Invertebrates: pitfall trapping

Version 1.0

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Contents

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Synopsis

Pitfall trapping is commonly used for capturing invertebrates that are active on the ground. As the name suggests, the trap works on the principle that an invertebrate moving on the ground simply falls into an open (usually circular) container dug into the ground. The invertebrates are usually killed in a preservative solution and are taken back to a laboratory for later sorting and identification. Pitfall traps are generally unspecific in what they capture, although they can sometimes be modified to targeted specific groups (e.g. by baiting). Typically, large numbers of invertebrates are caught in any one session—usually large-bodied, mobile species such as predatory ground beetles (Carabidae), rove beetles (Staphylinidae), wandering spiders (e.g. lycosids, clubionids) and ants. Pitfall trapping measures the activity of species that may be susceptible to capture in pitfall traps. It can be used to estimate an index of relative abundance from one time to another or between different places. Such indices have different relationships to the actual abundance of different species because they are influenced by a huge range of factors such as meteorological conditions, time of year, sex and developmental stage of the invertebrate, etc. As a result, when pitfall traps are used to monitor changes in numbers of individuals then each species should be treated individually (i.e. such studies should be intra-specific). The technique is also sensitive to variations in how the traps are set so the quality of the data depends on paying attention to the details of how to set the traps (e.g. the diameter of the pitfall trap affects the size and number of invertebrates caught; lips of traps left projecting above the soil surface will reduce catch rates).

Pitfall trapping is suitable for studies of community richness (number of species present) or habitat assessments of mainly surface-dwelling invertebrates, but be aware that other trapping methods may reveal an entirely different range of invertebrates at a given locality. Pitfall trapping has been extensively used in forested habitats in New Zealand, whereas elsewhere it has been considered ideal for open habitats such as grasslands and arable land. There is extensive literature and the reader is directed initially to New (1998), Southwood & Henderson (2000), and Leather (2005) for reviews. Two case studies (Pawson et al. 2008; Sinclair et al. 2005) are provided to demonstrate the use of pitfall traps to examine invertebrate fauna. Other examples that include distributions are studies by Stevens et al. (2007) and McCartney et al. (2007), and a good example of an inventory study is outlined in Winks et al. (2004).

Pitfall trapping has been used in the following types of studies:

- Investigating community structure
- Habitat associations—which species may be found in various vegetation communities
- Activity patterns of individual species or pre-determined groups of species
- Mapping spatial distribution
- Total population estimates (possibly coupled with another technique such as mark-recapture and necessarily using a live-capture trap)
- Pest monitoring programmes (e.g. invasive ants)
- Studies which require sampling/monitoring species that occur at low densities

- Comparative studies of relative abundance (though note the caution regarding this application and the possible implications for using pitfall trapping in monitoring studies which seek this information)

Assumptions

Explicit assumptions that relate to pitfall trapping depend on the details of the study in question and may include the following:

- The target species is/are catchable using pitfall traps.
- The capture of one animal will not interfere with the capture of another.
- There is a direct (but unknown) proportional relationship between the numbers of a particular invertebrate species caught in a pitfall trap and the abundance of that species that are active in the environment.
- Every trap is equally effective at trapping if set in the same position.
- The design and method of laying out traps does not compromise the 'trappability' of the target species.
- The effectiveness of the sample design or layout is the same between each sampling event.
- The proportionate relationship (described above) will not change with time.

Advantages

- Pitfall traps are simple, easily transported, erected and serviced.
- Easily repeatable between studies and over time.
- Cost-effective in terms of materials, labour and skills required.
- Pitfall traps can catch a large number and wide diversity of primarily ground-active invertebrates.
- Specimens are usually caught undamaged and are therefore easy to identify.
- Pitfall trapping produces large amounts of data.
- Pitfall traps can be set for long periods if necessary.
- Pitfall traps do not require a power source.

Disadvantages

- Not suitable for catching most flying or arboreal invertebrates, and of limited use with rare ground-dwelling species unless live-capture traps are used.
- Not suitable for long-term studies as the traps can reduce the overall abundance of invertebrates in the local area.

- Occasionally, pitfall traps catch lizards and frogs and should not be used in areas where threatened species occur.

- Pitfall traps are not suitable for use in areas where weka are present or in flood-prone areas (see 'Case study B').

- Except for relative abundance, the method is not usually suitable for estimating other population parameters (such as survival, reproductive rate, fecundity, lifespan, etc.).

- The catch is affected by the time of year, meteorological conditions and the particular micro-habitat characteristics where they are situated.

- Numbers caught can also be influenced by the attributes of the catchable invertebrates (such as population size, relative abundance, sex ratio, developmental stage, etc.).

- The relative abundances of species caught in pitfall traps differ from their actual relative abundances in the habitat because species vary in how trappable they are. However, see Woodcock (2005) about minimising this shortcoming.

- Studies involving baits with pitfall traps are most suitable for qualitative studies such as determining which species are associated with particular habitats or for making species inventories because of the exaggerating effects attractants have on the numbers caught.

- The materials used to make covers over pitfall traps may introduce a source of bias (e.g. covers that prevent letting light through). This may be attractive to species that hide during the day and a detraction to others that are active in bright daylight. A more neutral approach is to use a clear cover which lets light through. Despite this, most studies in New Zealand have used opaque covers.

- A significant (and difficult to manage) problem is the relationship between increasing structural complexity of vegetation or habitat and capture rate—it is possible that catch rate may be reduced because dense vegetation dilutes numbers or impedes the free movement of individuals.

- The data collected from pitfall trapping are typically fraught with problems when it comes to statistical analysis—despite their apparent suitability. See a review on the subject by Spence & Niemela (1994).

Safety issues

Ethyl alcohol (ethanol):

- Poison: Treat ethyl alcohol (ethanol) 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 (ethanol) is highly flammable (both liquid and as vapour).

- Do not use ethylene glycol. It is not environmentally friendly and may impose health risks (Cresswell, 1995).

- Propylene glycol antifreeze **should** be used as a substitute for iso-propyl alcohol.

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 city council disposal site

Suitability for inventory

Pitfall trapping is suitable for inventory of ground-active invertebrates but not for other invertebrates.

Suitability for monitoring

- Pitfall trapping is not suitable for measuring abundance indices over short periods of time because it is primarily an activity measure.
- It may be used to obtain an approximate index of relative abundance if used in the same way, in the same location(s), at the same time of year and if set over a long period of time.
- May be used for monitoring relative changes in species composition between sites and over time. However, these changes are indices and do not reflect the actual relative abundance of species in the habitat.
- There is the potential to use a customised trap or special location to target single species or few species if it is known that the trap will efficiently capture these species without killing them (such as using a pheromone lure).

