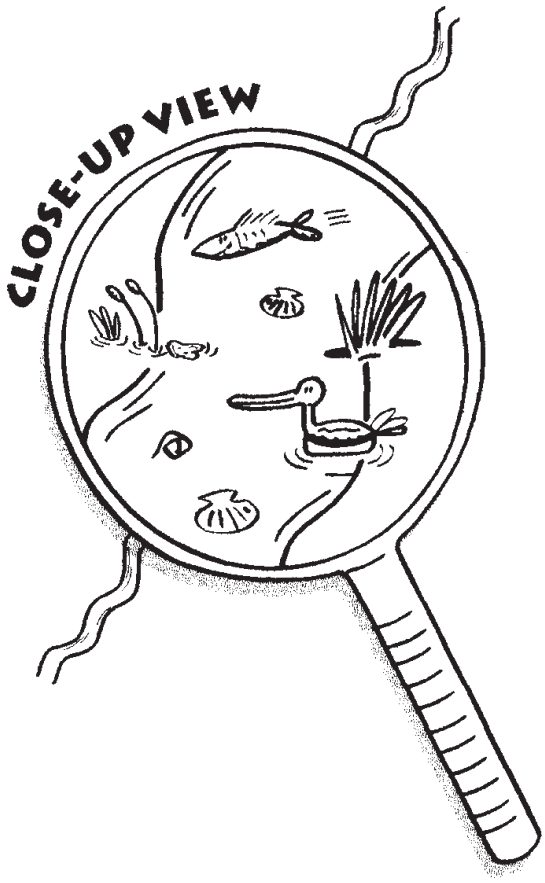
Biological Surveys

Biological surveys involve sampling aquatic life.

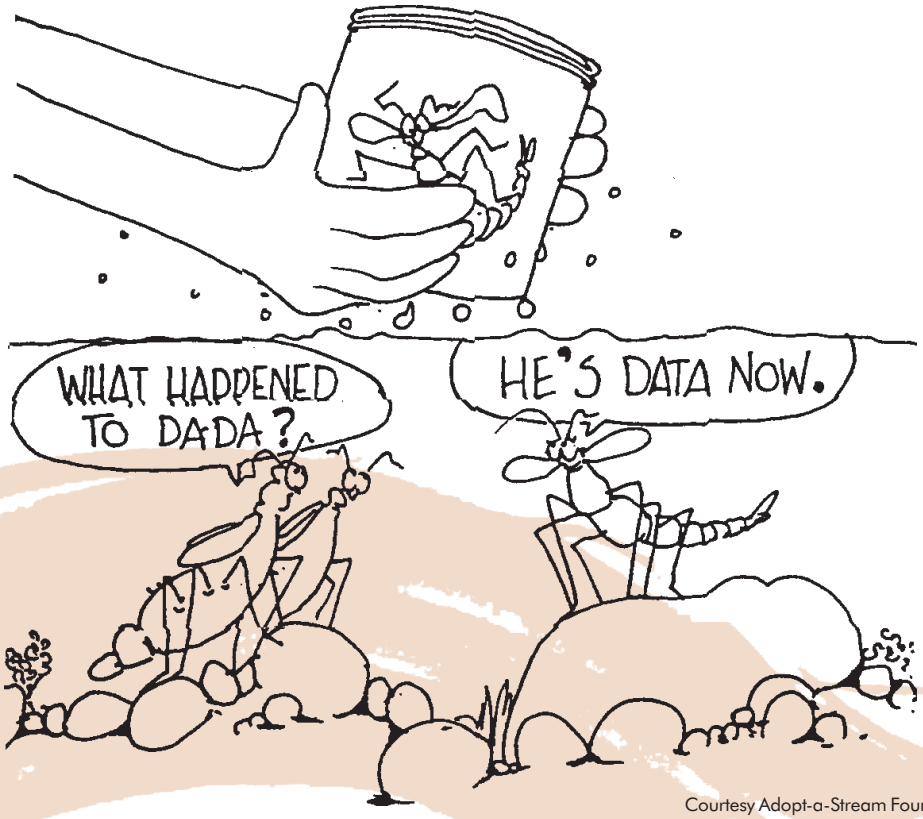
You can focus on any or all of the types of organisms in the aquatic community: algae, **macroinvertebrates** or fish.



Macroinvertebrates are animals that do not have backbones, but are visible to the naked eye. In this section, we use the terms macroinvertebrates and bugs interchangeably. Many stream bugs are insects, but many other types can also be found, including worms, snails and crustaceans such as koura.

In this section you will learn how to gauge the general health of your stream using various sampling and sorting methods. The method you choose will determine how much information your sampling will provide.

At the simplest level, gauge the overall health of the stream by seeing what is present or absent. This is known as a **qualitative** method, because you don't consider the number of bugs. To detect more subtle effects on the stream, more precise sampling and detailed sorting is needed. Keys and photos may be needed to help you identify bugs and you will have to become familiar with the tiny body parts you use to identify the different types of bugs. While most of the bugs can be identified in the field and released back to the stream, this **quantitative** method may require taking 'mystery bugs' back to the lab for identification. Removing bugs should be kept to an absolute minimum.



Courtesy Adopt-a-Stream Foundation

How Macroinvertebrates Indicate Stream Health

Bugs are often used to indicate stream health because:

- They are sensitive to chemical and physical changes to their environment.
- A range of sensitivity/tolerance levels (1-4 or 1-10) can be given to the many different types of macroinvertebrates. The presence, absence or relatively high numbers of the different types can then indicate stream health.
- They move very little, making them a captive audience for the effects of pollution. Being relatively fixed in position also makes them suitable for assessing effects on specific sites by comparing upstream and downstream.
- There are many different types of stream bugs. Each type has a specific set of needs which the stream must provide for it to survive. Altering the stream may have a major effect on the abundance and distribution of different macroinvertebrates.
- They are relatively easy and inexpensive to collect.
- Bugs are easier to identify than algae, which also vary in their tolerance to pollution.

