In this book you will find
- safety information
- equipment checklists
- instructions for conducting:
  - visual/smells tests
  - water chemistry tests
  - water flow measurement
  - biological assessments
  - habitat assessments
  - procedures for ensuring accuracy
  - pollution clues for each parameter
- blank data sheets for each type of assessment
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2 Things to keep in mind
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10 Monitoring stream water quality and flow
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14 Water clarity (turbidity) test
15 Measuring water clarity
16 Nitrate and nitrite test
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18 Phosphate test
19 Measuring phosphorus
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22 Dissolved oxygen test
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This Field Manual is designed to be used by Wai Care groups carrying out biological, physical and chemical tests in the field. It includes information and instructions for each test, reference values that will help you interpret your results and suggestions of other tests and observations that will confirm your findings. If you are concerned that your results might indicate a problem please follow the procedures outlined in the decision support tables. A laminated summary version has been prepared as Book 4 'The Field Handbook' for a ready reference guide once you are familiar with the tests.

The tests outlined in this manual follow carefully developed procedures which will give reliable results if followed closely. The tests are generally simple and easy to perform but are capable of generating very useful information about the state or health of any waterway.

A set of record sheets is included. Photocopy the set that you require for your Wai Care project and store completed sheets systematically. Suggestions for data storage, presentation and interpretation can be found in Book 5 'Data Management and Interpretation'.
Before you begin your Wai Care project, there are a few things to keep in mind:

**Courtesy**
- Ensure that your local Iwi/Hapu/Marae have been involved as part of your planning process.
- If you must cross private property to reach your monitoring site, first seek the property owner’s permission.
- If you need to cross any farmland remember to close all gates after you. If you need to climb a fence, do this at a post to avoid damage.
- Provide property owners with a copy of the results.

**Your environment**
- Minimise damage to stream banks and beds at monitoring, entry and exit points.
- Return all live sample organisms to the stream.
- Take away more litter than you brought in.
- Avoid sampling macroinvertebrates from any site more than four times a year.
- All chemical waste used in water quality tests should be collected in a leak proof bottle and disposed of into the sanitary sewer system.

**Safety and health**

* a. Sampling sites
  - When selecting a site, choose one that has safe and easy access.
  - Check that banks are not slippery or unstable. Floods often damage banks - check for change regularly.
  - Complete a safety/hazard checklist for each site and include these with your record sheets, as a reminder of site specific safety considerations.
  - Do not enter the water barefoot and be careful of hidden objects, holes, prickly vegetation etc. Never put your feet in places where you cannot see.
  - Conduct macroinvertebrate sampling in water less than knee deep.

* b. Gloves and safety gear
  - Wear gloves so you don’t contaminate yourself with polluted water or with chemicals used for testing. Some procedures also require safety glasses.
  - Wear suitable clothing and footwear, including sturdy waterproof shoes with a good grip and waterproof gloves. Dress for the weather and don’t forget your hat and sunscreen in summer.
  - Bring a fully stocked first aid kit with you to the sampling site, including sterile saline eyewash.
Things to keep in mind

- If ever sampling near water that is deep or fast flowing wear a life jacket. If you do fall in, swim across the current to safety, not against it.

**c. Safety in numbers**

- Never survey alone. It is best to work with at least two others. If one of you is injured, one person can go for help and one can stay with the injured person.
- Let someone else know where you will be sampling and approximately how long you will be gone.
- Do not allow children to sample without adult supervision.
- Know where the nearest phone is located, carry a cellphone if possible or carry coins/phonecards in case you need to make an emergency call.

**d. Others**

- Carry water with you to drink. Do not drink water from the water sources you are testing as it may be polluted. In particular, when sampling in urban areas, do not put your hands near your mouth, eat, drink or smoke while testing the water.
- Do not sample if you have broken skin (cuts etc.).
- Put all solid waste such as used gloves, empty reagent containers and any other rubbish from the day into a rubbish bag.
- If your site is a heavily vegetated area wear bright easily visible clothing.

If many of these risks are not easily overcome then consider using the visual/smells check method as your main form of assessment. You may be able to link in with another monitoring group or agency to obtain other kinds of data.

**Before you take the plunge......**

A quick visual/smell survey is advisable before sampling from any site. Many pollutants can be detected in this way and will help you to decide the method of sampling you should use. Pollution events should be quickly reported to the appropriate authority, and evidence collected if possible. If you have access upstream you may be able to locate the source of the pollution.

