Cave invertebrate collecting guide

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CONTENTS

Abstract

The cave environment and the habits and density of cave invertebrates demand important modifications to the techniques used to collect invertebrates on the surface. This report describes the equipment and techniques that have been trialed for collecting both terrestrial and aquatic cave invertebrates, highlighting those aspects requiring special awareness underground. Both the merits and disadvantages of the techniques trialed are discussed and recommendations made. The methodology of handling and preserving the invertebrates collected is also detailed. This report is intended for use as a guide for future invertebrate survey work in cave habitats and at this stage it is not envisaged that cave invertebrate monitoring would have a significant role in cave management work. While the live capture techniques described could possibly be adapted for use in cave monitoring work, the nature and scarcity of cave invertebrates mean that they are unlikely to be easy to monitor in any meaningful way.

Keywords: cave fauna, invertebrates, terrestrial, aquatic, monitoring, sampling, collecting, trapping, New Zealand

1. Introduction to cave biology

1.1 DEFINITIONS OF CAVE FAUNA TYPES

The New Zealand cave fauna includes a wide variety of animals, but is dominated by invertebrates. The use of caves as roost sites for bats and birds as seen overseas is uncommon here and the only live vertebrates commonly encountered underground are frogs, eels, rats, and possums. The invertebrate fauna however is diverse with both aquatic and terrestrial species, and it is this fauna we wish to investigate.

Some invertebrates in New Zealand are known to be truly troglobitic, cave adapted species, but a large proportion of the invertebrates found underground are surface species which are there accidentally, or surface species which may choose to visit or live in caves but which cannot complete their entire lifecycle underground (trogloxenes). Rats and possums which have dens in caves fit in the trogloxene category, as do cave weta which live and reproduce in the cave, but have been observed on vegetation outside the cave at night (Richards 1966).

Other species found in caves can be classed as troglophiles, which are not true cave species either. Troglophiles are species which can live out their entire life cycle inside caves, but which can supposedly also live in other habitats given the appropriate conditions. For example, the glow-worm is just as at home on damp, shady banks in the bush as over a stream in a cave. However some species presumed to be troglophiles have never actually been found living outside of the cave environment. These are still considered to be troglophiles simply because they don't have the *appearance* of being physically adapted to the cave environment. That is, they have perfectly good eyes, coloured bodies, and their body hairs or feelers are not especially elongated with respect to those of their surface relatives. So, even though the protected cave spider *Spelungula cavernicola* has never been shown to complete its life-cycle outside of the cave environment, it would still be considered to be a troglophile as its physical appearance shows none of the typical adaptations of invertebrates to the cave environment.

Troglobites are species which appear to be totally adapted to life in caves and unable to survive outside of the cave environment. As with the troglophiles this implies that considerable knowledge of the life history of any species must be acquired before we can definitively call it a troglobite. Again the reality is that we take the appearance short-cut: it is troglobitic if it displays morphological characters which limit it to subterranean habitats (Howarth 1983). So any species with eyes reduced or absent, reduced pigmentation, elongated sensory hairs or antennae (in the case of terrestrial species), etc. is assumed to be a troglobite until proven otherwise. Hamilton-Smith (1971) gives a review of this classification and terminology.

1.2 WHY SURVEY AND COLLECT CAVE INVERTEBRATES?

To say our knowledge of the New Zealand cave invertebrate fauna is limited would be a gross understatement. A few entomologists worked on New Zealand cave invertebrates in the 1960's (May 1963; Richards 1962; Townsend 1963, 1971; Johns 1991), but since then cave work has been more piecemeal; the majority of material being collected by observant cavers rather than as part of a concerted surveying programme. There has been little systematic sampling work completed in New Zealand caves, and new species of troglobite are still being found.

The Department of Conservation (DOC) has funded a Science & Research Unit project over the last year to investigate cave invertebrate survey methods, to review the status of New Zealand cave invertebrate work and to renew work on surveying and cataloguing the invertebrates found in New Zealand caves throughout the country. This report describes the various techniques used for surveying cave invertebrates, and is intended as a guideline for DOC staff involved in cave invertebrate work, initially as part of this project. Precise details are given so that a lack of experience does not hinder survey work, and to encourage people to try using more than one technique by describing the advantages of that approach.

Though the supporting taxonomic work is lacking for many species, surveying and collecting cave invertebrates is valuable, and interesting specimens spur taxonomists on in their work. In this small project we have collected several new species of troglobite. The cave faunas of the Nelson and Waitomo areas are the best studied of any in New Zealand. Despite this, a new species of troglobite was collected in a Waitomo cave in this project. There are karst areas in New Zealand where little or no invertebrate searching or collecting has yet been done, and where the potential of a troglobite presence is relatively high. In New South Wales, Australia, the numbers of known troglobitic genera increased by one third when an intensive survey program was undertaken (Eberhard & Spate 1995). This could also be the case in New Zealand, if the work were to be undertaken.

Troglobites require very specific habitats and conditions, and may have extremely restricted distributions. This means that they are likely to be vulnerable to becoming endangered. In order to direct conservation effort to protect cave faunas most effectively, we need an awareness of what we are aiming to protect—which can only be gained by survey work. There are documented cases of cave faunas being degraded and species lost in some Australian caves, due to direct disturbance in the cave or to changes in water flow or quality due to surface activities (Eberhard & Spate 1995). Little is known of the cave fauna in a number of areas in New Zealand, let alone its conservation status, for example, caves on the East Coast of the North Island (Whakapunake, Wairoa) and alpine caves in Kahurangi National Park. Without surveying and documenting cave faunas we run the risk of losing species without knowing they were even present.

Habitat protection is probably the best technique to conserve vulnerable cave species, as it will benefit whole cave communities. But it would be easier to sell

the idea of cave habitat protection and karst system management if we could give more examples of the species which are at risk, or which would benefit directly from protection efforts. More survey work of cave invertebrates is, therefore, important.

1.3 WHAT TO SAMPLE, AND WHAT INFORMATION DO WE NEED?

The invertebrates of most interest in a cave invertebrate survey are those with a limited distribution and which may be at some degree of conservation risk. These are most likely to be the troglobites and those species of troglophiles which seem to be restricted to the cave environment. As the latter will be generally more difficult to identify and separate from surface accidentals or trogloxenes for example, then we should concentrate on troglobites in the first instance.

To sample as thoroughly as possible, invertebrates should be collected from a broad range of cave habitats using a number of different techniques.

There is no absolute number of specimens of each species which should be collected. Personal judgement and common sense must be applied in deciding how many invertebrates of each species to collect so that the cave population is not damaged. Some cave invertebrates have low population densities and reproduce slowly, so are particularly vulnerable to over collecting (Chapman 1985).

The number of animals which can be collected without impacting on the population depends very much on what is being collected. If the collection target is crustacea which are common in large numbers (such as Amphipods), then removing more than thirty from a pool is unlikely to have an impact on the total population. In contrast, collecting half that number of Carabids using bait could have a much larger impact. The bait may draw these mobile animals into the trap over long distances, so a large proportion of the total population could be captured.