Skills

Using pitfall traps in the field does not require highly specialised skills. As a minimum an operator needs to be able to record accurately the location of the traps with GPS equipment and log these and capture data onto a digital file (using software such as Excel) for subsequent analysis (which would probably be done by someone with specialised statistical skills). Establishing the traps in the ground is simple, although it is essential to pay attention to details and follow a standard procedure strictly (such as placement of the trap in the ground and recording sample data methodically and accurately). Sorting and identifying captured animals (even to recognised taxonomic units—RTUs) requires highly technical skills which, typically, will require outsourcing.

Resources

One person is required with the relevant skills to set up and service pitfall traps, although a second person can be advantageous when a large number of traps are being used. In calculating the time required, take into account the return travelling time to the site, and add about half an hour to set up a set of five pitfall traps on a 5 × 5 m grid (one trap in the centre). If the substrate is difficult to dig (e.g. stony or compact soil), allow extra time to establish the traps. Allow about 2 minutes per trap to collect the samples and either reset the traps or remove them.

What you will need for pitfall trapping

- Notebook and pencil to record date, trap identification number and position, and note any other relevant details such as how to find the trap again.

- Permanent marker pen to write identification numbers onto traps.

- Flagging tape to help find the trap again.

- GPS (preferably one that works in the bush; an external aerial may be required) to obtain precise locations for the traps. Try to obtain positions with errors less than 10 m.

- Hand trowel to dig holes for the traps.

- Container of antifreeze (use undiluted propylene glycol) to preserve anything that falls into the trap. You will need about half a beer cup full per trap.

- Materials for pitfall traps (lengths of downpipe, beer cups, metal covers). See materials in the following list.

Materials required for each pitfall trap

- 170 mm length of standard 80 mm outside diameter white plastic downpipe with about six holes about 1 mm in diameter drilled about 60 mm from the top. This forms the pitfall trap hole (drilled holes allow excess water to drain and prevent 'floating' the trap out in excessive rain).

- Clear plastic beer cup or similar about 130 mm long and 76 mm outer diameter that fits snugly within the downpipe. This collects insects that fall into the downpipe.

- Aluminium cover (about 250 mm square) with corners bent down 30–40 mm to prevent rain and debris from entering the pitfall trap.

- Approx. 50 mL antifreeze—undiluted propylene glycol. This preservative is non-toxic and environmentally safe.[1]

- About 1 teaspoonful of table salt as an added preservative in case the antifreeze is diluted by rain.

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[1] Propylene glycol is a less common antifreeze than ethylene glycol. Both cost about the same. Propylene glycol is an environmentally friendly antifreeze. It can be disposed of by tipping down the pitfall trap hole whereas ethylene glycol is not environmentally friendly and should be taken to a city council disposal facility.
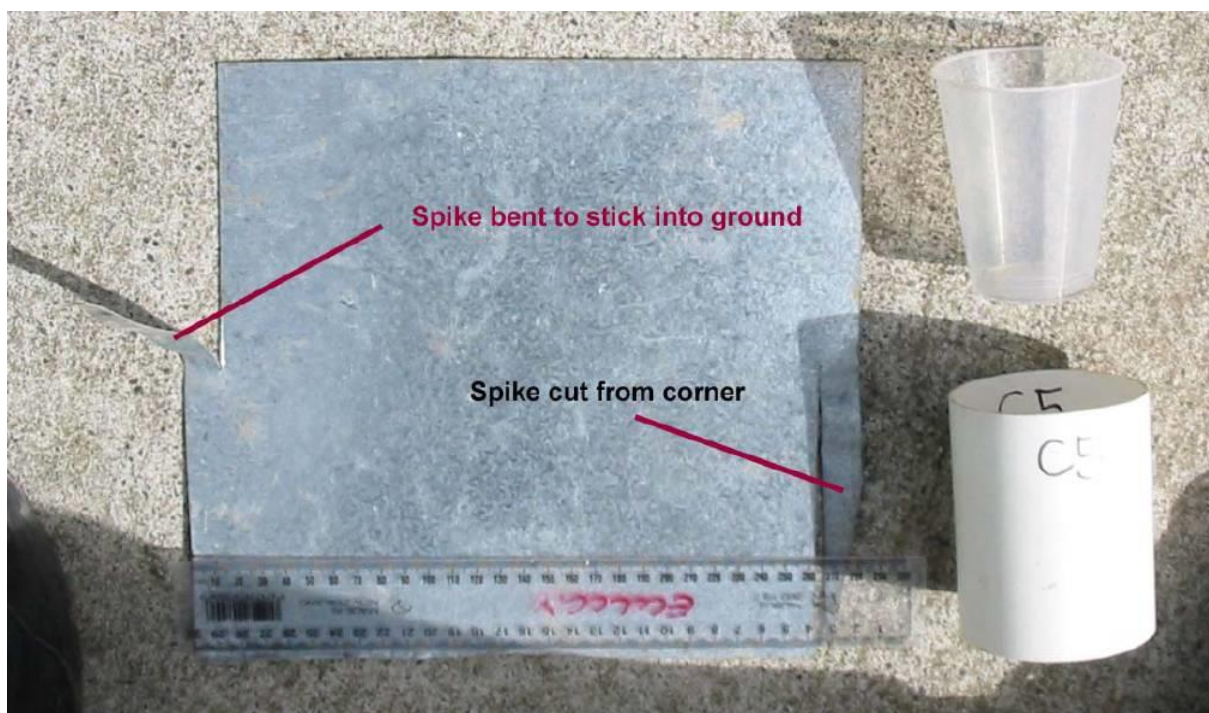
Figure 1. Materials for a pitfall trap (ruler included as a scale only).

The materials required for establishing pitfall traps in the field need to be scaled up depending on the size of the project. Economies of scale regarding costs and using standard materials should result if others in DOC are using the same standard method. Use Table 1 for costing out the materials. In addition, you will need to get an estimate for the cost of sorting the samples if you are using a contractor.

Table 1. Materials purchase list.