**After you take the plunge......**

Complete and check field record sheet entries at each site. You may need to repeat some tests if the values appear unusual. When you return from the monitoring trip enter your data into your database, or use a systematic method for storing the sheets. Information about data management can be found in Book 5 'Data Management and Interpretation'.
Protecting property owners

Under the Occupational Safety and Health Act (1992) all landowners (e.g. industrial or commercial operators and farmers) have a duty to warn visitors of any significant hazards that they know of on their property. This Act has led to most landowners having much more stringent rules about visitors and makes them unwilling to allow non-work-related visitors to their properties.

For site owners to fulfil their obligations under the Act and safely allow groups and individuals to visit their properties the owner will need to complete a hazard checklist. This is one reason why many property owners may be reluctant to allow you or your group access to their properties unsupervised. If contact has been made before, it is advisable for you to contact the property owner a day or two before the trip to ensure that no additional hazards have cropped up since your last communication.

The field trip and equipment checklist will help you prepare for your monitoring trip and act as a reminder to cover all the safety aspects.
**Field trip and equipment checklist**
(please photocopy)

| Date |
| Field Handbook |   |   |   |
| Data Record Sheets |   |   |   |
| First aid kit |   |   |   |
| Pens, pencils |   |   |   |
| Marker pen (waterproof) |   |   |   |
| Sticky labels |   |   |   |
| Sample bottles |   |   |   |
| Waste container |   |   |   |
| Rubbish bag |   |   |   |
| Sun cream and hat |   |   |   |
| Drinking water and food |   |   |   |
| Camera and film |   |   |   |
| Emergency telephone number |   |   |   |

**Chemical/physical tests**

| Armoured thermometer and bucket |   |   |   |
| Water clarity tube |   |   |   |
| Nitrate test strips and colour chart |   |   |   |
| Phosphate test kit |   |   |   |
| pH test strips |   |   |   |
| DO test kit |   |   |   |
| Stopwatch, meter ruler (or head rod) and 20m tape |   |   |   |
| BOD bottles |   |   |   |
| Filter paper |   |   |   |
| Distilled water |   |   |   |
| Safety glasses and gloves (as required for test kits) |   |   |   |
| Paper towels |   |   |   |

**Biological assessment**

| Kick net |   |   |   |
| Bucket |   |   |   |
| White sorting tray |   |   |   |
| Bug box |   |   |   |
| Bug suckers and/or soft paint brushes |   |   |   |
| Identification guides |   |   |   |
| Magnifier |   |   |   |

**Before you leave the site, check the following**

| Is the equipment cleaned? |   |   |   |
| Is there any rubbish left behind? |   |   |   |
| Has the stock in the kit been checked? |   |   |   |
| Is any equipment broken or lost? |   |   |   |
Wai Care assessments

Your visual/smells check

Most water monitoring groups use equipment to collect information, however this is not always necessary. Two of our basic senses, sight and smell, are incredibly powerful when combined with common sense and are capable of collecting very useful information about our chosen stream. In fact we would suggest that any monitoring group use this basic assessment method before they go anywhere near the water to conduct other tests.

This simple assessment is based entirely on things you can observe and smell. Conducting a visual/smells test is so simple and quick that you could consider doing it regularly, for example, each time you walk the dog or go to the dairy - the more often the better. Pollution events can happen in an instant, and early warning of such an event can help minimise the extent of damage and help trace the source more easily.

The areas of the stream to check can be remembered by the term “SoSMART” which stands for:
- S-mell
- o-bstruction
- S-tream bed
- M-argin
- A-pearance
- R-ate
- T-op

Each of these 7 areas of your stream could show the effect of a pollution event. On the record sheet, there are criteria for each area that you may wish to use to rate how degraded your stream is. Over time this may be a useful tool to track recovery.
Scoring

<table>
<thead>
<tr>
<th>Score</th>
<th>Description</th>
</tr>
</thead>
<tbody>
<tr>
<td>0</td>
<td>Indicates a severe problem</td>
</tr>
<tr>
<td>1</td>
<td>Indicates there is a minor problem</td>
</tr>
<tr>
<td>2</td>
<td>Indicates no problem</td>
</tr>
</tbody>
</table>

These values are added to give a total score out of 14, the lower the score the more compromised your stream is by chemical or physical influences. Any severe problem (score of 0) should be related to your Wai Care co-ordinator as soon as possible.

