data from each field on the data sheet (date, time, location, plot designation, number seen/collected, identity, etc.) in columns, with each row representing the occasion on which a given survey plot was sampled.

If data storage is designed well at the outset, it will make the job of analysis and interpretation much easier. Before storing data, check for missing information and errors, and ensure metadata are recorded.

Storage tools can be either manual or electronic systems (or both, preferably). They will usually be summary sheets, other physical filing systems, or electronic spreadsheets and databases. Use appropriate file formats such as .xls, .txt, .dbf or specific analysis software formats. Copy and/or backup all data, whether electronic, data sheets, metadata or site access descriptions, preferably offline if the primary storage location is part of a networked system. Store the copy at a separate location for security purposes.

Data from Malaise trapping can be of fundamentally different types:

- Voucher specimens for curation, identification and storage
- Numerical data (number of species or morpho-species collected)
- Descriptive data (location of trap, weather conditions, time of day, vegetation, etc.)

For inventory, voucher specimens will be necessary so that accurate identification can be made by a specialist taxonomist in the particular insect groups. Ultimately invertebrates collected for inventory should be kept for future reference and those of conservation value or taxonomic interest should be deposited in a museum or in the National Arthropod Collection administered by Landcare Research.[4] Institutions should be contacted first to find out their requirements.

Data should be stored in Excel spreadsheets for ease in manipulation and interpretation. DOC staff should enter records into the BioWeb database.

[3] http://www.doc.govt.nz/Documents/science-and-technical/inventory-monitoring/im-toolbox-vegetation-reece-plots.pdf

[4] http://www.landcareresearch.co.nz/resources/collections/nzac

Analysis, interpretation and reporting

Introduction

The following outline is intended to highlight some of the practical considerations of dealing with Malaise trap data and to provide an overview of the types of analysis appropriate for the collecting method used. Once the data have been collected and sorted into recognisable taxonomic units (RTUs), it is recommended that the data is summarised and presented either in a table or graphically. Basic data summary statistics and an overview of the common types of analysis used for invertebrate data (with worked examples) are provided in 'Introduction to statistical analysis of invertebrate monitoring data' (doccm-525907)[5]. However, extensive training in statistical methods is required before attempting any statistical analysis. Seek statistical advice from a biometrician or suitably experienced person during study design and prior to undertaking any analysis. The information provided in this section and in the 'Introduction to statistical analysis of invertebrate monitoring data' is intended to familiarise staff with some of the options available so that informative discussions can be held with a statistician. The information is not intended to be a comprehensive guide to data analysis.

Practical considerations

Malaise traps are most commonly used to assess relative abundance and diversity of invertebrates active in the subcanopy of forests or in shrublands where there is a reasonable amount of shelter. At the time studies are designed, decisions need to be made regarding which key groups are going to be analysed. This is usually based on the likely relative abundance of the target groups, the suitability of Malaise traps to collect the target groups and the capacity to identify the target groups. For example, wētā and spiders are occasionally collected in Malaise traps but this is not a suitable method for monitoring these species. Malaise traps are not suitable for monitoring moths as they are difficult to identify once submerged in ethanol. Malaise traps are often used to assess beetle assemblages as they represent a range of trophic groups, are more easily identifiable, and are thought to reflect the general characteristics of the invertebrate fauna occupying a site (Hutcheson & Jones 1999). The placement of the trap in relation to natural corridors and prevailing wind direction can bias results and will need to be taken into consideration. Although Malaise traps are not generally used in replicated studies, they are useful for monitoring temporal patterns in relative abundance (e.g. changes in the seasonal abundance of wasps) and to get an appreciation of the invertebrate community in a particular habitat type.