Enter number of pitfall traps required here $\longrightarrow$		$\longleftarrow$ **this is** Z **below**
Item	**Number/quantity to purchase***	**Cost**
Antifreeze (propylene glycol)	25 mL × Z = mL	
Ordinary table salt	0.20 g × Z = g	
165 mm lengths of 80 mm diameter plastic downpipe	Z / 18 = 3 m lengths	
Plastic cups that fits snugly into the downpipe	Z =	
Trap covers (250 mm squares of aluminium or galvanised iron sheet)	Z =	
Sample containers (100 mL clear square-sided polycarbonate jars)	Z + 1 =	

Chux multicloth	Z / 8 = sheets	
Small plastic funnel	1	
'Laundry-tag' paper	Z / 30 = sheets	
Undiluted alcohol (or methylated spirits)	Z × 50 mL = mL	

* '×' indicates multiply; '/' indicates divide

Materials for collecting samples

- A sample container (100 mL clear square-sided polycarbonate jar) for each trap. Pre-pack each sample container with a multicloth square and a label. Note: check the weave and porosity to make sure captured invertebrates will not pass through.

- A square of Chux multicloth (c. 100 × 100 mm) for each trap for straining the insects out from the preservative. Each should be large enough to fit comfortably in the collecting funnel without folding when the liquid is poured in.

- One spare large (e.g. 200 mL) sample container to collect the antifreeze when straining the insects out.

- A small plastic funnel for holding the Chux multicloth while straining out the insects.

- One label (c. 3 × 7 cm) of 'laundry-tag' paper label for each trap, and one pencil. 'Laundry-tag' paper does not fall apart in most solutions—waterproof notepaper eventually falls apart. Do not use a pen or marking pen as the ink will dissolve.

- Undiluted alcohol (you can use methylated spirits), about 50 mL for each trap, for preserving the samples. Note: the volume of preservative spirit to that of captured invertebrates should be about 3:1.

- Indelible marking pen for labelling the lids of the sample jars after the samples are in them.

- Fresh 80% antifreeze (100 mL per trap) and table salt ONLY if you are resetting the traps.

Materials for removing traps

- Everything for collecting samples (see previous list) except you will need no spare antifreeze

- A large watertight plastic bag (optional) to hold the beer cups (these will dribble propylene glycol)

- A pack or bag (optional) to carry dismantled traps out if you have far to walk

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]

For each site, record:

- Site name
- Brief explanation of what the study is for
- Range of pitfall trap identification numbers
- Experimental design (layout, trap groupings if any, etc.)
- Start and finish dates

For each pitfall trap, record:

- Trap identification number
- Date collected
- Name of collector
- GPS coordinates

Optional attributes include taking photographs of the sites and any other qualitative information that may influence the sample (e.g. vegetation description, flooding, interference by mammals or people).

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.

Once the sample has been sorted, data for each species or RTU are entered into an Excel spreadsheet using the following suggested fields:[3]

- Person who collected the sample
- Location of trap
- GPS coordinates (eastings and northings in separate columns)
- GPS estimated accuracy (if available)
- Altitude (from GPS)
- Trap number (individual identification number for trap)

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

[3] For an example spreadsheet, see 'Example data sheet invertebrate monitoring' (doccm-428226) http://www.doc.govt.nz/documents/science-and-technical/inventory-monitoring/invertebrate-monitoring-datasheet-example.pdf

- Order

- Family

- Species/RTU

- Number of individuals

- Sampling series

- Date trap set (opened)

- Date the sample was collected

Note: considerable taxonomic revision will occur over time because taxonomic revision is a standard part of all entomological studies. The creation of a 'look-up' table in Excel may ease this process.

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.

Ultimately insects collected for inventory 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.

DOC staff should consider entering location records (GPS coordinates) of identified species into the BioWeb database.

Analysis, interpretation and reporting

Introduction

The following outline is intended to highlight some of the practical considerations of dealing with pitfall trap data and provides an overview of the types of analysis that may be appropriate. Once the data have been collected and saved into a spreadsheet (see 'Example data sheet invertebrate monitoring'—doccm-428226)[5], they should be summarised and presented either in a table or graphically. Basic data summary statistics and an overview of the common types of analysis used

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

[5] http://www.doc.govt.nz/documents/science-and-technical/inventory-monitoring/invertebrate-monitoring-datasheet-example.pdf

for invertebrate data (with worked examples) are provided in the 'Introduction to statistical analysis of invertebrate monitoring data' (doccm-525907).[6] It is recommended that advice from a statistician or biometrician be sought at the start of projects to check that the study design is appropriate for the hypothesis being tested, and again when a basic summary of the data is complete. It is also important to seek statistical advice from a biometrician or suitably experienced person prior to undertaking any analysis. The information provided in this section and in 'Introduction to statistical analysis of invertebrate monitoring data' (doccm-525907) 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

Pitfall trapping has been widely used to monitor invertebrates in New Zealand (Moeed & Meads 1985; Berndt 1998; Green 2000; Norbury et al. 2009) but the data collected are a reflection of invertebrate density, activity and coincident environmental conditions (Uetz & Unzicker 1976). The method is useful for comparing relative abundances of invertebrates between sites but not between species because some species are more active than others and this leads to bias (Dufrêne & Legendre 1997). Normally, the first collection from pitfall traps is discarded because of the 'digging in' disturbance effect. However, this should be retained when pitfall traps are used for inventory.

Other considerations when interpreting pitfall data include the size of the pitfall traps, the surrounding vegetation and the biology of the target species (Spence & Niemela 1994). Assuming that a study has been designed to minimise both biotic and abiotic bias, and that any bias is constant between experimental sites, this trapping method can be successfully used to compare spider communities (Berndt 1998) as well as other ground-dwelling groups such as beetles, ants, millipedes, centipedes and ground-wētā. Pitfall traps are not suitable for long-term repeated sampling since there is a possibility of reducing the local population of invertebrates (Southwood & Henderson 2000).

Analysis of pitfall trap data

The statistical methods used with pitfall traps depend on the hypothesis 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:

- 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 used.

- At the time studies are designed, decisions need to be made regarding which key groups are going to be analysed. Some invertebrates such as ants are aggregated in groups (or colonies) and this may influence how your study is designed. It is common to place pitfall traps in groups of four or five, and when the data are summarised/analysed, the results from these traps are combined and treated as one independent sample giving a single data point.

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

- 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.

- If the traps have been used to assess a particular group of insects occupying an area, it may be possible to summarise the data in terms of functional groups. This can provide important information about the role of the insect community in the local environment.

Once the data have been summarised there may be trends in the data that warrant further investigation. Statistical tests for unreplicated studies (e.g. one treatment group and one control group) will be limited to testing whether one group of data differs from another, whereas replicated studies (e.g. three treatment groups and three control groups) allow for more sophisticated tests that enable you to determine whether treatments differ from each other and to what extent they differ. If information regarding associated environmental variables has also been collected quantitatively, it will be possible to explore the data for relationships with environmental variables using multivariate analysis techniques (see 'Exploring data for relationships' in 'Introduction to statistical analysis of invertebrate monitoring data'—doccm-525907)[7]. 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.