Macroinvertebrate communities reflect the presence of most environmental stresses through changes in community composition, and many provide general indications about types of pollution. Chemical testing may be used to confirm the presence and particular type of pollutant/s.

However, chemical testing will not usually detect occasional pollution and information will be limited by what tests you do. Phosphate and nitrate tests will not show faecal contamination for example.

Biological surveys using only macroinvertebrates have some drawbacks:

- They do not respond directly to all pollutants such as herbicide.
- You can't identify a particular pollutant. Species which may be sensitive to one pollutant may tolerate another.
- Macroinvertebrates may be missing due to factors than just water quality, e.g. habitat damage or recent flooding.
- Natural variation in community structure must be taken into account. For example, stonefly larvae are important in monitoring water quality but there are naturally fewer in lowland streams. This is not necessarily because of pollution but rather that many stoneflies prefer upland streams rich in dissolved oxygen.
- Macroinvertebrates often show a very patchy distribution and large numbers of samples are needed to be accurate.
- Numbers may vary seasonally as insects hatch and emerge from the stream.
- The need for the samplers to have skills in classifying plant and animal groups.

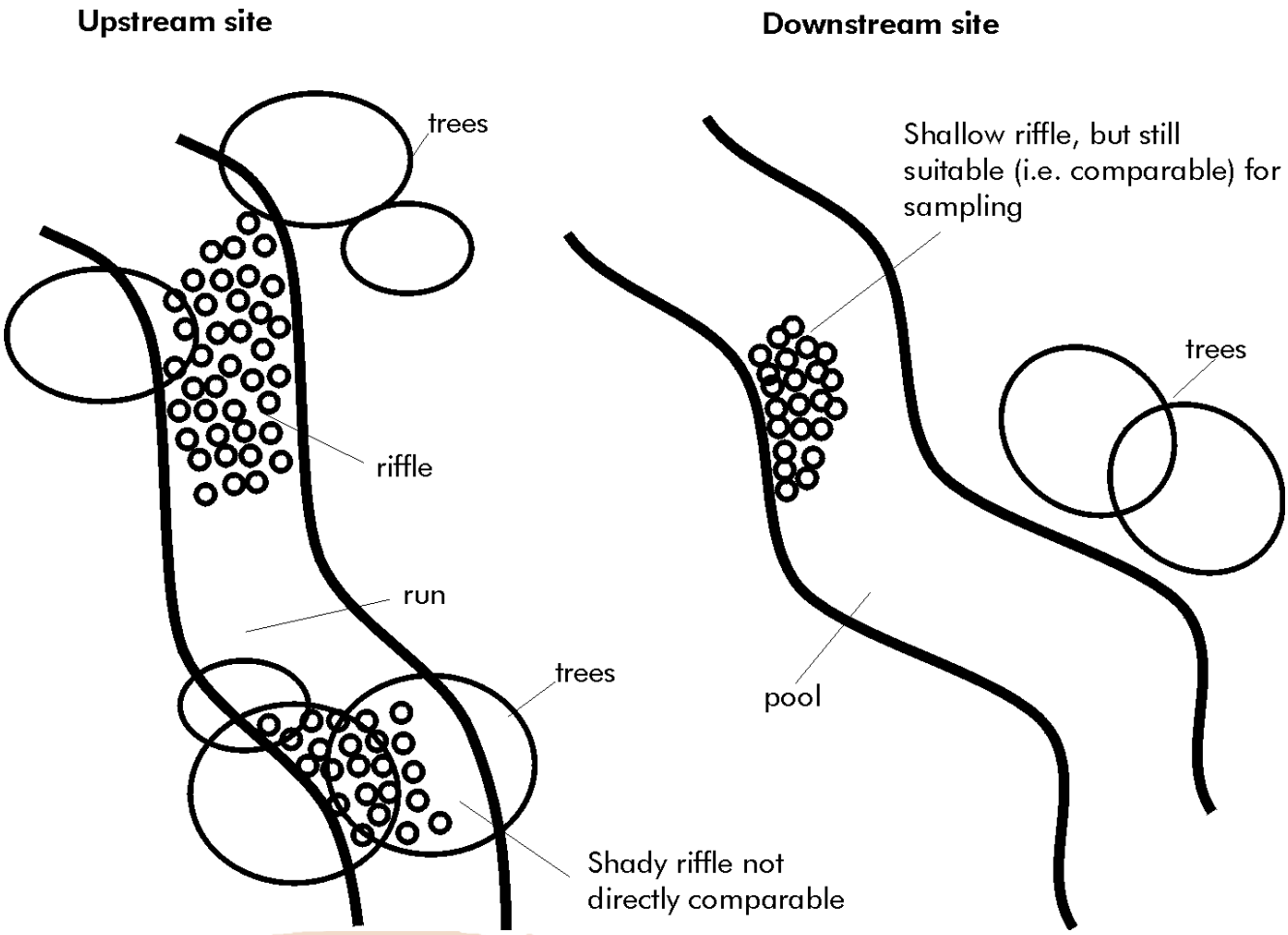
Sampling Frequency

Ideally, macroinvertebrate sampling should be undertaken quarterly, certainly no less than twice a year, preferably in late summer and late winter. Late summer sampling will collect larger specimens of insects that emerged during summer, making identification easier.