While care must be taken to avoid over-collecting cave invertebrates, it should be remembered that most invertebrates which are actually seen are probably 'the tip of the iceberg'—only a fraction of those which are actually present. Just because only one animal of a certain species is seen in the cave, you shouldn't necessarily decide against collecting it on the grounds of its possible rarity. Many invertebrates in caves are found as isolated specimens, but it is highly likely that there are further animals tucked away in the fissures and cracks of cave habitat which cannot be surveyed.

We need to collect enough material to allow identifications to be made, but again without reducing the cave population irreparably. A taxonomist can do a much more thorough identification job on a small series of 6–10 animals than on just one or two. In some groups the key characteristics used in identification may be found only on adults of one sex, so the chances of having a useful animal are increased if more than one is collected.

Labelling specimens carefully is vital if they are to be of use. Record labels on good quality paper, in pencil when you are underground or waterproof pigment liner ink on goatskin parchment paper in the lab. Include the cave name (add a grid reference later) and locality in the cave, region, collector's name, date, and habitat information.

In addition to labelling samples, take careful notes while you are collecting, recording what you collect and where, along with general observations such as: behaviour, numbers of organisms active, evidence of flooding, etc. Particularly important are habitat descriptions. Notes should be made using pencil in a waterproof notebook.

2. Sampling for terrestrial fauna

2.1 SELECTING TERRESTRIAL SAMPLING SITES

Early biospeleological work was based in temperate limestone caves, and theories developed around the findings in that situation. It was only in the mid-1970s that the way we think of troglobite habitats was revolutionised by cave biologist Francis Howarth with the realisation that troglobite faunas are also present in non-limestone caves and in the voids of fractured rock, in non-temperate climates, and that many obligate cave species are not relicts (Howarth 1983).

Working mainly in Hawaiian lava caves, as well as other tropical lava and limestone caves, Howarth recognised two terrestrial habitats within caves:

- 'Macrocaverns'—those passages or voids which are sufficiently large that humans could move through them (assuming that they were accessible through negotiable entrances)
- 'Mesocaverns'—smaller voids from c. 20 cm down to 1 mm wide which often penetrate the rock well beyond the macrocaverns (Howarth 1983)

Howarth's work indicated that the primary habitat for the most of the species he studied was the mesocavern habitat, an environment characterised by saturation humidities and the build-up of potentially noxious gases such as CO_2. Howarth reasoned that only when key conditions of the mesocavern habitat were met in macrocaverns would troglobitic species be found there. He went on to demonstrate this by finding many more troglobite species in those parts of certain Australian caves where there was a build-up of 'foul air' (i.e. CO_2) than could be found in sections without the foul air build up (Howarth & Stone 1990).

To what extent Howarth's findings hold for limestone and marble karst caves in temperate New Zealand is uncertain. Our caves are not usually prone to foul air build-up and most of the troglobite species which have been collected here so far have been taken from macrocaverns with high humidity, but normal levels of CO_2. This could mean that there are species which we have not yet found!

Work by Howarth (1983) and Juberthie & Delay (1981) implies that it is likely that our troglobites also make heavy use of the mesocavern habitat and we should bear that in mind when choosing collecting sites. Mesocavernous connections between macrocaverns (i.e. caves) probably explain why a species found in one cave in a discrete block of limestone is usually found in other caves in the same block. Therefore it is useful to think in terms of sampling discrete karst units for their troglobites, rather than individual caves. However, some individual caves may present a better range of accessible habitats for sampling than others.

There are no hard and fast rules about where to look for troglobitic animals: they can turn up in a variety of situations, often unsought. Generally, it is best to start by looking for passages or sites *well away from the cave entrance*, with two characteristics:

- Little or *no obvious air movement*
- *A source of food*, such as a stream or seep

Significant air movement raises the possibility of drying of cave substrates and most cave-adapted fauna cannot survive in much less than about 95% relative humidity. Air movement can be created by a chimney effect in caves with more than one entrance and large draughty passages between entrances are usually not suitable. The movement of an active stream can also induce air movement.

Cave ecosystem energy inputs are limited to whatever falls or gets washed into the cave from outside (silts, plant detritus, dead animals, etc.) and to the bodies or wastes of species which occasionally or habitually use the cave for shelter. Streams, seeps, etc., are the primary means by which these energy sources are carried deeper into the cave. Generally, fauna will not be found in old, dry upper-level passages without some sort of food input. (However, there is at least one cave beetle species in the Nelson area which seems to occur in just such passages—which means that we don't know enough about its way of life to recognise its food sources.)

Large clean-washed stream passages subject to regular, large-scale flooding are not likely to be worth searching for terrestrial cave fauna. However, searching small muddy ledges, nooks, and crannies with detritus on the passage walls above the regular flood levels can be worthwhile. Passages carrying smaller streams, even temporary streams or seepages, not prone to excess flooding, and passages occasionally subject to flood overflows or backwaters from stream passages below, are a good place to start looking. Any passages containing wet seeps running down walls or even just permanently wet walls are worth making a note of also. These seep zones can contain a variety of small species.

Streamways with silt banks which are occasionally inundated by floodwaters, with deposits of small amounts of leaf litter, twigs, etc., are worthy of careful investigation, even if the stream does create some degree of air movement. Detritus feeders and scavengers may be attracted to this material and predators may come to hunt. As a general rule, large detritus deposits may not have very many troglobites in residence, as in these situations surface or threshold species washed in with the detritus may prove more competitive. Smaller, more remote deposits are likely to be best.

Although these sites are totally within the dark, many troglobites still prefer to be in situations where they are in close contact with surfaces both above and below them, i.e. small cracks. Furthermore, many troglobites, although blind, can sense light (or possibly heat), and will seek refuge in holes or crevices if disturbed by lamplight. Therefore, within these sites the animals are just as likely to be found underneath detritus as on top of it, including burrowing into twigs and pieces of wood. They may also occur under stones and other debris on or near silt banks. Rockfalls and breakdown areas, especially with a seep or small stream associated, are worth searching, baiting or pitfall trapping in, as they provide conditions similar to those found in mesocavernous habitats.

In shallow caves with plant roots penetrating from the surface, especially where this occurs at a reasonable distance from the nearest entrance, it is worth spending some time searching on or around the roots. Troglobitic root sucking species are common in shallow lava caves in Hawaii (Hoch & Howarth 1993). There is at least one species of cave-dwelling sap-sucking bug in New Zealand and there is no reason there shouldn't be more. Other species may be attracted to plant exudates on the roots.

Lava caves

Sampling in lava caves should take Howarth's findings into account. Basalt lava forms extensive networks of cracks on cooling and this crack (mesocavern) habitat is normally far more extensive than any network of lava tubes which can be entered by humans in the same basalt field. Methods are required which sample these mesocavern systems, whether from within caves or tunnels or from the outside by digging into the larva/scoria. Baiting may be especially important in lava caves where suitable habitat in the macrocavernous areas is uncommon—bait may draw organisms out of mesocaverns which are otherwise impossible to sample. Again, penetrating tree roots may be a potential source of fauna.