Case study A

Case study A: understanding beetle diversity in heavily modified landscapes

Synopsis

Pitfall trapping can provide a consistent, and therefore reliable, method for accurately assessing insect assemblages. Pawson et al. (2008) demonstrate how pitfall trapping can be used to understand beetle communities over a range of exotic habitat types.

Habitat loss and fragmentation are widely recognised as causal factors in species decline. In some regions of New Zealand over 90% of the original forest cover has been removed and significant changes in land use are still occurring (Ewers et al. 2006). Approximately 25% of New Zealand's total forest cover includes exotic plantations (primarily *Pinus radiata*) (New Zealand Forest Owners Association 2005), although exotic forests have similar characteristics to native forests when compared with pastoral farming—the dominant feature of the New Zealand landscape. Exotic forests are therefore increasingly recognised for their potential contribution to conservation in terms

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

of preservation of forest biodiversity. Plantation forests can also contribute to the maintenance of ecosystem integrity by buffering native forest remnants from external influences.

Objectives

The objectives of Pawson et al. (2008) were to use pitfall traps to determine the role of non-native plantation forests in heavily fragmented landscapes as alternative habitats for native species. Two specific aims were proposed. First, beetle diversity was used to compare native forests, mature production forests, recently clearfell-harvested production stands, and pasture across the highly fragmented landscape of the central North Island. A second aim was to sample using pitfall traps along gradients between habitats in order to understand edge-mediated changes in the abundance of beetles (Fig. 2).

Sampling design and methods

Sampling design and study sites

Pitfall traps were laid along three independent edge gradients in each of the six possible comparisons between pairs of the aforementioned habitats. This gave a total of 18 edge gradient sites (i.e. pitfall traps were laid at three independent sites where native forest merged into exotic forest, three independent sites where native forest merged into pasture sites, and so on). Along each of the 18 edge gradient sites, single pitfall traps were set at seven distances ranging from one habitat into the next. These were set at −125, −25, −5, 0, +5, +25 and +125 m perpendicular to the margin. Point 0 refers to the central point between the two compared habitats. A total of 126 pitfall traps were used and these allowed comparisons to be made within and between sites as well as within and between habitats. Pitfall trap samples were collected six times at monthly intervals between November 2002 and February 2003 and between December 2003 and February 2004.

Figure 2. Beetles, such as this lucanid (*Geodorcus helmsi*), are abundant in native forests and can be used as 'quality' indicators for studies using pitfall traps.

Trapping methods

Pawson et al. (2008) used a standard pitfall trap design which consisted of a circular 680 mL polypropylene plastic container of 100 mm diameter. The container was buried so the lip of the container was flush with the surface of the ground. In an attempt to increase trap catch area by channelling invertebrates into the pitfall trap, two white plastic guide panels, measuring 1.2 m long and 10 cm high, were placed cross-wise on the ground and centred above each collection container. Antifreeze solution (70% monoethylene glycol) was used as a preservative and this was replaced approximately every month when samples were collected. During sample collection, samples were transferred from the container into 70% alcohol and the traps were reset.

Beetle diversity and identification

Beetle diversity was assessed by using three families—ground beetles (Carabidae), chafer beetles (Scarabaeidae) and bark beetles (Curculionidae: Scolytinae)—because these taxa reflect habitat use and they were collected in sufficient numbers for statistical analysis. Beetles from these families also provided greater trophic breadth than relying on single invertebrate groups—Scarabaeidae are herbivores, Carabidae are generally predators and Scolytinae feed on wood. All beetles from these groups were identified to species and stored in 70% alcohol. The exotic status of carabids was determined using Larochelle & Larivière (2001), and the exotic status of scarabaeids and scolytines was determined by the authors.

The number of individuals of each species captured was used in estimated species richness, and the capture rates of each species was standardised by converting to catch per 100-trap-days. This was done to account for variations in pitfall trap exposure durations.

Measuring environmental variability

Importantly, in order to compare beetle diversity with native and exotic habitat gradients, Pawson et al. (2008) had to accurately measure the degree of actual environmental changes in each chosen gradient. For this Pawson et al. (2008) measured abiotic factors, understory vegetation, ground cover complexity and the proportion of native and exotic plantation forest at each site centred over each pitfall trap. Underlying abiotic variables (climate, soil and slope) for each pitfall site were obtained from the Land Environment New Zealand (LENZ) protocol (Leathwick et al. 2002) and incorporated into composite variables in analysis of the beetle data. Understory vegetation surveys were also conducted within a 2.5 × 2.5 m quadrat centred on each pitfall trap, and relative plant abundances were quantified in four vertical strata for each plant species (for details see Pawson et al. 2008). Resulting scores, after principle component analysis, provided a measure of the understory vegetation community associated with each pitfall trap. These were included as environmental variables associated with the relative abundance data of the beetles. Estimates of ground cover complexity around each pitfall trap were made concurrently with the vegetation surveys using a five-point qualitative scale and were then incorporated into the analysis.

Pawson et al. (2008) also measured the proportion of native and exotic plantation forests. This was done by using data from the Land Cover Database V2 (LCDB2) (Terralink 2004) within a 500 m, 1000 m and 5000 m radius of the centre of each pitfall trap. The proportions calculated were then included as explanatory variables in the analyses.

Results

With regard to the first aim of Pawson et al. (2008)—to determine the difference in beetle diversity across four habitats (one native and three exotic)—clearfell habitats had the greatest beetle species richness and native forests had lower species richness than any production habitat. The main cause of this seemed to be the low level of exotic beetles found in native habitats (i.e. high numbers of both native and exotic beetles were found in exotic plantations), and to the fact that the abundance of native species was relatively uniform across all habitats. Recent clearfell areas had the highest relative beetle abundance of all four habitats, and this was largely due to the presence of one native tiger beetle species (*Cincindela tuberculata*).