Lower flows and higher temperatures in summer may make the effects of stresses such as pollution worse - a chance to decide what to do in the worst conditions.

Choosing Monitoring Sites

It is often useful to select at least two different monitoring sites on each stream, an upstream reference site and a downstream site. The reference site can be used to set a 'benchmark' for the catchment and you should select it on how it compares in physical features with the downstream site(s).



Stream monitoring sites - illustration showing:

- characteristics (riffle if possible, or alternative suitable substrates)
- easy access to 50-100m to allow everyone to spread out and flat areas for groups to work
- the stream must be able to be waded into under normal flow conditions and safe for sampling.

Results from your monitoring programme may be used to determine a trend in stream health or to decide the effect of various land uses or activities within the catchment. To make sure that you can make reasonably valid conclusions it is vital to select sites carefully.

Use a number of criteria to select suitable sites:

- It should be as similar as possible to the downstream site. Biological communities are sensitive to differences in habitat qualities as well as differences in water quality;
- The reach or section of stream chosen for monitoring should have physical features that are typical of the whole stream;
- Suitable comparable substrate to the reference site is available to sample.

Selecting Appropriate Habitat

Riffle areas - sections with fast flowing, shallow water - are most frequently used for sampling and assessment purposes because they support the most varied groups of bugs, and they tend to be easier to sample than other habitat.

Ideal riffle characteristics include:

- a current velocity between 0.3m/s and 0.7m/s
- a depth of about 10-30cm
- plenty of gravel or cobbles (greater than 50 percent of riffle if possible)
- at least two 1m^2 areas of riffle with a gravel or cobble bottom

However, gravel or cobble substrate is not always available so you will need to select alternative **habitat** to sample.

The sampling procedures outlined below enable samples to be gathered from the most common types of habitat: riffle or run, large woody debris (snags), banks or submerged aquatic plants.

Sampling from only one type of habitat at each monitoring site, such as a riffle area, will allow you to compare results between sites.

Note: If you want to determine bug distribution versus aquatic habitat type you will need to sample all main habitat types separately. If you sample each site in the same way (duration or area) you'll be able to compare population densities at each site.

Note the results from each sample separately for the distribution and habitat analysis but may be totalled later for the calculation of different stream health indices.

Field Sampling

Collection procedures should be standardised as much as possible:

- Sample from similar habitat types within each section of the stream.
- Try to sample for the same time, distance or area.
- Use the same techniques for collecting samples throughout the reach if possible.

1. Kick Sampling

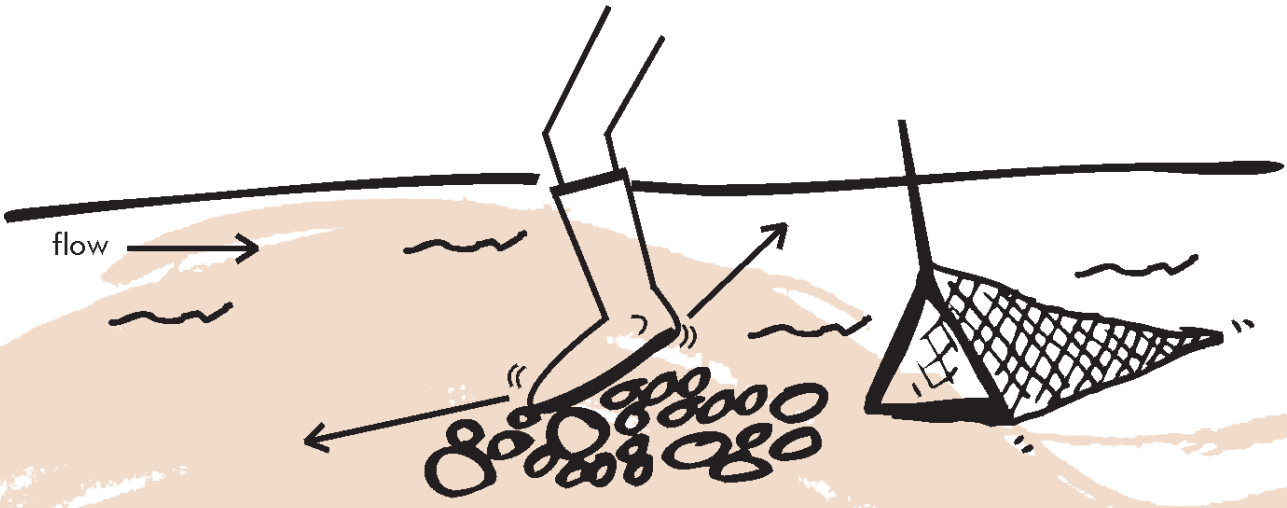
Kick sampling is a simple but effective method used to dislodge animals living in or on the rocky bottom of the stream and allows the current to sweep them into the net. Two sampling areas within the riffle or run are needed, each about 1m^2 in area and preferably with different flows: one with fast current and the other an area of slower current.