Analysis of Malaise trap data

Statistical methods that can be used to explore data collected from Malaise traps depend on the hypotheses being tested (the questions that you want answered) and the design of the study. The following issues should be addressed as part of your study design but may need to be considered when analysing the data as well:

[5] http://www.doc.govt.nz/documents/science-and-technical/inventory-monitoring/im-toolbox-statistical-analysis-of-invertebrate-monitoring-data.pdf

- As with many other collection methods, the results are a reflection of insect activity and the relative abundance of the species present at the time that the trap was active.
- The sampling unit (point) for Malaise traps is 'one' for each discrete collection of invertebrates.
- Malaise traps are rarely used to compare invertebrate fauna between sites or different treatments as you would need a number of them set up simultaneously.
- It is difficult to know what area they collect from and if additional traps are set up, it is important to ensure that they are set up some distance away (> 100 m) so that they collect independently.
- It may be important to accurately quantify environmental variables (e.g. associated richness of plant communities, distance from forest edges or other gradients such as altitude) so that they can be incorporated into multivariate analyses.
- Malaise traps catch such huge numbers of insects that it is sometimes advisable to take a subsample. This is usually done by taking a known fraction of the total volume ensuring that the sample is completely mixed beforehand.

Once the data from the Malaise trap have been summarised, without replication, the analysis is limited to calculating and reporting the diversity of various invertebrate groups (e.g. diversity of beetles) and exploring the data for relationships with environmental variables using multivariate analysis techniques (see 'Introduction to statistical analysis of invertebrate monitoring data'—doccm-525907)[6]. This might include determining whether certain species are indicative of specific environmental variables using a program such as PC-Ord or using ordination techniques to determine whether the species collected are responding to environmental gradients. If the results suggest that there are obvious associations between particular species or groups of species with the key environmental variables or gradients that have been measured, it is important to present these results in the context of the biology of those species. For example, can there be conclusions drawn about the relative abundance of fungal feeding flies in mature forest compared with the abundance on the edge of the forest where there are likely to be higher temperatures and less mature trees? If the Malaise traps have been used to assess the beetle fauna occupying an area, it may be possible to summarise the data in terms of the functional groups. This can provide important information about the role of the beetle community in the local environment (Hutcheson et al. 1999).

There are a large number of possible analyses and interpretations depending on the purpose of the sample. Most analyses require specialised statistical advice. For example, a short resume of some of the considerations in relation to beetle communities is provided in 'Case study A'.

It is important to check that the Excel data are transposed (i.e. columns to rows) and saved in an appropriate form for a specialised application for multivariate analyses procedures (e.g. PC-ORD; note that there are several other proprietary programs). The program CANOCO, which is an extension of the program DECORANA (Hill 1979), can be used to ordinate the data and display ecological affinities between catches in a representation of three-dimensional space.

[6] http://www.doc.govt.nz/documents/science-and-technical/inventory-monitoring/im-toolbox-statistical-analysis-of-invertebrate-monitoring-data.pdf

Although the multivariate procedures themselves use the inferential statistical tools to normalise data, these are only applied within species, thereby retaining the common denominator that makes quantity meaningful, and they describe community affinities based on both the qualitative and quantitative information, rather than claiming to be strictly quantitative. Like both the Malaise-trapped samples and the RECCE plots, the aim of the multivariate tools is for a general accuracy rather than for a focused precision which is not ecologically informative and may be entirely misdirected.

Species abundance may be transformed in Excel into the default abundance classes of TWINSPAN, as these classes were found best for discrimination of communities according to habitat by Hutcheson (1990). However, abundance distributions of most communities sampled to date show similar distributions, echoing the conclusions of May (1975), Zak (1992) and Tokeshi (1993), that all abundance distributions are essentially similar, with few common and many rare species at any one time. Some very high carbon-turnover habitats (e.g. late-rotation exotic pine forest) can show 'fatter' curves because of higher numbers of moderately abundant species (Hutcheson & Jones 1999).

The TWINSPAN abundance classes have also been used to create a simple diversity index which includes trophic information. However, this has since been recognised as being less informative than simple side-by-side comparisons of graphs which show mean trophic structure in terms of individuals and species. This enables immediate recognition of whether trophic components are composed of a few species in high abundance or of many species in low abundance.