2.2 METHODS AND EQUIPMENT FOR COLLECTING TERRESTRIAL SPECIES

2.2.1 Hand searching

Aim to collect as many species as possible, but not in large numbers. With this in mind, the selective nature of hand collecting makes it the most preferable method of sampling terrestrial cave fauna. A bright electric light and good eyesight is necessary as many of the invertebrates, such as Pseudoscorpions and Springtails (Collembola), may be only a millimetre or so in length.

Search likely sites such as silt banks and detritus deposits carefully, turning over and then replacing rocks, boulders and other debris. Invertebrates such as cave beetles, spiders and harvestmen may be found out on silt and debris deposits looking for food. Other spiders, millipedes, and springtails may be found in or under plant debris or rocks, etc. Gently break open larger twigs, branches and pieces of wood which are well rotted and burrowed and look for troglobites within them.

Water seeps running down walls, especially walls with small nooks and crannies, should be carefully examined, preferably close-up with an electric light. These waters can sometimes harbour a variety of small species, troglobitic or surface, and may also have associated predators. Cave springtails, millipedes, snails and other species may be found in such places.

Water seeps on walls, and wet walls generally, should also be closely searched for troglobitic spiders. (Look up!) Shine a torch along the wall at an angle to your field of view and examine it closely. Fine webs hanging from small irregularities may indicate the presence of such species. Some troglobitic spiders are extremely small and easily overlooked. In the Mt Owen karst there is a species which weaves an extremely fine sheet of web suspended below small overhangs in wet seeps on walls. The spider is often found suspended on a thread below this sheet, a few centimetres off the wall. The spiders and their webs are so fine that they are almost impossible to find unless by chance. The best searching method is to focus on a likely section of wall from 20–30 cm away (close to, but not at, our limit of focus) and blow gently on the wall. If any out-of-focus movement becomes visible in front of the wall, one can focus back and find a spider swaying on its thread.

Searching tree roots should be done with care. It is easy to dislodge fauna from roots hanging in the middle of a passage. This could in fact be a method for sampling a large root mass which is too dense to be easily searched: spread a dark coloured sheet on the ground beneath the roots and vibrate or shake the root mass to see what falls on the sheet. Cave invertebrates are usually light coloured and will show up against the dark material. Such root masses will not always be conveniently placed for this method to be used, but if it is adopted, the roots should be visually inspected first and must not be damaged in any way. The one species of cave root-feeding bug which we do know of is capable of jumping up to a metre away if it is disturbed, so the sheet would need to be extended some distance out from the base of the root mass.

Avoid over-collecting specimens of a single species. There is some evidence from overseas caves of troglobite populations being severely depleted by over-collecting. If you are uncertain whether a series of similar specimens consists of one or more species, collect a sample of 6–10 animals, rather than all of them.

Vials

Specimens should be collected into plastic vials underground. An alcohol proof seal is required for these vials. See-through vials are best so you can see immediately whether the vial you pick up is already full or not. However if you use a system of clearly separating full from empty vials while in the field, this is not necessary.

If you collect your specimens live, predator species should be placed singly in containers, and certainly not with potential prey species. To minimise the number of containers you need to carry, half fill several containers with a solution of 80% alcohol, and collect specimens directly into these, using one container for specimens from one site, passage or habitat type within a cave. (Label!) Don't mix specimens from different caves. The 80% solution allows for the high humidity of the cave environment which will cause the alcohol to

become diluted by absorbing extra moisture every time that the vial is opened. Two drops of glycerine can be added per vial to keep specimens flexible.

Labels

It is important that all samples are labelled at the time of collecting. Mix-ups happen easily if labels are added later and an unlabelled specimen is useless. Labels should be on good quality white paper that can be left in liquid (such as laundry tag manilla or goatskin parchment, although photocopy paper will do in the short-term), and written with either pencil (easiest underground) or permanent pigment pen (Staedtler pigment liner). If using laser-printed labels, ensure they are oven-baked (180° for 5 minutes, or less, so paper doesn't brown) after printing to set the ink, otherwise over time in alcohol the ink can peel off the paper. Labels should be placed inside the vial with the sample, as there is a high risk of sticky labels or marker pen on the outside of the vial coming off, especially when there is alcohol in use. Ensure you label each vial with the cave name and details of the site in which the sample was collected, and include the date and the collector's name.

Tools

Larger animals can be picked up directly or otherwise coerced into the collecting vial, but many will resist and avoid the vial. If your vials contain alcohol, and for smaller specimens in general, rather than scooping animals directly into the vial you will need to use some basic tools to pick up specimens.

Forceps: a pair of soft forceps which require little pressure, such as fine-tipped 'Feather light' entomological forceps (available from entomological equipment suppliers) are useful for picking up specimens. Forceps which are very fine at the tip must be used with care as specimens are easily damaged by squeezing too hard. The forceps themselves are easily damaged by dropping them and will need to be protected when taken underground. Wrap them up, or push the tip into a tube or sleeve.

Paint brush: a child's camel hair watercolour paintbrush (available cheaply from toyshops or stationers) is useful for flicking specimens into the vial. Alternatively, if you push the end of the bristles directly on to a small specimen they will often secure it long enough to transfer it to the vial. You can also moisten the bristles so that the specimens will adhere to them.

Pooter: an old-fashioned entomologist's pooter can be very useful for securing small specimens which are difficult to handle, or which take refuge in small crevices. Remember, however, that this is a humid environment, so damp specimens may adhere to the sides of the inlet tube, especially if water droplets are sucked in with the specimen. The inlet tube should preferably be made of clear glass or plastic so that the specimen can be seen if it does get stuck in this way.

2.2.2 Baiting

A good way of collecting cave animals is to place baits in likely habitats and leave them for a few days, then revisit to see what has been attracted to them. A variety of baits can attract troglobites and different baits will attract different

groups of species. Again, this selective method of collecting is preferable to killing invertebrates indiscriminately.

It has been shown that baited pitfall traps produce different results to a visual census (Poulson & Culver 1968) probably due to organisms being drawn out from the inaccessible cracks and fissures that comprise the mesocavernous zone, and from large distances throughout the cave. Baiting may, therefore, be an extremely important method of surveying cave invertebrates.

Baits should be placed in sites where they are unlikely to be trodden on or washed out by flooding over the period that they are to be left. Mark and carefully note down these sites so that they can be re-found easily. Place the baits under a degree of cover (e.g. under rocks or surrounded by cobbles) to provide shelter for organisms attracted to them. This may also prevent large, non-target species such as cave weta from getting to the baits and eating them before the troglobites are attracted. When re-visiting the site, carefully turn over and search the rocks, cobbles, or other debris in the vicinity of the bait. In Jamaican caves, cheese was placed under tinfoil and invertebrates which stayed under the tinfoil were collected (Stringer pers. comm.).