Equipment

- Kick sampling net (preferably with a mesh size of 250 μm)
- Bucket
- Suitable footwear, to protect your kicking foot
- Sorting tray

Taking A Kick Sample

1. Select a shallow riffle area with a depth of 10-30cm and stones that are gravel or cobble sized (2-64mm and 64-256mm across the middle axes respectively), if available.
2. Mark the position of your sampling site on the site map and note any reference points.
3. Sampling begins at the downstream edge of the selected site and moves upstream, covering a predetermined area, say 0.25m^2. The area you decide to sample will vary with bug density. Size your sampling area to enable you to collect a minimum of 50 bugs.
4. Position the net about 0.5m just downstream of your foot, with the bottom edge lying firmly against the stream bed.
5. Using the toe or heel of your foot (whichever is best protected!), dislodge the upper layer of cobble or gravel and scrape the underlying bed.
6. Use a forward scooping motion to lift the net from the water to prevent any of the organisms escaping.
7. Carefully empty the net contents into a bucket part filled (about 5cm) with water. Rinse clinging organisms off the net by splashing with water from the stream.



net placed about 0.5m downstream of the kicker

8. After each sampling, wash the net in the stream to remove debris.
9. Transfer some or the entire collected sample into the white tray for sorting. If you get lots of stones in the sample, swirl the water in the bucket to dislodge and separate any clinging bugs.
10. Record the percentage of each habitat type in the area sampled. Note conditions at the time of sampling, such as higher than usual flows, that may influence the results and interpretation. Also record observations of fish or other fauna.

Larger substrate may not be moved by kicking. See below for an alternative method for large substrate.

Where riffle or run habitat with coarse gravel or cobble bottom is not available, other submerged fixed structures such as snags can be used.

2. Sweep Sampling

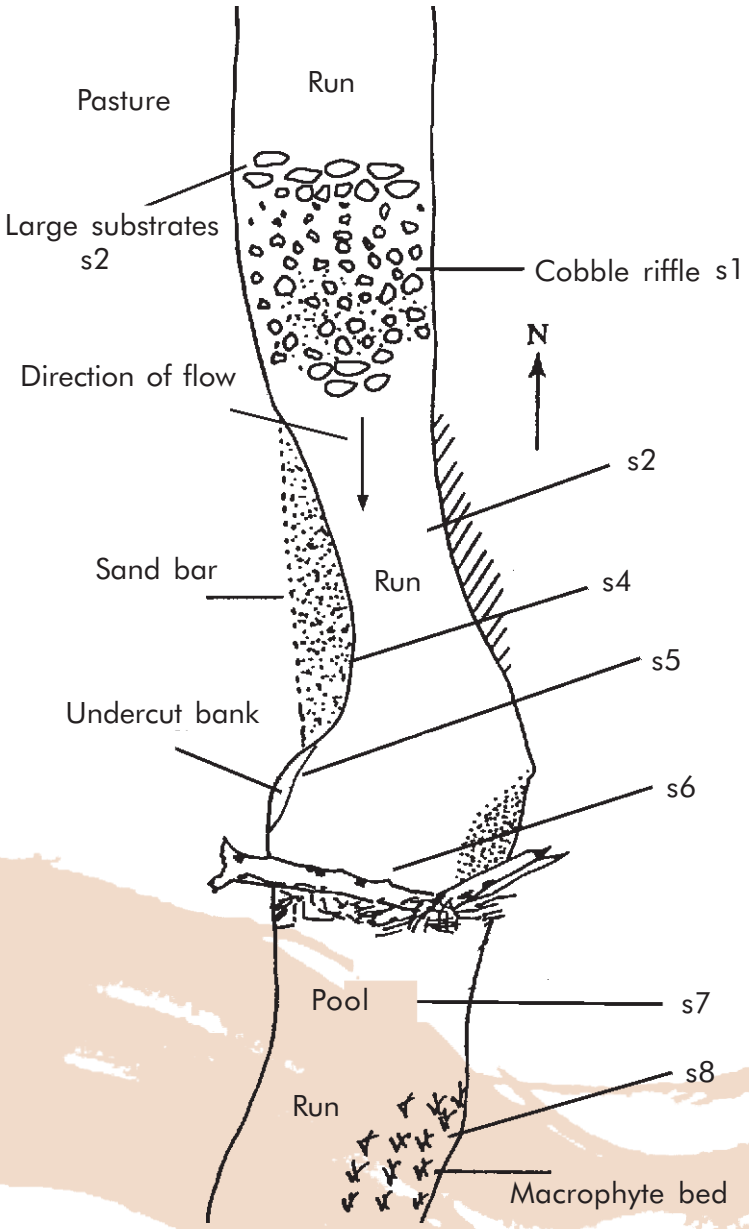
Use this method to collect organisms living in and around vegetation or edges of the stream. When rocks or other fixed substrates are unavailable macroinvertebrates may be collected from submerged vegetation, bottom sediments or piles of leaves. If you sample from submerged vegetation, you are less likely to collect too much debris.

Equipment

(as for kick sampling)

Taking A Sweep Sample

1. Decide the length of stream margin you will sample. As a rule of thumb, three individual samples of 1m in length should be enough.
2. Mark the position of your sampling site on the site map and note any reference points and near the base of the plants.
3. Beginning from the downstream end of the sampling site sweep the net around and through the vegetation. Avoid disturbing sediments where possible. A jabbing or bumping rather than scooping motion usually results in less plant material being gathered in the net. Always move in one direction so that any animals you catch are forced towards the bottom of the net.
4. Use a forward scooping motion to lift the net from the water.



5. Carefully empty the net contents into a bucket part filled (about 5cm) with water. Rinse clinging organisms off the net by splashing with water from the stream. If any plant material has been collected in the net, wash it briskly in the bucket of water before returning it to the stream.
6. After each sampling, wash the net in the stream to remove debris.
7. Transfer some or all of the collected sample into the white tray for sorting.
8. Record the percentage of each habitat type in the reach used for sampling. Note the conditions at the time of sampling, such as higher then usual flows, that may influence the results and interpretation. Also record observations of fish or other fauna.

You can use unvegetated banks and muddy bottom streams if there are no other more suitable types of habitat. To reduce the amount of debris collected with the sample, bump the net along the surface rather than drag it through the soft sediments.