The second aim of Pawson et al. (2008) was to assess beetle diversity and abundance across habitat gradients in order to better understand edge-mediated effects of exotic and native habitats on insect diversity and abundance. All four habitat types formed statistically distinct groupings from each other and 12 of the 29 environmental variables tested were significant predictors of variation in beetle community composition between sites. Variations in beetle species composition were best explained by the proportion of native forest within 500 m of the sample location followed by understory variation. Variation in native beetle species between native areas was relatively uniform so it was not surprising that Pawson et al. (2008) found relative beetle abundance did not change

with respect to distance into each habitat. There were marked differences in beetle composition between habitat types but this was dependent on the adjacent habitat type and not distance from the habitat edge. There was little invasion by exotic species into native forest habitats even when adjacent areas were exotic. However, there was evidence that exotic beetles were encroaching on pasture habitats. Mature forest also had a much lower abundance of exotic beetles than pastoral grassland even when pastoral land was directly adjacent to recent clearfell.

Limitations and points to consider

Pawson et al. (2008) found that species groups of the three families assessed through pitfall trapping were well characterised by the level of sampling effort employed (127 traps collected six times over 2 years). With reference to future pitfall trap studies these results are important for at least two reasons. First, relatively few pitfall traps may be needed in a study to obtain well-resolved results on species diversity and abundance. Second, very few invertebrates need to be sampled and identified in order to obtain well-resolved environmental patterns. Major limitations to large pitfall surveys often include the large amount of time required to service both the traps and process the invertebrates caught in them. Often expertise is not available for determining all of the vast number of organisms that can be caught throughout a pitfall trap season. Limiting both trap number and the number of taxa that need to be assessed may greatly reduce the expense and duration of a study making this method more practical for determining a wider range of questions relating to conservation and ecology.

It is widely recognised that pitfall trapping has inherent biases (Ward et al. 2001). Capture rates in pitfall traps cannot necessarily be used as direct measures of local population density nor can it be assumed that the area sampled between traps has the same size. Variations in local invertebrate densities and wider-scale invertebrate activity along with trap position and variations in the surrounding environment influence the catch rate of the trap and, as a result, bias results in an unpredictable manner. Pawson et al. (2008) countered this by using 'abundance' to refer to activity-density, i.e. relative number of individuals present in a pitfall trap in a given habitat. Furthermore, conclusions regarding changes in relative abundance of species refer to changes in the relative activity-density of different species, rather than to identifiable changes in local population density. Another way of reducing variation due to variations in activity levels of the organisms is to set and service the traps at the same time so all the traps experience similar meteorological conditions. It is also important to remember with pitfall trapping, as with all passive trapping systems (e.g. Malaise, pitfall-flight-intercept), that true measures of population density are difficult to ascertain.

A last point to consider, one that is highlighted well in Pawson et al. (2008), is that pitfall traps catch and kill most (if not all) vagrant ground invertebrates in large numbers. This is not an ideal situation when few species are targeted or when there is a large likelihood of catching endangered species (possibly in large numbers). Collecting non-target invertebrates is in many cases superfluous to answering specific environmental questions and could, in some cases, have large deleterious effects on the local invertebrate fauna.

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Case study B

Case study B: effects of rat eradication on invertebrate assemblages from Kapiti Island, New Zealand

Synopsis

In this example Sinclair et al. (2005) show how pitfall trapping can be a major tool in understanding invertebrate population changes over time.

Kapiti Island (Fig. 3), is a large (1965 ha) nature reserve, and has been the focus of intense restoration over 60 years. In 1996 the last two exotic pests, the Norway rat (*Rattus norvegicus*) and Pacific rat (*R. exulans*) were removed. Resident invertebrate fauna were expected to benefit from removal of rat predators.

Invertebrates make up a significant component of a rat's diet, yet the benefits to invertebrate communities following rat eradication are either anecdotal or inferred. Predator removal on islands is usually followed by an increase in insectivorous birds (Miskelly & Robertson 2002; Towns & Broome 2003), yet few, if any, studies have documented the relationship between invertebrate densities and bird assemblages. The Kapiti Island rat eradication programme therefore provided the

perfect opportunity to employ pitfall trapping methods in order to understand the effects of predator removal on invertebrate communities.

Pitfall traps were chosen because they were cheap, portable and gathered a large range of invertebrates most likely to have been depleted due to the presence of rats (Fig. 4).

Objectives

Sinclair et al. (2005) had four main objectives:

1. Compare invertebrate community composition and capture frequency before and after rat eradication, serving as a baseline study for future research.
2. Determine whether future sampling could be simplified by using higher taxonomy as a surrogate for more specific identification.
3. Assess which techniques were most effective at summarising data in an ecologically meaningful manner.
4. Relate changes in invertebrate communities to studies on changes in bird populations.