Small quantities of smelly meat and cheese can be very effective for attracting scavenger-predators such as cave beetles. The meat can be any raw or cooked meat which has gone off, or a suitably smelly processed version such as canned cat food. Similarly, the cheese could be rancid cheddar or something naturally smelly such as feta. Only very small amounts are needed at any one site (one tenth of a teaspoonful is plenty). Try different baits in the same area and see if some are more effective than others. These baits can be left for a period of a couple of days, up to a week, or so before checking. If left too long they will be eaten and the animals will have dispersed again.

For those who prefer the traditional approach, on long caving expeditions the usual bait used by the expedition biologist is human excrement, often available in plentiful supply. It is easily as effective as ripe meat, but is a bit messy to use.

Smear some cottage cheese or cooked sweet potato on one face of a large stone or small rock, then place it, smeared face down, supported slightly above the ground on pebbles, etc. Place other stones or rocks round about to provide shelter for animals attracted to the site. Leave in place for one to two weeks to allow the smear to grow mould (this happens fast in the humid cave environment), then return and search under the rocks and stones for fauna. These baits should attract certain detritivores and fungus feeding species and they could also attract predators intent on feeding on these species (Eberhard pers. com.).

Other baits worth trying include grains, such as weetbix flakes, and dried fruits, such as apricots. These could probably be left for longer periods of time also (e.g. one to two weeks). Be experimental with different baits and different time periods. Cave carabids in New Zealand have been observed by one caver to be attracted to the syrup of a dissolving barley sugar left on a nook in a cave wall.

2.2.3 Baited pitfall traps

Baited pitfall traps are a technique which can place the cave fauna at great risk, due to their potential to collect invertebrates to extinction if left out for a long

period. In fact their use may be frowned upon in some biospeleological circles, and we must be extremely careful in our use of this technique. **Never leave a baited pitfall trap set in a cave indefinitely**.

A pitfall trap consists of a vial or jar sunk into the ground to its full depth, with the ground surface level with the opening. A suitable preservative (Gault's solution, see Section 5.1) is then placed in this container and bait (usually cheese/rotted meat) placed or suspended within it, in such a way that it does not end up in the preservative. A flat rock is placed over this, propped up slightly on one side by a pebble to leave a small gap for fauna to get to the trap. The rock keeps out water drips, which might flood the trap, and larger animals such as cave weta or rats. Ensure that the ground surface is carefully flattened flush with the rim of the container, as many species will be put off by the presence of a glass or plastic rim. Baited pitfall traps in caves need only be small. Vials 55 mm high with a diameter of 45 mm were large enough to trap the invertebrates which we were interested in, but small enough for reasonably easy placement.

Unbaited pitfall traps, although commonly used above-ground where there is a large ground-dwelling fauna moving around, are next to useless in the cave environment as the density of invertebrates in the cave community is so much less.

The trickiest part with baited pitfall traps is finding a method for placing the bait without creating an escape route for the catch. One simple method is to use a small vial (c. 7 mL) to contain the bait, placed inside the pitfall container (c. 80 mL) (Fig. 1). Don't allow the small bait vial to float or to touch the side of the pitfall container—weigh the bait vial down with a small stone inside or use more bait. The bait (catfood, or smelly cheese) should be placed carefully into this central vial and a depth of approximately 2 cm of the Gault's solution placed in the main container.

Pitfall traps should be checked 7–10 days after being set, or sooner if possible. A pitfall trap should be removed if it has collected more than five or six of one species, to avoid over-collecting from a site. Careful judgement must be used in deciding when to remove pitfall traps so that they do not endanger any population, while still collecting the less common species. Pitfall traps should

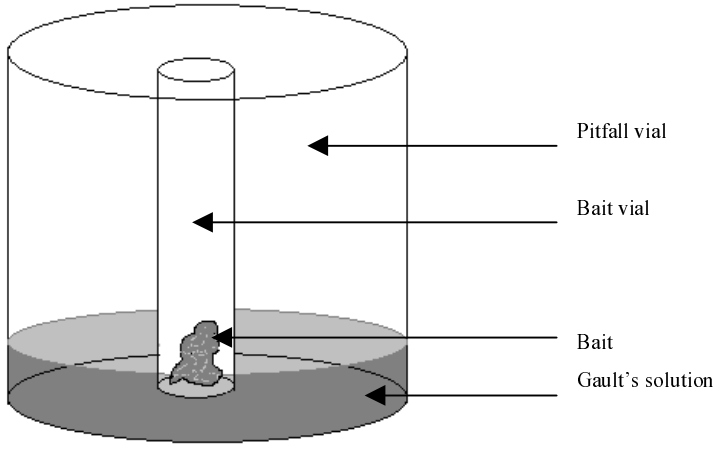

Figure 1. Baited pitfall trap, for sampling terrestrial cave invertebrates.

be spread throughout the suitable habitat of a cave and not clustered too closely together. Pitfall traps must be checked regularly if you are leaving them in place (at least every 10 days) and must be closed down if you will be unable to check them, because of their potential to collect to extinction.

Ideally, specimens should be carefully removed from the pitfall container using forceps and placed in 75–80% ethanol for transport out of the cave. With the bait vial lidded and removed, the pitfall trap container could be carried out as is, however, remember to transfer specimens out of the Gault's when you get home. To remove large numbers of invertebrates from the Gault's solution it may be easiest to use a piece of mesh fabric (e.g. net curtain) to strain out the animals, while transferring the Gault's solution to another container for use in a new pitfall trap or to take out of the cave. Label each container clearly!

Other options for pitfall baiting methods you may want to try are:

- A thin wire with a small hook at either end to fit on the rim of the trap, and a series of bends in the middle to form a small 'platform' on which the bait can be placed. (It needs to be thin wire or some species will negotiate their way to the bait without falling off.)
- Some sort of miniature wire stand with a flat platform at the top for the bait.

The latter has been successfully made out of two crossed paper clips, with half of one of them bent straight up into the air and the end of this then bent around parallel with the base to form an attachment point for mounting a small disc (c. 1 cm diameter) of plastic cut from an ice cream container which held the bait. The second clip is slid across the base of the first to give better support. The two are held together by a large glob of epoxy resin, which is also used to attach the plastic disc at the top. An alternative method is to smear bait on the underside of a rock directly above the pitfall trap, but some organisms are likely to feed on this bait without falling into pitfall trap.

Baited pitfall traps should *always* be examined within a week to ten days of placement and removed if there are more than five or six specimens of any one species in them. Baited pitfall traps will keep on catching animals as long as the bait lasts, which could have a big impact on cave communities. **Remember: Never leave a baited pitfall trap set in a cave indefinitely!**

2.2.4 Terrestrial leaf litter packs

Damp leaf litter traps have been used to good effect in Australian cave invertebrate studies (Weinstein & Slaney 1995). The basic idea is to place damp leaf litter in a mesh pouch and put it on the cave floor for a length of time to allow cave invertebrates to utilise the material. This is not so much a trapping method, as a matter of providing habitat for invertebrates and then surveying those organisms which are attracted to it, similar to baiting. The leaf packs we used were a simple 80 × 100 mm pouch of plastic mesh with 5 mm holes.