Case study A

Case study A: Malaise trapping: understanding beetle composition over a range of vegetation types

Synopsis

This example from Hutcheson & Kimberley (1999) demonstrates how Malaise trapping can be used to study insect community composition over a range of vegetation types. They investigate the potential of using Malaise-trapped beetles to represent community composition and successional habitat types across the central North Island volcanic plateau. Hutcheson & Kimberley (1999) focus on beetles (Order: Coleoptera) because they represent close to 50% of known insect species in New Zealand (Watt 1982) and are well represented across terrestrial systems and functional groups.

Malaise trapping methods are passive and suitable for a wide range of habitats. When set correctly, they sample a wide range of flying, and some ground-living invertebrates, and they provide a simple and repeatable method for sampling insect communities.

Objectives

This particular Malaise trapping study had two specific objectives: (a) to confirm that a short sampling period in early summer provided good discrimination between different beetle communities (as implied by Hutcheson 1990) and (b) to illustrate that beetle communities are characteristic for vegetation type, and not location alone. Samples were included from similar habitats separated over space by 5–100 km, and over time by 4 years.

Sampling design and methods

Study sites

The study included two sampling series from the Waipapa Ecological Area (50 km northwest of Taupō) taken 4 years apart, and a sampling series from Rangitāiki, about 100 km southeast of the Waipapa sites. Successional habitat stages sampled at these three areas included heathland, advanced shrubland and mature podocarp forest. In the first year, at the Waipapa Ecological Area, Malaise traps were set in advanced shrubland and in mature podocarp forest. Four years later, a second Waipapa series was taken in a range of successional habitat types from heathland to mature podocarp/broadleaf forest. This involved seven sites with one trap at each. The Rangitāiki collection occurred over 1 year and included one trap at each of three heathland sites. Standard RECCE plots were measured at each site to document habitat and vegetation structure.

Trapping methods

Standard full-sized Malaise traps (2 m long by 1.2 m high) of the dimensions outlined by Townes (1972) were used to sample beetle populations as smaller, commercially available traps had earlier been found to collect an insufficient sample for characterising communities (Dugdale & Hutcheson 1997). A reasonably flat, log-free area of around 2 m^2 was chosen for each site and the base of the Malaise trap was pegged to the ground. Trap corners were tied out so the structure was erected like a tent. The collection jar is located at a high point at one end of the trap and the collection jar filled with 70% ethanol. The traps were tied to existing vegetation in order to minimise trap-site disturbance.

Beetles were caught between weeks 47 and 3 (counting the first week of January as week 1, these weeks in December and January are when most southern hemisphere adult insect activity occurs) for the first of the Waipapa series, and weeks 47–52 for the other two series. Samples were collected weekly.

All beetles were curated according to Walker & Crosby (1988) and organised into RTUs (see Hutcheson 1990 for detailed methods and discussion). Most taxa were able to be identified to species level but many were simply coded within family, subfamily or genera. The National Arthropod and Forest Research Institute collections were used in combination with a range of specialists to determine the taxonomic status of all beetles. The beetles were counted and assigned to simplified functional groups defined as: detritivores, herbivores, predators and aquatic species.

Analysis

Sørensen's similarity index (Krebs 1978) and polythetic divisive classification using TWINSPAN were compared in assessing sample affinities. The former only uses presence/absence of species, while the latter also uses abundances to assess community variation over time (the four 7-day samples per site), and space (i.e. between sites).

Results

The authors determined that Malaise-trapped beetles supported both hypotheses: a) beetle catches showed good discrimination into groups relating to habitat types when trapped over a 4-week period during the early part of peak adult beetle activity; and b) samples were characterised by habitat type and not just site. Although the study design had limited replication, the Malaise trapped beetles did show progressive change in community composition in relation to vegetation succession, from heathland through shrubland to tall podocarp/broadleaf forest. The approach also showed that beetle biodiversity was highest at the advanced shrubland successional stage. This implies that the value of advanced diverse shrubland to biodiversity conservation is currently grossly undervalued.