Sampling design and methods

Invertebrates were sampled over 5 years from April 1995 until June 1999, using standard pitfall traps. Traps were set on 14 occasions; five before and nine after rat eradication. Sampling periods were classified as follows: First year = April/May, and May/June 1995. During the next 4 years Sinclair et al. (2005) took a cycle of three samples representing summer (November/December), autumn (February/March) and winter (May/June), finishing in the winter of 1999.
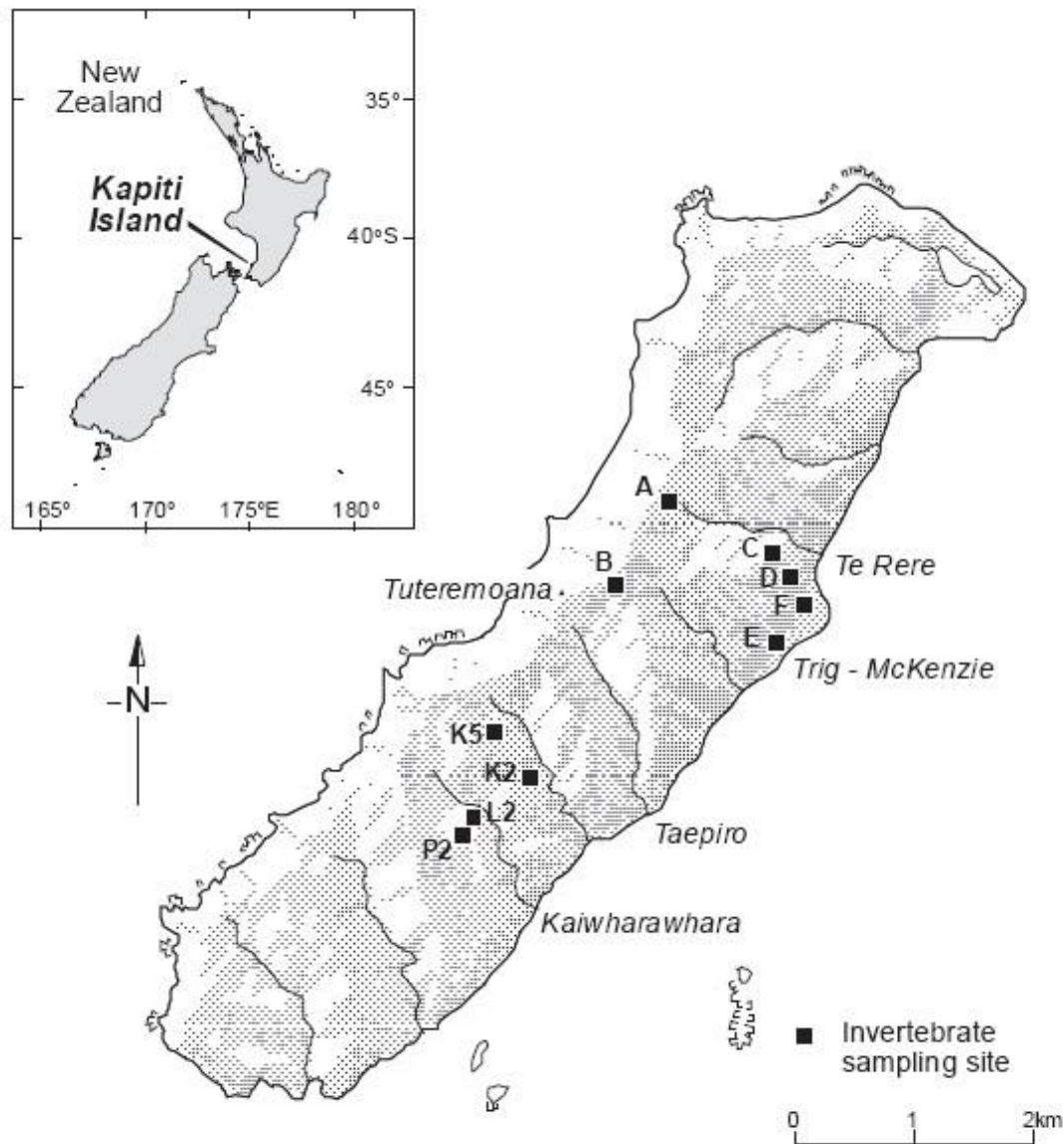
Figure 3. The 10 pitfall trap sites located along old possum-trapping lines and catchments of Kapiti Island, New Zealand (with shading to accentuate relief).

Ten pitfall trap sites were chosen. The sites ranged in primary vegetation from kānuka, mānuka, kohekohe, coastal māhoe and tawa forest to bracken fern and grassland. Each site comprised 10 randomly located traps at least 1 m apart. Traps were 7 cm diameter (8 cm deep) plastic cups that fit snugly within a plastic retainer tube. They were covered with a square 30 cm^2 plywood 'lid' staked 2 cm off the ground with wire. The cups were 1/4 to 1/3 filled with Galt's solution, exposed for 1 month and then either reset for another month or closed by filling with debris and pushing the lid down, until the next cycle. On collection, the whole contents of the trap were transferred to a water-tight container and labels were written on both the outside of the container and on waterproof paper that was inserted into the container to avoid any later misidentification. Soon after, the contents of the container were stored in 70% ethanol.

Specimens that were longer than 5 mm were sorted into morphospecies (RTUs) using a wide array of published keys, reference books and specialised advice (for details see the methods of Sinclair et al. 2005).

Various statistical techniques were used to explore the data, such as simple abundance, species richness, evenness and diversity indices. Taxa were also lumped into higher taxonomic groups to see if this could mimic more detailed species data. Vegetation type, season and year were also considered with analysis of variance (ANOVA), ordination, cluster analysis and data splitting.

Figure 4. 'Big eye' harvestmen belong to the widespread Opilione group that are abundant and readily caught in pitfall traps.

Results

There were 366 RTUs recognised, with 28% keyed to genus level. In general there was a significant decrease in invertebrate catch frequency and diversity, most obvious in the Carabidae and Amphipoda following the mammal pest eradication. However, results varied depending on the statistics used. After eradication, two of the three richness indices, and one of the two diversity indices, showed a significant decrease, and evenness measures showed a significant increase. Trends seemed gradual and not immediately detectable after eradication and, as expected, there was a strong influence of site, season and year on the variation within the data.

In answer to the second objective, Sinclair et al. (2005) found that finely classified taxa revealed very similar trends to the seven higher taxonomic groups. However, the richness scores obtained from finer levels of classification were interesting for ecological interpretation, and these required sorting to morphospecies, which took considerably more time (months).

The third objective revealed that exploratory techniques of data splitting, the presence and absence of taxa after eradication, and cluster analysis did not improve the ecological understanding of the data in Sinclair et al. (2005). Of all the analysis techniques used, the ANOVA modelling with comparisons using Akaike's information criterion (AIC), ordination and standard richness, evenness and diversity indices were the most interpretable.

Limitations and points to consider

Pitfall trap sampling is a passive technique that can trap a vast number of species across a wide range of taxa. While this may be important in larger studies aimed at understanding long-term trends, this can present logistic problems such as processing time. Sinclair et al. (2005) avoided these problems to a large degree in three ways: firstly, invertebrates that were smaller than 5 mm were ignored; secondly, identification to the species level was for the most part avoided by classifying invertebrates into recognisable groups; thirdly, results were examined using 'morphospecies' and RTUs, which may allow examination of the data with respect to trophic structure—often much more useful in ecological terms. Furthermore, Sinclair et al. (2005) examined the data at high and low levels of taxonomic identification giving support to the idea that an understanding of diversity and variation at the species level is not always necessary when understanding long-term population trends.

A small point mentioned in Sinclair et al. (2005) is that some samples were destroyed by weka (in four sites; p. 296); it seemed that the weka had learnt to go from trap to trap pulling up the pegs used to stake down the lids, and started pecking at the bottom of the cups. This is not only frustrating for the researcher, but, depending on the preservation solution, is potentially harmful to the birds. This highlights the need for securing traps properly and shows that it is necessary to think of the terrain and wildlife that is present at each site before leaving traps exposed for any length of time.

The terrain and weather patterns meant that a dry powder of Galt's solution was far easier to carry and use than other types of preservative. It could be made up on-site with nearby water—and was therefore easy to carry across the rugged terrain. It could also withstand some dilution with rain and still preserve invertebrates. However, Galt's solution can damage insect cuticle, making them harder to identify (Brennan et al. 2005). Care is therefore needed when planning the location and preservative needed for ideal sampling. Factors to consider may include terrain, local wildlife, trap exposure duration, rainfall/humidity, etc.