3. Sampling Large Substrate

The following method may be useful if the stream bed is composed of large cobbles and/or boulders. Kick sampling this type of habitat is impractical, and painful.

Equipment

- Kick sampling net
- Sorting tray
- Scrubbing brush

Taking A Sample From Large Substrate

1. Select a shallow riffle area with a depth of 10-30cm.
2. Mark the position of your sampling site on the site map and note any reference points.
3. Begin sampling at the downstream edge of the selected site and move upstream, covering a predetermined area, say 0.25m^2. (The area you decide to sample will vary with bug density. Size your sampling area to allow you to collect a minimum of 50 bugs).

4. Position the net about 0.5m immediately downstream of each of the cobbles or boulders you are going to remove from the stream bed. The net will catch the bugs that become detached as you lift stones from the water.
5. Place the selected rock into the sorting tray in a small amount of water.
6. Scrub the entire surface of the rock, with a sweeping downward motion. This should ensure that most bugs are flicked downwards, into the tray. Place the rock back in the stream when you have removed all the bugs.
7. Continue removing and scrubbing stones until you have collected at least 50 bugs. Alternatively, scrub each rock in front of the net, below the surface of the water. Place each rock to one side before selecting the next.
8. After removing the stones, use the handle of the brush or your foot to stir up the underlying bed, catching dislodged bugs in the net. Use a forward scooping motion to lift the net from the water to prevent any of the organisms escaping. Place this sample into the sorting tray (or bucket if it contains too much debris).
9. Note conditions at the time of sampling, such as higher than usual flows, that may influence the results and interpretation. Also record observations of fish or other fauna.

You can use this method to collect samples from **snags and other woody debris.** Woody material that has been submerged for a relatively long time (more than 6 weeks) provides a suitable habitat for sampling. The snag material may be kicked or surfaces rubbed by hand or brush to dislodge organisms. Large logs should be avoided since they are generally not productive.

Sorting And Identifying Bugs

Equipment

- Sorting tray (containing field sample)
- Bug handling tools, including 'bug sucker', brush and/or forceps
- Bug box
- Identification guides
- Hand lenses
- Sorting table (or flat area)
- Tally sheet
- Pencil.

For Level Two below, you will need the same equipment as for Level One, plus:

- Sorting tray with a grid of 12 numbered 'squares' ruled on the bottom
- Soda water (optional)
- Field microscope (at least 20x magnification)
- Extra bug boxes/containers (to allow you to separate out the bugs into more different types)
- Detailed macroinvertebrate keys and/or identification guides.

Level One - The Beginner's Guide To Boxing Bugs

This is the simplest way to classify bugs into the arrangement on the bug box:

1. Swirl then pour your sample from the bucket into the large, shallow white sorting tray. Spread the sample over the bottom of the tray in a thin layer so that the bottom is still clearly visible. The white bottom of the tray contrasts the bugs, making them easier to distinguish from the debris.
2. Partly fill the cells of the bug box with water. Work out where each type of bug will be placed - the arrangement corresponds to the pictures on the lid. These groupings represent different invertebrate **orders.**
3. Use your bug handling tools to pick the invertebrates out of the tray and sort them into the right places in the box. Forceps can be used on the hard-bodied bugs, the 'bug sucker' for the small fast moving ones and the brush for the larger softies.

   Mystery bugs can be trapped in a small drop of water on the lid of the box if you need to inspect them more closely with the hand lens.
4. Continue sorting until you have at least 50 bugs in the box. Be thorough as you search and try your best to find as many different groups as you can. Don't be satisfied with just the large slow moving ones - go for the little racers too!
5. Check that there are no strays; all bugs should be in their proper place. Size and colour alone cannot distinguish different types, or species. While some species are smaller than others, individuals within a species may vary in size, depending on age.
6. Use the tally sheet (which is set out like the cells in the bug box) to record the number of each of the different types of bug you have identified.

7. Follow the directions in the 'Calculating Indices and Analysing your Results' pages to calculate the indices that suit the goals of your programme. Each of these values provides information about the health of the stream.

If you are not confident about your classification skills it is best to concentrate on simply looking for **different types** of bugs. The Sequential Comparison Index (SCI) indicates the diversity of an invertebrate community, a reflection of the health of the waterway.

You can perform this activity without knowing the names of the bugs, but you will need to be observant and try to look for differences between the bugs in your sample (see comment in 5 above).

1. Pick a bug out of the sorting tray and place it into one of the cells in the bug box.
2. Record a tick in the tally sheet that corresponds to the position of the cell.
3. Randomly select another organism. Is it is the same as the first?

   Yes - Record another tick in the same square and put the bug back.

   No - Place the bug in the next cell and tick the appropriate square.
4. Randomly select another organism. If it is the same as any you have selected so far tick the appropriate square. If it is different place the bug in the next cell and tick the appropriate square.
5. Continue this process until either all the bugs are used or at least 50 have been selected.
6. Count the number of cells used in the box. This is the macroinvertebrate diversity.
7. Count the total number of organisms you have selected.
8. Calculate the Sequential Comparison Index (see 'Calculating Indices and Analysing Your Results') and rate your stream.

Level Two - Taking A Closer Look

At this level you will be entering into the realm of the advanced bug hunter. You will be expected to **distinguish between individuals** within each major group.

For example, the lid of the bug box shows three different types of mayfly. They all belong to the Order Ephemeroptera, but each of the three have enough distinctive features for them to be classified into more specific groups, or species.