The “SoSMART” criteria

Although most of the observations are straightforward and can be committed to memory in the field it is well worthwhile recording these on paper when you get home. Reference notes can be helpful when you are looking for change over long periods of time, or are attempting to look for patterns. The visual/smells record sheet can be used, or modified, to record your observations. Or if you do witness a pollution event, immediately reporting it to your local or regional council pollution hotline will help the source be identified.

S-mell

This can be the first warning that something is not right. Ensuring that the smell is actually coming from the stream and not a nearby land based source is important and may require some ingenuity or simple equipment. The classic problem in urban areas are sewage smells venting from the sewer manholes next to the stream being mistaken for pollution of the water itself. Collection of a sample of the water for a better sniff using a bucket or other container will help to confirm the source. Where the stream bed sediment potentially harbours the source of the smell, sticks can be used to stir it up for confirmation. You may be able to think of other simple practical devices that meet your particular sampling requirements.

Many other aromas may be wafting around and may prove more difficult to locate. The ‘smells’ table in the record sheet has grouped common odors into types, but you will no doubt want to add some of your own as you become more familiar with the pollutants in your area.

O-bstruction

These could be blocking the flow of water causing it to pond and thereby reduce dissolved oxygen and also provide a further trap for rubbish to collect. Where materials get trapped against or inside pipe or culverted sections of stream, serious flooding problems can result if they aren’t cleared.
Stream bed

Look for materials that cover the bed of the waterway, such as sediment, slime or scum, water plants and the colour of the covering material. In particular look for material that appears to be smothering the bed. For example, if the streambed is rocky but is being covered in sediment then record this, and the approximate depth of the covering. Accumulations of organic matter and ‘dead’ stream life should also be noted.

Margin or bank

This part of the stream environment is very important for stream health. The ‘bank’ may be in a condition that protects the stream from adjacent threats, or it may provide the pathway for polluting materials to enter the stream.

Look for signs of erosion (and whether this is ‘fresh’), debris/rubbish, and potential or actual pollution sources. Changes in vegetation should also be noted.

There may be other undesirable material on the edge as well - people often use these areas to dump all sorts of junk. Please contact your Wai Care co-ordinator to advise of the presence of waste or if you have some concern about the presence of pipes or drains that look like a private and maybe an inappropriate connection.

Appearance of the water

The two important indicators of stream water appearance to look for are colour and murkiness. Both can vary with flow but it is possible to establish a ‘feel’ for the acceptable, natural range through frequent observations in different conditions. Some colours are obviously unnatural, and water generally clears quite quickly after rainfall ceases, especially in smaller catchments. Persistent murkiness or discoloration should be reported and/or investigated further especially when this occurs outside of rainfall events.

Rate

How fast is the water flowing? This will change considerably and quickly with rain but could affect the other factors of the stream. It is therefore important to record the weather and previous rainfall on your sheet. Water may be pumped out of streams during dry summer conditions leaving little for aquatic life.

Top of the water

Water is a great solvent but many pollutants do not dissolve or not completely and so remain floating on top. These floating materials can cause serious short-term damage (toxic effects) to stream life and where they persist in the environment, long-term damage. In urban areas these forms of pollution are all too common and are found as oily films, rainbow coloured sheens and globs, scums and foams.
SoSMART Assessment Record Sheet

<table>
<thead>
<tr>
<th>CATCHMENT NAME</th>
<th>GROUP NAME</th>
</tr>
</thead>
<tbody>
<tr>
<td>DATE</td>
<td>TIME</td>
</tr>
<tr>
<td>CURRENT WEATHER</td>
<td></td>
</tr>
</tbody>
</table>

**PREVIOUS RAINFALL** (circle) Within 24 hours 1 - 7 Days > 7 Days

**RAINFALL DURATION** (circle) Light Medium Heavy

**SITE CHANGES** (if any since last visit) / COMMENTS

### 1. SMELL
- e.g. Sewage and/or grey water
- Chlorine/chemical
- Petroleum/ solvents
- Organic - including dead things
- Detergent