Limitations and points to consider

There are four main limitations to using Malaise traps. Firstly, a huge number and diverse range of invertebrates were collected, but only beetle data were used. A large by-catch of insects were killed but this probably had relatively minor effect on the overall insect fauna of the areas. Although not analysed here, all specimens were stored and these data represent a snapshot of the habitats and their insect communities which may outlast the actual habitats. The stored material is available for future studies and may reduce future sampling requirements. Material from this study, for example, has been used to evaluate the spread of adventives into natural systems, and new species in various orders have been recognised. Collecting was restricted to a short time during peak insect activity and collected species relative to their abundance, to minimise the catch.

Secondly, the traps do not take a representative sample of ground-dwelling species and they do not catch species that fly at other times of the year.

Thirdly, trap efficiency is affected by all sorts of weather conditions which are hard to quantify and may significantly bias results. However, in this example although some traps in low vegetation were exposed to wind, this did not significantly affect results (J. Hutcheson, pers. comm.). Weather has a far greater effect on approaches to sampling which do not use a simple continuous trap system. Wind may potentially damage traps, but if necessary a frame such as that reported in Faulds & Crabtree (1995) can be used to reduce this (J. Hutcheson, pers. comm.).

Fourthly, ethanol was used as a collection fluid and this could affect results if insects respond to it. This applies to all collection fluids, and using ethanol was found to be the most pragmatic and safest option. Ethanol evaporates, particularly in exposed sites, and its loss may be compensated by using a larger collection jar (and hence more fluid) on the collection attachment (Hutcheson 1991), or by replenishing fluid during the week.

References for case study A

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Full details of technique and best practice

How it works

Malaise traps are passive traps: they catch insects that fly within about 1 m of the ground (this is the zone where the highest insect activity occurs), as well as some that emerge under the trap and some that climb up from the ground. The trap is essentially a vertical panel of fine dark netting and dark end panels that intercept flying insects. These move up towards a pitched and sloping roof of white tent material and are guided into a collection jar at the highest point (Fig. 1). Townes' (1972) full-sized design is just over 2 m long (Fig. 2).

The numbers of flying insects caught relate to their abundance. Hence rare species are not over-collected, and abundant species are not ignored. Huge numbers can be caught even over relatively short periods such as a week and, as only about one quarter of the New Zealand insect species have been described, the catch will often include new or unnamed species.

Malaise traps have been used to assess insect community richness in forests (Moeed & Meads 1987; Hutcheson & Jones 1999) and shrublands (Dugdale & Hutcheson 1997) and to study wasp numbers in mainland islands. Malaise traps collect a wide diversity of invertebrates that usually include moths, beetles, flies, wasps and bees, as well as adult aquatic insects.

How to set a Malaise trap

Malaise traps are usually set on reasonably flat and clear areas of at least 2 m × 1.5 m. The peak of each trap is set pointed toward the light (i.e. northward) and the bottom edges are pegged touching the ground. The corners of the roof are tied off to pegs or trees and two tent poles may be required if there are no suitable trees nearby to keep all surfaces taut.

Figure 2. A full-sized Malaises trap is just over 2 m long (Townes 1972). The photo shows how the vertical panel is in contact with the ground, enabling ground-active invertebrates to climb the netting and be collected at the top of the panel.

Malaise traps can also be set above ground, and the beetles trapped in native forest canopy usually form a subset of those caught in adjacent ground-based Malaise samples (Hutcheson unpublished data). When set above ground the Malaise trap is held in place by a frame.

If trapping in an exposed site, stronger netting and a frame is necessary (e.g. Faulds & Crabtree 1995). Using smaller Malaise traps is discouraged because samples from them are not as useful for comparative studies (Dugdale & Hutcheson 1997).

The simplified collecting attachment described by Hutcheson (1991) (Fig. 3) has since been modified to use a collection jar of the same dimensions as those used to construct the attachment. Larger jars are also available which fit the same lid. Half fill the collection jar with 70% ethanol and use either larger collection jars or propylene glycol if the trap is to be left unattended for more than 2–3 weeks.