This method also refines your sampling process by bringing in **random sub-sampling.**

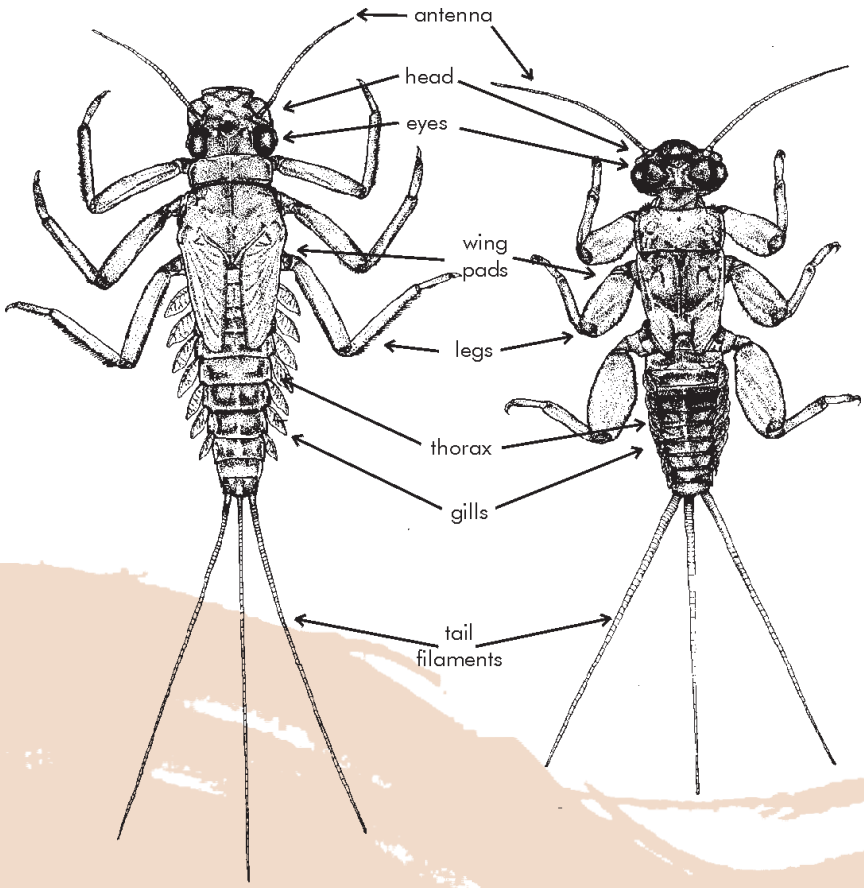
You can use keys, photos or drawings of bugs to help you get to this level of classification. These guides help you to zero in on the key features that distinguish the different bugs. You will need to look carefully to see these differences. The shape of the body and head, the location and arrangement of gills, tails, eyes, mouth parts, antennae, and prolegs may be used to key out a particular invertebrate.

You need to:

- carry out random sub-sampling
- identify bugs to the family and/or genus level
- apply sensitivity ratings to the different species you have collected.

Method

1. Transfer your sample to the shallow white sorting tray.
2. Spread the sample over the bottom of the tray in a very thin layer so that the bottom is still clearly visible. The bugs may need a little rearranging if they are not evenly distributed over the tray, including the corners.



All mayflies are not equal. Look at the range of features highlighted here to distinguish between these different types (genera) of mayfly.

3. You will pick one square at a time until you reach about 50 or 100 bugs total, picking every individual out of each square before moving to the next one. Check that you haven't left any bugs behind under leaves, within clumps of algae or lurking in organic matter.

4. Each time you need to select a square roll a pair of dice so your choice is random (select the next square if you roll the same number twice).

   If you find that the bugs are not 'behaving', and tend to migrate between squares you will need to subdue them. Scientists use alcohol, but this is rather severe causing almost instant death. A gentler approach is to drug your bugs by pouring soda water into the tray. This increases the levels of carbon dioxide and slows their metabolism, and movement, sufficiently. Nicotine may be added to the water to achieve the same result.

5. Partially fill the cells of the bug box with water. Work out where each type of bug will be placed, according to the pictures on the lid.

6. Use your bug handling tools to pick the bugs out of the squares and sort the organisms into the right places in the box.

7. Continue until you have enough bugs, say 50 or 100, sorted into the bug box cells. Make sure that there are no strays. All bugs should be in their proper place in the box. Have someone check your sorting before you tally your sample.

   Remember **size and colouration** alone cannot distinguish different types, or species.

8. Record the number of each of the different types of bug you have identified and their names.

   You may also want to record the number of squares you picked for each replicate bug. You can calculate the density of the sample (see the extension activity).

9. Work out the sensitivity score for each of the invertebrates you have identified and record on the data sheet.

10. Follow the directions in the 'Calculating Indices and Analysing your Results' pages to calculate the indices that suit the goals of your programme. Don't forget to state the species classification level you have used for your assessments on the tally sheet.

Bug Glossary

Abdomen	The posterior portion of the body, made up of similar segments, and containing reproductive organs and part of the digestive tract.
Gills	Respiratory organs of aquatic animals, usually a thin-walled projection from some part of the external body surface.
Larva	A juvenile form of an adult, usually rather different in appearance from the adult.
Proleg	A short, stumpy, unjointed leglike process in certain insect larvae.
Tentacle	Long, flexible protrusions located about the mouth of many macroinvertebrates, usually tactile.

Extension - For The Enthusiast!

This more detailed method will allow you to evaluate the health of your stream using a more precise macroinvertebrate analysis.

The procedures are more complex and may need more skill than the methods described above. However, this level of analysis will enable you to detect more subtle effects on your stream's bug community.