It is important to consider confounding factors that may influence the overall community trends and include these in the sample programme. A control site (with no eradication) is impossible on an island destined for eradication. However, sampling a control site, or for a longer term, would have clarified short-term aberrations in longer-term trends due to El Niño/La Niña weather changes.

When 'restoring' an island biota, a distinct 'success' end point should be defined. It was clear on Kapiti Island that birds had replaced rats as insectivores, which may have suited conservation of bird species but not necessarily the conservation goals of the invertebrate fauna.

References for case study B

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Full details of technique and best practice

Pitfall trap design

The standard pitfall trap comprises a 165 mm length of plastic downpipe (80 mm internal diameter), a plastic cup usually containing a preservative fluid, and a cover to keep out rain. The downpipe is dug into the ground and soil packed around it so its rim is either level or just below the ground (1– 5 mm). If the lip of the downpipe projects, it will present a barrier to some species (especially small ones) and bias the capture. The cup should fit snugly into the downpipe so nothing can escape between it and the inner surface of the downpipe and yet be loose enough to be pulled out when the sample is collected. The cover sits 30–40 mm above the trap.

Different species respond differently to the physical makeup of the trap such as its size or shape and so it is important that identical traps are used throughout the study. The modal diameter of traps derived from literature is between 60 and 80 mm with a depth of 80–100 mm, hence the dimensions given above. There is, of course, an infinite variety of possible designs for pitfall traps but we suggest that the standardised design should be used except where there are specific reasons for varying it. For instance, when surveying for an invasive ant species, it will be more efficient to set a larger number of smaller traps rather than fewer standard traps over the same area.

An opaque cover over the pitfall trap (usually made of plastic or metal) keeps litter and rain out and gives the invertebrate the illusion of safe passage as it walks over the edge of the trap. Outside New Zealand a clear cover is often used, but this is not standard practice in New Zealand.

The trap itself is partially filled with a solution whose purpose is to drown and preserve the invertebrate. When the sample is collected, the contents of the trap are drained through a sieve and the specimens are stored typically in ethanol. Propylene glycol is most commonly used in pitfall traps as a killing agent and preservative but a variety of solutions are available depending on the requirements of the study. Considerations include avoiding the risk of attracting or putting off invertebrates with the smell of chemicals (such as alcohol). If this is considered a risk, a strong solution of table salt may be used with a little non-scented detergent (as a wetting agent) to help captives sink. If water alone is used, then the traps need to be cleared before the invertebrates decay. Occasionally pitfall traps are used to catch live invertebrates and then they are set without killing/preserving solution. For example, pitfall traps have been used to catch live ground beetles for a study on tracking their movements (Hutchinson 2007). This may result in some captives eating others, but it may not be a problem if the target species is a predator. If live traps are to be used then glass containers work best to prevent captives from climbing out. The risk of escape may be further reduced by using funnels. Also, of course, pitfall traps are checked as often as possible for live trapping to ensure that animals remain alive. Otherwise, plastic traps are adequate with killing solutions. Another modification is to bait traps to attract specific groups. This is a specialisation that is rarely employed and so is not explained further here. It requires knowledge about what attracts the invertebrate of interest. See Southwood & Henderson (2000) for further details.

Pitfall traps may be used in conjunction with 'drift fences'; these are baffles whose purpose is to intercept invertebrates and guide them to the pitfall trap. The first catch, after the trap is installed, may be higher than normal and this is often referred to as a 'digging in' or 'settling in' effect. As a result, this sample is usually discarded for studies involving comparisons between subsequent sequential samples as, for example, is done when samples are taken from pitfall traps one after the other to investigate what time of year a particular species is active.

Woodcock (2005) discusses ways of minimising the shortcomings of pitfall trap design. Some of these considerations include not using killing solutions which may attract particular taxa (e.g. antifreeze is commonly used and acts as an attractant to Carabid ground beetles), using specialised designs such as gutter traps (essentially elongate pitfall traps which will engage specimens over a wide frontage) and ramp traps for rocky surfaces. Sampling regimes may be modified to reduce variability such as extending the duration of the time the trap is set. For a detailed discussion of sources of error encountered in pitfall trapping see Adis (1979) who lists 18 main sources.

Sample design

Pitfall traps are laid out in a consistent pattern and opened for set periods before the samples are collected (usually referred to as 'clearing' the traps). For a detailed discussion of experimental design see Woodcock (2005). The standard spacing and layout is five pitfall traps on a 5 × 5 m grid with one in the centre. All five pitfall traps at this one site are a single sampling unit, although the

individual trap catches are kept separately. The number of sampling units and the experimental design should be discussed with a statistician before the study commences. Spacing and layout may also vary depending on the specific questions addressed by the study (Woodcock 2005).

Possible variations on how pitfall traps are set out in the field are almost infinite but common designs in published studies are 10, 20 or 25 pitfall traps set out on grids 1–5 m apart; lines of traps along transects or traps set randomly within a plot. Often, in New Zealand, pitfall traps are set in groups of five—a central one surrounded by four others 5 m apart, and the catches from each group are summed and used as a single sample. The number of traps or groups of traps necessary depends on the aim of the study and the level of accuracy required. A preliminary trial employing a power analysis will usually be required to determine the number of traps needed. Once set out, the traps are inspected at regular intervals (after a 'settling in' period has been allowed for)—up to 14 days between checks. When sampling is intermittent such as when yearly monitoring is required, the traps are often left *in situ* and then they are filled with sticks and leaf litter to allow anything falling in to escape until the traps are needed again. Finally, the design of pitfall trap studies should consider the risk of adversely influencing the species' populations. This is a risk for highly mobile species and others which may be particularly 'trappable' because they may be 'trapped out' from an area.

Establishing traps

Locate a suitable site for the trap

This should be level, but can be on a narrow ledge on a slope provided the ledge is wider than the collecting funnel. Take care not to set traps in situations which are likely to flood in rain.

Describe how to find the trap again. Use flagging tape (not too obvious at the entry point) along the path to the trap. Describe in your notebook how to get to the entry point. Get a GPS fix at the trap site (you may well need a GPS with an external aerial). Record the accuracy (try to obtain an estimated accuracy of less than 10 m by averaging your reading) and altitude if possible. If a GPS is not available get a grid reference with a detailed description. (Note: grid references only have an accuracy of ±100 m.)