Collect forest leaf litter—individual leaves, not too broken down—and wash thoroughly in water to remove forest floor invertebrates. Pack the leaves loosely into the mesh bag and set in position in the cave. Check the leaf pack after a few days and re-moisten if it has dried out. (Australian studies set up dripper bottles so that the litter remained moist, but this is unlikely to be necessary in New Zealand caves.)

If you are able to sort the litter in the cave you will be able to release all but one or two of each species, and the length of time the leaf packs are left out is of no concern. This is probably the best approach, but ensure your light is good and bright or you will miss small organisms. It is easiest if the leaf litter is tipped into a small tray for sorting through, and a small amount of water in the bottom of the tray can be used to float invertebrates off the leaf litter.

However, if you want to take the whole sample out of the cave to be sorted, then sampling over short periods to begin with is a good idea. You do not want to over-collect, if the method works brilliantly. If you are removing the mesh bag and the litter it contains from the cave to sort the sample, the leaf pack should be sealed in a plastic bag (with a label), separately from other leaf packs. These sealed bags can then be placed together in a plastic container so they aren't crushed while being carried out of the cave. Invertebrates contained in the litter should be removed to 70% ethanol with glycerine.

2.2.5 Other methods

Trial other methods as you see fit, keeping in mind the impact which you will have on the cave environment and the invertebrate populations. Discuss such techniques with Ian Millar or Maree Hunt if you wish.

Live capture pitfall traps

An alternative which allows you to be more selective of your catch is to set live-capture pitfall traps. These are essentially baited pitfall traps without preservative, which allow you then to select the specimens you wish to keep and to free the rest. For these to work without the animals eating each other, a larger container is necessary so that pebbles, etc. can be provided for shelter for the catch. This will mean that the mouth of the container should be sufficiently wide to insert your hand to place or remove these. Essentially this trap is the same as simply placing a bait, except that the animals can't escape once the bait is eaten. A trap of this size may require that a wooden or plastic cover be placed over it if a suitable size of rock cannot be found. Stones or small rocks can then hold the cover in place. Larger pitfall traps like this are likely to be the least flexible in where they can be used, because they require a substantial area and thickness of silt for burial. They will also be substantially bulkier to carry into the cave. If you decide to try live-capture pitfall traps you will need to look for containers with a distinct bulge out ('shoulder') below the section where the lid screws on, to ensure that your catch cannot climb out again once it has fallen in. You may also find that some new plastic containers give off gases which may repel the animals before they can fall in. Placement of pitfall traps of this size may be difficult, or require disturbance to the habitat beyond that which can be justified. Judgement of how useful this method might be must be made according to the available habitat.

Habitats outside caves

Troglobitic beetles have been collected from mines and tunnels dug into many different types of rock in non-limestone areas of Japan (Ueno 1877). Artificial tunnels which cut into the mesocavernous zone in loose fissured rocks, and which maintain environmental conditions favourable to troglobites provide good habitat in which to sample those species. Ueno (1976) set baited pitfalls in

abandoned man-made mine adits (horizontal tunnels) for 6 months before collecting specimens of these beetles.

Another method which may allow us to sample invertebrates from the interstitial habitats (i.e. mesocavern) is to set baited pitfall traps in karst or broken rock outside caves. This has been done successfully by Juberthie (1981) and Millar (pers. comm.). Mesocavernous habitats are described in shales, granites, soluble (limestone), and volcanic rock types (Howarth 1983; Juberthie 1981).

The idea is to get down into the interconnected cracks, fissures, and voids that anastomose through the rock and which make up the mesocavern habitat. To access this habitat, you need to be able to dig through the soil to a point where you find rocks with a degree of open space between them. Once this depth is reached the baited pitfall trap can be installed as usual. Good places to try would be in a stable scree slope, with soil and vegetation cover, below a limestone bluff, or in scoria.

Wedge a board or large flat rock across the hole, preferably some distance down. By closing the hole off from the surface as much as possible the pitfall trap is likely to catch organisms moving out of limestone fissures, rather than collecting surface or soil species. Replacing some of the removed turf over this might help to maintain the humidity and temperatures required by any cave dwellers in the mesocavern (Fig. 2).

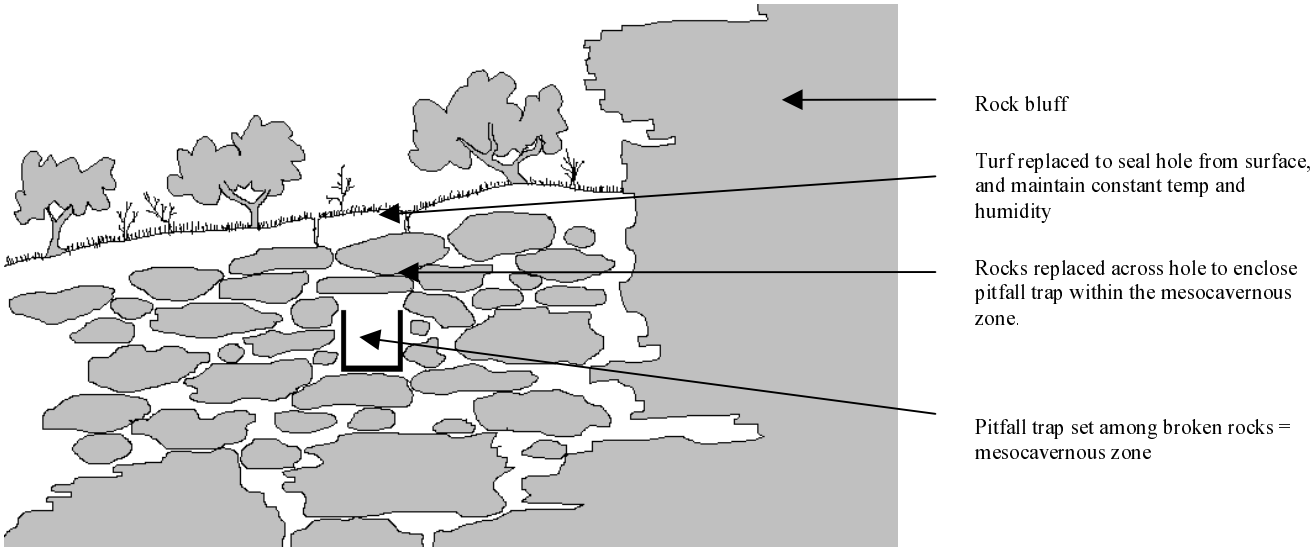

Figure 2. Pitfall trap set outside cave, buried within mesocavernous habitat.

3. Sampling for aquatic fauna

3.1 SELECTING AQUATIC SAMPLING SITES

To avoid collecting samples comprised mainly of surface species, it is important to select cave waters which are generated within the cave by percolation, rather than those which are fed directly from surface streams. Looking at a topographic map or going for a walk over the surface may answer this question. There are obvious clues if the source of the water is a surface stream, but this may take some working out. Think about the following things, starting with the most obvious.

- Can one walk up the stream and out of the cave?
- Is there significant litter such as leaves and sticks in the stream?
- Is the water stained brown with humics?
- Is there froth on pools? Propensity to froth is related to levels of humics.