For this method you will have to collect three identical samples per site. Biological surveys don't get much more exacting than this, and you can be sure your biology/science teacher will insist that you do it!

You will need the same equipment as above.

Collecting The Bugs

Follow the collection procedures above, using the method for the type of habitat being sampled, preferably riffle habitat.

If you are sampling riffle habitat you may be able to collect all samples from within one riffle area. (Make sure you work upstream as you sample to avoid disturbing substrate before it is sampled).

If you collect bugs from other habitats besides riffles, collect three for each habitat type. You cannot count samples from different habitats.

Sorting The Samples

Keep the samples separate, sorting and identifying the bugs in each, using the methods above with these changes:

Transfer each sample to a separate sorting tray, each marked with a grid.

The grid lets you estimate the total number of bugs in your sample by picking a sub sample of say 50 or 100 bugs.

For each sample, record the number of squares you picked for your sub-sample. You will use these figures to work out the projected density of the sample.

Bug 'x' density from a sample:

$$= \frac{\text{total no. of bug 'x' picked}}{\text{total no. of squares picked}} \times 12 = \text{projected density}$$

To get the average of your three replicates add the values together and divide the total by three:

$$= \frac{\text{replicate 1} + \text{replicate 2} + \text{replicate 3}}{3} = \text{average projected density}$$

This can be taken a step further if you have collected your sample from a known area, say 1m x 1m.

For example, if you estimate that 63 mayflies were in the sample taken from $3m^2$ ($1m^2$ per replicate sample) then mayfly are present at a density of 21 per m^2.

Use the keys and other references to work out what sort of feeding group (shredders, scrapers, gathering collector or predators) your different bugs belong to. Record this information on the data sheet.

Calculating Indices And Analysing Your Results

Using Biological Indicators To Assess Stream Health

Any of the following indices may be used to assess the health of a particular waterway. Scientists use many different indices to discriminate between degraded and undamaged sites, and each has its strengths and weaknesses. The following are relatively simple to calculate and each will give you some useful information about your stream. You can use a number of different indices if you want to have a more complete understanding of the health of your stream.

Richness Measures

Measuring the health of the community by the total number of different types of invertebrates.

1. Taxa Richness

Count the number of taxa in the whole sample and record on the data sheet. Numbers usually increase with larger samples. (You can easily standardise your richness scores by calculating the richness represented in a particular sample size, say 50 or 100.)

The species richness, or total number of species, tells you important information about the variety of the bug population in your stream. Knowing that there are caddisflies in your stream is useful information, but knowing there are three different families (taxa) of caddisflies is even more useful. The greater the species richness, the larger the variety of your bug population.

In general, streams with a larger range of bugs are considered healthier than those with fewer. However, some organic pollution (excess nutrients from silage pits or animal wastes from a stock crossing) sometimes increases the number of species, especially in high altitude streams that are naturally low in species diversity and number of individuals.

2. EPT Richness

Count the total number of EPT 1 (Ephemeroptera - mayflies, Plecoptera - stoneflies, and Trichoptera - caddisflies).

The EPT richness, or number of mayfly, stonefly, and caddisfly species, provides important information about your stream because these creatures are generally more sensitive to pollution. EPT numbers usually drop with pollution, although some mayflies and caddisflies tolerate some pollution (the bug box pictures shows two exceptionally tolerant ones).

[1] *Taxonomy can be to any level (order, family, genus/species), but should be consistent among samples. State the taxonomic level used in your Biological Survey Record Sheet.*

Many species of midges, sand flies, crustaceans, aquatic worms and snails tolerate more pollution, and tend to move into habitats vacated by mayflies, stoneflies and caddisflies when areas become polluted. This shift tends to simplify and destabilise the structure of the bug community, and reduces the biological soundness of the stream ecosystem.

Although EPT richness values above 12 are considered good, some naturally unproductive high altitude streams may have lower EPT numbers and yet be pristine.

EPT Richness	Stream Quality Assessment
More than 15 families/genera	Excellent
12-15 families	Good
8-12 families	Fair
Less than 8 families	Poor

Composition Measures (or relative abundance)

Measures of relative abundance provide information on the make-up of the community. A community composed of many different types of bugs will score higher values. A stream community that supports a wide diversity of invertebrates is considered healthy, and will be more stable.

1. Sequential Comparison Index (SCI)

Add up the number of different types of organisms and the total number of organisms, then calculate the diversity index:

$$\text{SCI} = \frac{\textbf{Number of different types of bugs in sample}}{\textbf{Total number of bugs in sample}}$$

$$= \frac{\qquad\qquad}{\qquad\qquad}$$

Compare your sequential comparison index with the table below:

SCI	Stream Rating
1.0-0.90	Excellent
0.89-0.60	Good
0.59-0.30	Fair
0.29-0.00	Poor

This index shows the variety and relative abundance of bugs. People unfamiliar with identification of invertebrates use this measure easily. The index is based on the idea of 'runs'. A new run begins each time an invertebrate picked from a sample is different from any already picked. The boxes in the tally sheet are simply ticked as each **new kind of bug** is identified.

2. Percent EPT

Add together the number of mayflies, stoneflies and caddisflies in the sample (i.e. total EPT).

Use the formula to calculate percent EPT:

$$\textbf{Percent EPT} = \frac{\textbf{Number of EPT in sample}}{\textbf{Total number of bugs in sample}} \times \frac{\textbf{100}}{\textbf{1}}$$

$$= \frac{\qquad\qquad}{\qquad\qquad} \times \frac{\textbf{100}}{\textbf{1}}$$

$$= \qquad\qquad \textbf{percent}$$

This index is based on the percentage of pollution 'sensitive' types in the sample. There are some variations in the sensitivity ratings of bugs in these orders, but as a general rule the percent EPT should be highest in unimpaired, pristine streams little affected by organic enrichment.