Installing and setting a trap

- Dig a suitably sized hole to just accommodate a length of plastic downpipe. Try not to dig the hole much wider than the downpipe.
- Set the length of downpipe base into the hole so the top of the downpipe is level with the ground (Fig. 5). Then pack the soil back firmly around the downpipe until it forms a smooth surface which slopes up slightly to the top edge of the downpipe. There should be no gaps between the soil and the top of the downpipe when you are finished so that even minute insects do not escape. The top of the downpipe should be, if anything, slightly below the surface of the ground (Fig. 6).
- Clear any leaf litter, sticks or other debris that might fall into the trap and offer a means of escape for insects, away from the immediate vicinity of the trap.

- Add about half a beer cup full of propylene glycol antifreeze and drop the cup into the downpipe (Fig. 6).

- Place the cover centrally over the pitfall trap. If using steel, first bend the spikes down and push these into the ground until the cover is 30–40 mm above the ground. If using aluminium with pre-bent corners (Fig. 6), simply place centrally over the pitfall trap and place a weight (e.g. small stone or log) on top.

- Write the trap number on the trap cover and, if using an aluminium cover, place a small weight such as a stone or log in the centre to keep it in place (Fig. 7).

- Take a GPS reading of the trap location. Try to average the location to an estimated error of less than 10 m.

Figure 5. Place a cup in the downpipe to collect overflow soil before building up and packing down the soil up to the lip of the downpipe.

Figure 6. After finishing packing down the soil, remove the cup, tip out any soil and half fill the cup with propylene glycol before replacing it in the downpipe. Note the design of the lid, which is an alternative pattern when using aluminium for lightness. Also, the soil slopes up very slightly towards the downpipe lip, although this is not evident in the photograph.

Figure 7. Write the trap number on the cover and if using an aluminium cover, complete the trap by placing a small weight on it (not shown).

Collecting the samples

- Open a sample jar and remove the label and Chux multicloth. Keep the lid handy.

- Pull the cover out of the ground and extract the beer cup. This is best done by inserting two fingers and spreading them to grip the sides of the cup.

- Place the square of Chux multicloth in the small plastic funnel, and sit the funnel in the spare sample jar.

- Pour the contents of the beer cup through the multicloth. Recover the antifreeze from the sample jar and return it to the beer cup. Swirl this around if necessary and repeat, pouring through the multicloth each time until the beer cup is clean.

- When all the insects have been sieved into the Chux multicloth:
    - Remove any large twigs or leaves (first check there are no insects attached).
    - Gather the corners of the multicloth together and place it in the sample jar.
    - Write the date, trap number, collector, and a brief location name on the label using pencil and add it to the sample jar. Screw on the lid.
    - Write the date, trap number, your initials, and brief location on the lid of the sample jar using a permanent marker pen.
    - Either reset the trap (screw the white collection jar back onto the trap, keeping the trap upright, return the trap to ground and secure it with the pegs) or remove the trap as appropriate (see 'Removing the traps').
    - If you are resetting the trap and the antifreeze in the trap has not been diluted by rainwater, re-use it and top it up with fresh antifreeze if necessary. If it has been

diluted with rain, tip it back into the pitfall hole, half fill the beer cup with fresh antifreeze plus more table salt, and replace it in the downpipe.

– Take the sample jar back to the vehicle with you.

– When you reach the vehicle, unscrew the lid of each sample and add 95% (undiluted) alcohol to the sample jar until it is about half-full, screw the lid on firmly, and invert the sample jar a couple of times so that the alcohol completely mixes with the sample. The sample will now keep indefinitely. It is important to add alcohol because the insects will slowly deteriorate if kept in antifreeze alone.

– Try to keep the sample jars upright from now on. Alcohol may leak out slightly and this will remove the labels on the lids if the samples are not kept upright.

Figure 8. Sample collection. Remove the cup and strain the contents through the Chux multicloth (left) then repeatedly return the strained fluid back into the jar, swish it around and strain it again (right) until the sample jar is clean. Remove any large sticks or leaves from the material in the Chux multicloth, making sure no invertebrates are attached to them.

Figure 9. Pick up the Chux multicloth by the four corners and drop it and the strained material into a collection container (left). Write out the label (right) and place it in the container so it can be read from the outside.
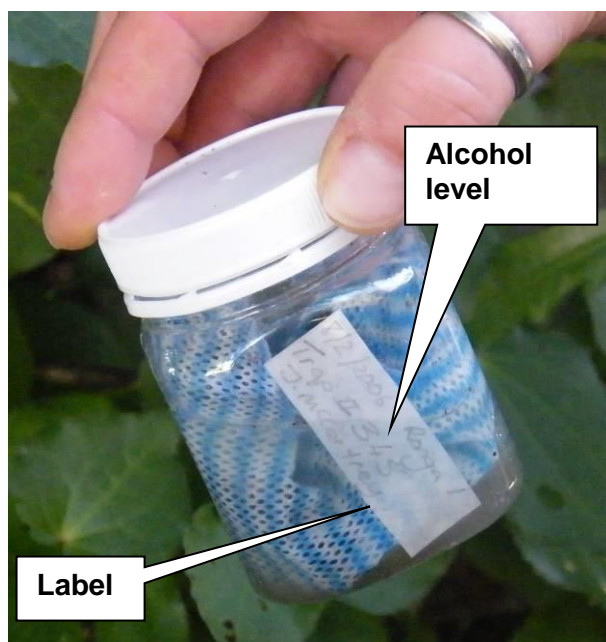
Figure 10. The completed sample should look like this.

Finally, recycle the antifreeze if you are setting the trap again, or tip it into the hole if you are removing the trap permanently. Collect up the Chux multicloth by drawing together the four corners and drop it into the empty collecting jar. Write out the label (date, person, trap number, brief locality) with pencil and add it to the sample. Close the lid, and write the date, trap number, your initials and brief location on the lid.

Remember to add undiluted alcohol when you get the sample back to the vehicle.

Removing the traps

- Ensure that you have a good GPS reading in case the trap sites have to be re-visited by someone else for other purposes later (e.g. taking another sample next year or doing a RECCE plot).
- Collect the sample as detailed in 'Collecting the samples'.
- Remove cover, bend the spikes back level with the rest of the cover if using steel covers, remove the downpipe from the ground. Leave the hole to mark the spot but place a bunch of sticks vertically into the hole so any insects that fall in can climb out. Attach some flagging tape as near to the hole as possible. Write the trap number on it with the indelible marker pen.
- Take the trap components and samples back out to the vehicle with you. **Remember to add the alcohol to the samples when you arrive at your vehicle.**

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Appendix A

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

doccm-428226   Example data sheet invertebrate monitoring

doccm-525907   Introduction to statistical analysis of invertebrate monitoring data

doccm-146272   Standard inventory and monitoring project plan