It may prove more profitable to sample pools, back eddies, and side flows rather than quickly moving waters.

3.2 SAMPLING METHODS AND EQUIPMENT FOR COLLECTING AQUATIC SPECIES

3.2.1 Hand netting aquatic samples

Tumble stones, etc., to stir up the invertebrates, scrubbing any boulders, pebbles, and the general substrate with the net positioned downstream while you scrub, so that the water flow is filtered through it. The invertebrates we are wanting to sample will collect in the net, as well as stones and debris.

The larger pieces of debris should be cleaned and removed, to reduce the bulk of the sample as much as possible, without losing invertebrates in the process. Stones and large debris can be scrubbed clean by working inside the net with water flowing through it, so that invertebrates which are dislodged are retained in the sample.

When as much as possible of the larger debris has been removed, the sample should be transferred to a tray where it can be spread out for closer inspection. Collect a small amount of water in the tray and invert the net, tipping material into the tray and dabbing the net into the water in the tray, so that the sample floats off the net into the water. Splashing water onto the net may help. Water can be washed through the net again so that material will collect in the bottom of the net, making it easier to remove the sample from the net. If necessary the net can be used to re-filter the sample to remove excess water. Invertebrates which are obvious can be removed from the sample at this stage, if you wish, and placed in alcohol. (Include a label in every vial!)

The sample should be poured from the tray into a vial for transport out of the cave, ensuring it is labelled immediately you put it in the vial.

For every day spent collecting a full day will need to be allowed for sorting aquatic samples. Large quantities of silt and gravel in a sample will take a long time to sort through, which is why we suggest taking time in the cave to filter the sample down to a manageable volume, while taking care that invertebrates are not lost in the procedure.

Sort the sample immediately following the collecting trip. Samples will hold in the fridge for a day or two, but sorting will be easier the earlier it is done, and specimens will be in better condition for taxonomic identification. Do not put off sorting for more than three days as specimens may be ruined and the effort put into collecting wasted.

Some of the amphipods and other species can be very difficult to pick out of the tray with forceps—they move too fast. For some of these, a thin glass pipette with rubber bulb is very useful. They can be sucked up and dropped into a vial of alcohol (which rapidly dilutes of course). Then lift them out with forceps into fresh alcohol, 70% ethanol with two drops of glycerine. The fine glass tubes are very delicate and get broken easily, so don't attempt to take them underground.

Nets with a mesh of 250 microns will collect very small organisms such as copepods and ostracods. When sorting samples it is important to search for these very small organisms, so the use of a dissecting microscope will be necessary. Sort by spreading small amounts of material in a petri dish or dark coloured tray and sift through it under the microscope, removing invertebrates to a vial containing 75–80% ethanol with 2 drops of glycerine added, except for snails and flatworms.

Invertebrates collected from the same site can be stored in the same vial, although particularly if there are large numbers it would be useful if this first sort produced separate vials for various taxa—to whatever level you can manage (e.g. 'Amphipods' or even more generally 'larvae'). Remember to label sorted vials with site information, etc.

After each sample, the net should be inverted and any residue washed out. Flicking them clean works well. Before sampling in a new cave the net must be thoroughly cleaned and dried so that organisms are not transferred between sites.

3.2.2 Baited aquatic traps

Small traps were designed out of plastic coke bottles, to use as baited traps for collecting aquatic invertebrates. This is a prototype and is likely to require further modification, so make improvements as they become obvious. Mesh covered holes in the bottom of the bottle allow water to flow through the trap, while the inverted neck of the bottle forms a narrow funnel, allowing easy access by invertebrates into the trap, but impeding their exit from the trap. The ridges and waist of a coke bottle ensures the inverted neck clips tightly into position (Fig. 3).

This trap should function either way round, but when it is set with bait it would be expected that many of the invertebrates which enter the trap would be moving upstream, following the scent of bait in the water. Therefore, if the open end of the funnel is pointing downstream, those invertebrates will be able

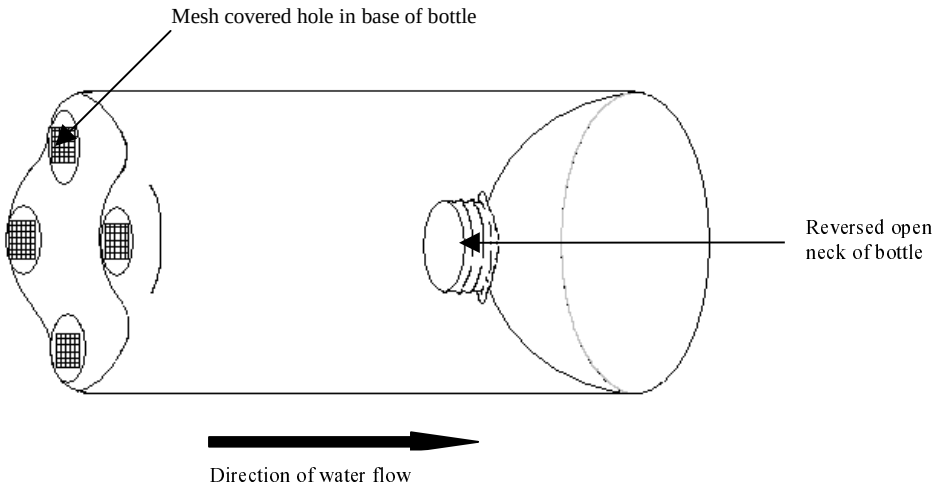

Figure 3. Baited trap for sampling aquatic cave invertebrates.

to enter the trap directly. Water will flow in through the meshed-over end and out through the bottle neck. In a still pool the orientation of the trap will not matter.

It may be useful to weight the trap by placing a stone inside. Also build some rocks or pebbles up around it on the outside to help hold it in place against the current, perhaps piling some inside the mouth of the funnel to aid the entry of crawling invertebrates. You may want to anchor the trap with a string or wedge it with pins or large stones, etc., so it can't wash away should the water rise.

This design of trap will probably not be much good in high-velocity water. Try a range of locations: gently flowing water, pools, and back eddies. Consider whether invertebrates are potentially able to travel to the trap from either the up or downstream directions.

A range of different baits may also be worth trying. By baiting the traps we hope to draw in invertebrates which may not be sampled in scrub or drift samples (but we may just catch eels and koura). Bait should be of a type that will hold together when left in water. Kippers in brine were found to be good in Australian studies (Eberhard & Spate 1995) and other possibilities include a chunk of stewing meat, or cheese (proteinaceous and smelly are the main bait criteria). Depending on how the bait holds together, you may need to contain it in a mesh bag or stocking to stop excessive contamination of the sample by bait. Careful handling to avoid further contamination by bait when removing invertebrates from the trap will be required.

Leaving traps in the water for longer than several days may allow debris to block the mesh or specimens may be spoiled (or have escaped). Leave the trap in place for up to five days to start with. If the catch is small you might wish to replace it for a longer period. You may need to replace the bait.