Compare percent EPT at your different sampling sites.

Tolerance/Intolerance Measures

1. Pollution Tolerance Index

Multiply the number of types of organisms in each tolerance level by the tolerance value[2] for that level (**4,3,2 or 1**).

For example, in a sample that contains mayflies (**4**), stoneflies (**4**), caddisflies (**3**) and some diptera (**1**), the calculation would be:

[2] *Use the tolerance values on the lid of the bug box.*

2 x **4** (2 different types of organisms each with tolerance scores of **4**)

\+ 1 x **3** (1 type of organism with a tolerance score of **3**)

\+ 1 x **1** (1 type of organism with a tolerance score of **1**)

Pollution Tolerance Index = 8 + 3 + 1 = 12

The Pollution Tolerance Index is based on the concept of indicator organisms and tolerance levels. Indicator organisms are sensitive to water quality changes, and respond in predictable ways to changes in their environment. By their presence or absence they indicate something about water quality. This procedure can be used to detect a relatively coarse level of degradation in stream quality.

Compare the index value with the scale below:

PTI Value	Stream Quality Assessment
23 and above	Excellent
17 - 22	Good
11 - 16	Fair
10 or less	Poor

2. SHMAK Index

The Stream Health Monitoring and Assessment Kit uses a scoring method similar to the PTI, but selects only key indicator organisms from the sample. Each of these species is assigned a specific sensitivity/tolerance score. The list of indicator species is slightly different for different parts of the country.

3. Macroinvertebrate Community Index (MCI)

Most regional councils use a more detailed version of the PTI called the Macroinvertebrate Community Index (MCI). Good identification and classification skills are needed for this advanced method. If you want to tackle this index, contact the environmental monitoring staff at your regional council for help.

Biological Survey Record Sheet For Levels 1 and 2

Bug counting using the bug box

RECORD SHEET

Name _____ Group _____

Date _____ Habitat Assessment Completed ☐

Site _____

Mayflies (type 1) = 4	Mayflies (type 2) = 4	Mayflies (type 3) = 4
Stoneflies (type 1) = 4	Stoneflies (type 2) = 4	Stoneflies (type 3) = 2
Caddisflies (type 1) = 3	Caddisflies (type 2) = 3	Caddisflies (type 3) = 1
Beetles = 3	Dobsonflies = 3	Damselflies = Dragonflies = 3
Hemiptera = 3	Crustacea = 3	Mussels = 3 / Snails = 2
Leeches = 2	Worms = 1	Diptera = 1
Flatworms = 2		

Site _____

Mayflies (type 1) = 4	Mayflies (type 2) = 4	Mayflies (type 3) = 4
Stoneflies (type 1) = 4	Stoneflies (type 2) = 4	Stoneflies (type 3) = 2
Caddisflies (type 1) = 3	Caddisflies (type 2) = 3	Caddisflies (type 3) = 1
Beetles = 3	Dobsonflies = 3	Damselflies = Dragonflies = 3
Hemiptera = 3	Crustacea = 3	Mussels = 3 / Snails = 2
Leeches = 2	Worms = 1	Diptera = 1
Flatworms = 2		

Sort:

- Determine the taxonomic level[1] you will use - not all mayflies are equal!
  If you don't see, or notice, any different kinds of mayflies etc record your totals for that group in one of the score boxes.
- Sort the bugs into the positions shown on the lid of the bug box.

Record:

- The number of each type of bug in the squares above.
- The total number of all bugs you have collected[2]. _____
- The number of squares you used (including half squares). _____
- Number of EPT in the sample. _____
- Number of orders represented in each tolerance level (tolerance levels are coloured blue in the above boxes).
  4 _____
  3 _____
  2 _____
  1 _____

Calculate:

- Your selected indices using the tallies you have recorded.

[1] State the taxonomic level you have used to define 'different'. The bug box lid labels represent invertebrate orders (e.g., Order Ephemeroptera and Order Hemiptera), but keys may be used to distinguish invertebrates to the family or genus level (e.g., the mayflies *Coloburiscus* and *Deleatidum*).

[2] Make sure you collect a minimum of 50 bugs.

Biological Indice Record Sheet For Levels One and Two

RECORD SHEET

Richness

Taxa Richness

Number of different types/taxa identified:

_____

Taxanomic level used for identification:

_____

(State order if you used the pictures on the lid of the 'bug' box)

EPT Richness

E (mayflies) _____

P (stoneflies _____

T (caddisflies)_____

Total EPT = _____

(Count the number of taxa you distinguished, not the number of individuals within each group)

Composition

Sequential Comparison Index (SCI)

$$\frac{\text{No. of different types of bugs in sample}}{\text{Total no. of bugs in sample}}$$

SCI = _____

Percent EPT

$$\frac{\text{No. of EPT in sample}}{\text{Total no. of bugs in sample}} \times \frac{100}{1}$$

Percent EPT = _____

Tolerance

Pollution Tolerance Index (PTI)

Tolerance	x	No. of types in each level
4	x	____ = _____
3	x	____ = _____
2	x	____ = _____
1	x	____ = _____

(Use the groupings and tolerance values on the lid of the bug box)

Taxonomic levels

Taxonomic levels used to define:

Taxa Richness _____

EPT Richness _____

SCI _____

Percent EPT _____

PTI *Order*

Other _____

(Aim to be consistent with the taxanomic levels used for classification at each monitoring trip)