Figure 3. Detail showing the collection container and its attachment. This attachment was made by cutting a hole in one side of a second collecting container and gluing on a funnel formed from a cut-off wide-mouthed drink bottle. Two lids with large central holes glued and clamped together back-to-back form the screw-top attachment for the collecting container. Note that blue polypropylene glycol, not ethyl alcohol, is used as a preservative in this collecting container.

When and how long to set Malaise traps

Malaise traps may be used over extended times provided they are emptied weekly. They are useful for long-term studies such as monitoring seasonality of species, or insect groups or communities (e.g. Moeed & Meads 1987; Hutcheson 1990). However, the large numbers of insects caught even when a Malaise trap is set for only a week means that only species or groups of interest are usually sorted out and if the entire catch is of interest then subsamples may be taken for analysis.

The best period for using Malaise traps for inventory purposes is to take four consecutive 7-day samples in December. This is the early part of the main adult insect activity period in New Zealand, and it is also when the greatest range of beetle species are caught (Hutcheson 1990; Hutcheson & Kimberley 1999).

If it is essential to use Malaise traps for single species work then the by-catch should be minimised by only setting the traps when and where the target species is active and if necessary reducing the collecting time.

Sample collection and storage

Obtain the sample by unscrewing the sample collection container from the Malaise trap and straining the specimens out by pouring the sample through Chux multicloth in a small funnel. Use the strained ethanol (or propylene glycol) to wash out the collection jar and repeat as required to retrieve any remaining specimens. Alternatively, top-up the collection jar ethanol (to prevent the sample from 'sloshing' around, which will damage the specimens) and replace the collection jar with a spare.

We recommend collecting the samples each 7-day period. Thus 4 × 400–500 mL (c. 2 litres), of 70% ethanol is required per trap for a 4-week inventory sampling period. For monitoring, use the same collection fluid (70% ethyl alcohol or propylene glycol) each time. All samples and any parts of samples (together with the collection label) should be stored in undiluted ethanol (Walker & Crosby 1988). Where the specimens will not be used for DNA analysis (e.g. to be pinned and dried later) use 70% ethanol and add a drop of vinegar (after collection) to retain specimen flexibility for taxonomic purposes (Klimaszewski & Watt 1997).

Preliminary sorting

Wash the specimens out in water in a white-bottomed plastic tray with suitably high sides. Use fine forceps (tweezers) to pick out the target specimens. You may need soft-nosed ('feather-lite') forceps or a small paintbrush to transfer small fragile insects within a drop of water. A low-powered (c. 10 ×) magnification aid is helpful for small insects. Transfer the specimens into pure ethanol in suitable separate small containers or glass vials, one for each group of interest. Additional information for sorting samples is available in 'Preliminary sorting of invertebrate samples' (doccm-388193). When you have finished sorting out the specimens you can keep them in the vials (expensive) or transfer them, together with some undiluted ethanol and each with a separate label, into small zip-lock plastic bags. Include a new specimen label (see 'Minimum attributes') and a few mL of undiluted ethanol in each bag. The bags are then sealed into screw lid plastic jars containing some undiluted ethanol.

Identification

This is usually done by an expert, but you can do some preliminary sorting if you have the skills. A list of identification aids is provided in 'Invertebrate identification aids' (doccm-388198). However, it

is important that you also keep all the other specimens you cannot or do not sort in case these are required later for checking by experts.

References and further reading

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Appendix A

The following Department of Conservation documents are referred to in this method:

doccm-525907	Introduction to statistical analysis of invertebrate monitoring data
doccm-388198	Invertebrate identification aids
doccm-2686377	Invertebrates: advice and diagnostic support
doccm-388193	Preliminary sorting of invertebrate samples
doccm-359575	Vegetation: RECCE plots
doccm-146272	Standard inventory and monitoring project plan