For transport out of the cave, invertebrates should be emptied from the trap into a small container, perhaps by washing material into the sorting tray. Untape the funnel which will then pull out. A squeeze bottle may be necessary to rinse invertebrates out of the trap. Invertebrates can be placed straight into 75–80% ethanol as described earlier for other aquatic samples (except Gastropods and flatworms), or sorted back in the lab, as with other aquatic

samples. (Label!) Don't carry the trap out of the cave without first removing the sample. Traps must be thoroughly washed and dried between uses in different caves or limestone areas so there is no risk of transporting animals between areas.

3.2.3 Aquatic leaf litter packs

Leave the leaf pack (described in Section 2.2.4 Terrestrial leaf litter packs) in the stream for several weeks to be colonised by stream invertebrates. Don't leave it in place for so long that the leaf litter disintegrates entirely. Anchor it in place using pins or rocks, or even tie a string to something fixed as extra security. When retrieving the leaf pack from the stream, hold the hand net open just downstream to catch any invertebrates which are disturbed as you pick the pack up. Sort through the litter in the tray and wash invertebrates off the leaves, picking them out with forceps and placing them directly into 70% ethanol.

3.2.4 Drift net samples

Drift sampling has been used extensively for sampling aquatic invertebrates at spring-heads and cave entrances internationally. Drift sampling can be used in a cave stream underground, or in the mouth of a cave resurgence or spring where a net can be placed so that invertebrates carried out of the cave in the water flow are caught in the net.

Site selection is important in the use of drift nets, as with other aquatic sampling. Once again the source of the water should be from percolation rather than being fed from a surface stream. This technique will be appropriate for a small number of caves, and may require modification of the net to a size which suits the resurgence (or passage if a suitable location is found within a cave).

The net can be left in place for whatever length of time suits, but if left for longer than 24 hours or in flow which carries a lot of debris, the net may become clogged up or be damaged. Standard practise when deploying the net is to note the start and finish time, proportion of flow captured by the net (you want it to collect as much as possible), depth of water, and velocity. From these you can work out drift rates (numbers/hr) and densities (numbers/m^3 of water filtered). While this may be necessary in a quantitative survey, such information is not vital.

Samples should be removed from the net and specimens sorted and transferred into 75–80% ethanol as described earlier for other aquatic samples.

4. Discussion and final recommendations

All techniques described, bar live-capture baited and non-baited pitfall traps, have been trialed in New Zealand cave situations. They are appropriate for use when collecting invertebrates in cave habitats, however, for various reasons some techniques proved to be more useful than others.

Collecting cave invertebrates by hand was the method that was generally preferred by staff because results are immediate, only those animals required for taxonomic purposes are collected, and much can be achieved in a single visit to a cave. More collecting was done in this way, therefore, than using the other techniques described. Hand collecting of invertebrates was found to turn up a wide range of cave invertebrates. The main drawback of collecting by hand is that cave invertebrates are usually present at very low densities, so search productivity was often low. Also those species which don't often venture into human sized passages or macrocaverns are unlikely to be sampled. In addition, not everyone is good at collecting cave invertebrates by hand. A combination of a bright light, good eyesight, and quick reactions are required to be successful when collecting cave invertebrates by hand because many are very small and move rapidly when disturbed.

It was difficult to get people to make a commitment to the repeat visits into a cave which are required to check or remove traps for techniques such as baited traps and leaf litter packs. This is a particular problem in caves that are remote, or difficult to access. As a result these methods were not utilised to the level hoped, although enough sampling was done to confirm these methods as useful. The subsequent sorting of aquatic samples was labour intensive which made this method unpopular, also. There is still strong potential with aquatic sampling, however, and continued collecting is likely to turn up new species.

Both baited aquatic traps and leaf litter packs were useful in aquatic situations, and many aquatic crustacea were collected using these. Stewing meat worked well as a bait in the aquatic traps, and held together well so the samples weren't too horrible to handle. Cleaning leaf litter effectively before taking it underground was difficult when litter was decomposing, but the more decomposed material seemed to attract invertebrates far more than fresh leaves did, so the effort was worthwhile.

Leaf litter packs set on silty banks were not so effective as those in more damp situations such as under seeps. Small snails and amphipods were collected from litter packs in damp locations, but no invertebrates had moved into litter left on dry silt. Invertebrate activity has been observed on silty banks, so it is likely that leaf packs in those drier areas may simply need to be left in place for longer than a week in order for invertebrates to move into them. This would vary from cave to cave.

Baited pitfall traps were very effective and collected a wide range of invertebrates. This is a technique that, if done carefully and with consideration to conservation issues, is extremely useful in New Zealand caves. Organisms

which are present in very low densities or which are generally present in the inaccessible mesocaverns are far more likely to be collected in baited pitfall traps than when collecting by hand because they are drawn from relatively large distances by the odour of the bait (Poulson & Culver 1968). Predators such as carabids are at particular risk of pitfall-trap overkill.

The biggest problem we encountered with aquatic sampling for cave-adapted stream invertebrates was finding cave waters which were from percolation, and which did not carry dominantly epigean (surface dwelling) species. This was harder than expected, and many cave stream and resurgence samples carried only surface species.

Baits left under foil were not effective over the short periods we were able to leave them out (several days) and with little invertebrate activity no difference in the use of sweet, proteinacous, or grain baits could be detected. Baits were not left in place long enough for fungi to develop, so the ability of such baits to attract fungus-feeding species is still unknown. In some caves rodent interference with baits was a problem.

The aim of developing techniques which could be used as cave invertebrate monitoring tools could not be realised in this project, mainly because of the nature of cave invertebrates. These animals are very small and live at low densities and are, therefore, difficult to monitor. Such monitoring would have to be non-destructive in order not to deplete cave communities.

Observing invertebrate activity on baits may be one useful method of monitoring cave invertebrates, however, densities of animals in caves are generally so low that results are likely to be poor. Recording numbers of observations of active animals may be useful, but also has problems. This would only be useful for species which are easily and often observed active in caves (e.g. *Spelungula cavernicola,* or cave harvestmen). Activity (and observation of activity) is highly variable and it must be recognised that lack of observations does not mean that the animals are not present in the cave.

Monitoring (and minimising) habitat change is a more realistic manner of managing cave communities.

5. Additional information

5.1 PRESERVATIVES

Most of the invertebrates collected can be placed directly into a vial with 70% ethanol (ethyl alcohol), and two drops of glycerine to keep invertebrates flexible. Use 75–80% ethanol when in a cave where humidity is high or for wet samples (i.e. aquatics), both of which will dilute the alcohol. When specimens may be held for some time before being sent to a taxonomist and in cases where there are large specimens or several specimens in one vial making up a significant bulk of material, then the alcohol should be removed and fresh added about 24 hours after first placing material in it.

Some organisms need special treatment when preserving them, however, and are of no use to taxonomists if not handled correctly.

Gastropods (snails and slugs)

If you are sending snails to Karin Mahlfeld (refer to Section 5.5) she is pleased to receive live material. Snails should be placed in a small tube with damp moss or similar, in which they will be able to survive for several days. (Otherwise they need to be narcotised with menthol crystals overnight, then fixed in 5% formalin, well buffered with sodium carbonate and then transferred to 80% ethanol within 24 hours. Note it is most important that formalin be properly buffered otherwise the shells will be destroyed or damaged.)

For more simple handling, snails may be relaxed by drowning in fresh water, which has been boiled to remove oxygen and then cooled. The snails can then be preserved in 75% ethanol (Ponder & Warren 1965). Snails preserved in this way may not be useful for dissection.

Aquatic planarians (flatworms)

Bertha Allison (Canterbury Museum) recommends that we fix aquatic planarians (flatworms) in Carnoy's fluid with the ether component omitted (recipe calls for 3 parts chloroform)

Carnoy's fluid, minus ether component (Walker & Crosby 1988)

Ethanol, commercial grade 95%	6 parts
Glacial acetic acid	1 part

Gault's solution (for pitfall traps) (Walker & Crosby 1988)

Sodium chloride (salt)		50 g
Chloral hydrate		10 g
Potassium nitrate		10 g
Water	mix to	1000 mL and add several drops of glycerine

Note that one can mix dry parts into a 1 L container, and add water in the field. Label the bottle indelibly.

5.2 POSTING SPECIMENS

It may be necessary to send specimens to experts for identification. To avoid damage to specimens, part-fill the vial containing the specimen with alcohol and push in a plug of cottonwool into the ethanol, close to the specimens, but don't squash them. This will allow you to squeeze out air bubbles and will reduce the movement of specimens during transit. Push fine forceps down beside the cottonwool plug to allow all the air to escape. It is not important to get the last of the air out of the top of the vial because the cottonwool will prevent it from reaching the specimen. Once air bubbles have been excluded, fill the vial to the top with ethanol. Wedge labels against the side of the vial with cottonwool so movement doesn't damage soft bodied specimens.

Double packing vials containing ethanol is essential. Packing small vials into a larger alcohol proof container gives extra protection, or alternatively vials can be sealed in a plastic bag. Parafilm may also be useful, stretched tightly around a container lid it is self-sealing and moisture resistant.

Depending how many and what sized vials are being posted, pack them into a box, or use an express-pac bubble-lined courier bag. Use whatever packing you have to hand such as foam chips or shredded paper. Pack carefully, expecting the package to be both crushed and dropped!

Fast Post or courier all samples, particularly live material. With live or unpreserved material it is vital that the recipient is warned of delivery prior to its postage, so they can ensure that the specimens do not sit around unattended. It may pay to use a refrigerated courier service for this type of specimen.

5.3 CAVE CONSERVATION

The first and foremost rule of caving is to be aware of the extremely fragile and pristine nature of the cave environment and the fact that nature will not repair any damage done by clumsy or unthinking people—even over many thousands of years.

In the 'heat of the chase' it might be easy to trample or destroy cave features which have taken a long time to form. These include not only the obvious features—flowstone, stals, and straws—but also more mundane things such as sculptured mud surfaces formed as a result of drying, or by dripping water. Damage to the cave cannot be justified by any scientific investigation (not even the collection of an extra invertebrate), so be aware of potentially vulnerable sites.

In addition, compression of sediments will destroy invertebrate habitat and should be avoided, so ensure you stick to the main routes, where damage has already been done. Care must be taken when placing pitfall traps that the area is not disturbed irreparably. Try to place pitfalls discretely.

Be careful where you put your hands and feet, and don't touch speleothems (formation) as even clean hands bear salts which will stain calcite forever. Be especially careful you don't smash things with your head. (Wearing a helmet mounted lamp will help a lot.) Be aware of your surroundings and your proximity to delicate features, and remind the people around you, especially where extra care is required.

As the Australians say (surprisingly expressively), '**Cave softly**!'

5.4 CAVE INVERTEBRATE COLLECTING-KIT CHECKLIST

General collecting gear

- Vials—use whatever you like so long as it is plastic and alcohol proof:
  —7 mL and 25 mL for collecting invertebrates into underground
  —70 mL vials for aquatic samples and pitfall traps
- Baited pitfall traps—use 70 mL vial with approximately 2 cm depth of Gault's solution, and a 7 mL vial containing bait positioned in centre. Bait vial may need to be weighed down with a stone, etc., under the bait so it doesn't float.

- Forceps—Fine tipped feather-lite, useful both underground and in sorting. Fine forceps for lab use mainly. (Be gentle on specimens and don't drop the forceps!)
- Paint brush—to pick up small invertebrates, especially useful, if tip is dampened
- Label pen—Staedtler pigment liner better for labels out of cave
- Waterproof notebook and pencil, and label paper
- Hand lens
- Leaf litter packs and well-washed leaves
- Dark-coloured cloth to spread under root masses—so light coloured invertebrates which drop off are easily seen. (A black plastic rubbish bag might be good for this.)

Aquatic sampling

- Sorting tray
- Hand net (short handle only)
- Baited trap (coke bottle)
- ?Drift net for cave resurgence
- 70 mL container—for sample, or may need some larger plastic containers (e.g. small peanut butter jars, etc., depending how you go at reducing the sample)
- Squeeze bottle—for rinsing aquatics out of trap or net

Additional requirements underground

- Catfood/other bait materials
- Ethanol (with glycerine). 75–80% underground and for aquatic species
- Gault's solution for pitfalls
- Carnoy's fluid for flatworms—these could be held in an empty vial and fixed in Carnoy's when out of the cave, but must be done within hours of collecting
- Remember label paper and a pencil for labelling vials as they are filled

Personal caving gear

- Overalls, preferably with polypro underneath (be prepared to get wet!)
- Gumboots or tramping boots (not good new leather ones!)
- Helmet and lamp (electric is best for this work)
- Back-up light sources—small maglite, or similar, with new batteries and spare bulb
- Bag—something without zippers, preferably, as cave mud and grit is insidious stuff and wrecks zippers. Army shoulder-type bags are good, with lots of pockets to keep full and empty vials separate
- You may also want kneepads or gloves for added comfort
- Small first-aid kit with essentials like foil survival blanket and a bandage

5.5 TAXONOMISTS

The following taxonomists are studying particular groups and would appreciate material collected from cave sources.

Graham Fenwick—aquatic amphipods from cave streams and resurgences
NIWA, PO Box 8602, Christchurch, New Zealand
Phone (Work) (03) 348-8987 email: g.fenwick@niwa.cri.nz

Karin Mahlfeld and Frank Climo—snails, aquatic and terrestrial
5 Imlay Crescent, Ngaio, Wellington 6004, New Zealand
Phone (Home) (04) 479-3829

Dr Brendan Moyle—Psuedoscorpions
Senior Lecturer, Department of Commerce, Massey University (Albany),
Private Bag 102 904, Auckland, New Zealand
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Dr Ian Townsend—cave carabids (Trechinae)
30b The Avenue, Levin, New Zealand
Phone (Home) (06) 368-8409

Phil Sirvid—spiders
Entomology curator, Museum of New Zealand,
PO Box 467, Wellington, New Zealand
Phone (Work) (04) 381-7000

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