### 2. OBSTRUCTION
- e.g. Weeds
- Large woody debris
- Rubbish
- Organic materials

### 3. STREAM BED
- e.g. New concrete
- Sediment covering
- Slime and /or scum
- Colour of covering

### 4. MARGIN
- e.g. Erosion
- Pollution source
- No vegetation
- Litter

### 5. APPEARANCE
- e.g. Murkiness
- Muddy
- Colour state
- Bubbles

### 6. RATE
- None 😞
- Slow 😞
- Fast 😞

### 7. TOP/SURFACE
- e.g. Oily film/sheen
- Globs
- Slime /algae bloom
- Scum/foam

### Score Table

<table>
<thead>
<tr>
<th>Site</th>
<th>Score</th>
<th>Site</th>
<th>Score</th>
<th>Site</th>
<th>Score</th>
</tr>
</thead>
</table>

**SCORE**

- 😊 2 = no problem
- 😞 1 = minor problem
- 😞 0 = severe problem

**SITE**

**SCORE /14**

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FIELD MANUAL
Wai Care assessments
Monitoring stream water quality and flow

The range of tests outlined in the following section are almost universally accepted as the most useful and practical tests for determining water quality and as indicators of pollution.

Typical pollutants are excess nutrients (nitrates + phosphates), oxygen demanding substances and suspended sediment (reduced clarity). But you also want to know the amount of dissolved oxygen, the water temperature and the pH because these tell you whether the water is capable of supporting aquatic fauna and flora - fish, invertebrates, water plants and algae, if the right habitat is present.

Biochemical oxygen demand (BOD) tells you how quickly the oxygen in the stream is being used up by bacteria and by chemical processes. If the BOD is high then you know that there will not be very much oxygen available for fish or other stream life.

Pathogens such as faecal coliforms and other associated bacteria and viruses are also major contaminants in many Auckland urban streams due to stormwater contamination or failure of the sewage collection network. Monitoring for bacterial indicator organisms requires specialised equipment and a higher level of expertise than most other tests. Therefore there is no proposal for universal inclusion of a bacterial test at this time. In special circumstances where Council or school laboratories are set up to provide appropriate support Wai Care groups may be able to undertake bacterial assessments in the future. Check with your Wai Care co-ordinator to find out if this test is available to your group.

Taking your sample

Where?

- Take the water samples as far away from the edge of the bank and as close to mid-stream as possible. Unrepresentative samples are often gathered from water at the edge: it is likely to be warmer, sediment is more easily stirred up and aquatic plants may trap a greater proportion of pollutants.

- If collecting samples while standing in the water, always take the sample upstream of where you are standing to avoid disturbing the bed and releasing sediments. Potential contamination from skin and clothing will also be avoided by sampling in this way.

- If you are working as part of a group it is important that some activities do not interfere with others. For example, water clarity monitoring should be upstream of any other activity to avoid contamination with sediment.

How?

- All sample bottles and buckets should be rinsed twice with stream water prior to collecting samples for testing.

- Keep samples out of direct sunlight.
If any samples are to be analysed at a later time it is essential to label all samples immediately on collection with the site name or site code, date and time of sampling.

A long-handled grab sampler could be useful if flowing water is not easily reached at your site/s (such a device is not provided as part of your kit but instructions on making one will be available from your Wai Care co-ordinator).

**Sample preservation and storage**

All water samples should be tested as soon as possible after collection. Where testing cannot be performed straight away, changes due to biological activity, physical changes or chemical reactions can be prevented by:

1. Filling to the top of the container before capping to prevent loss of dissolved gases, particularly oxygen;
2. Storing samples in the dark to stop photosynthesis;
3. Cooling the sample to reduce biological and chemical reactions.

Recommended sample storage and preservation techniques used by analytical laboratories are shown in the table below:

<table>
<thead>
<tr>
<th>Test factor</th>
<th>Container</th>
<th>Preservation</th>
<th>Storage time (maximum)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Conductivity</td>
<td>P,G</td>
<td>Refrigerate</td>
<td>28 days</td>
</tr>
<tr>
<td>pH</td>
<td>P,G</td>
<td>Analyse immediately</td>
<td>2 hours</td>
</tr>
<tr>
<td>Turbidity</td>
<td>P,G</td>
<td>Analyse immediately or store in the dark. Refrigerate.</td>
<td>24 hours</td>
</tr>
<tr>
<td>Temperature</td>
<td>P,G</td>
<td>Analyse immediately</td>
<td>No storage</td>
</tr>
<tr>
<td>Dissolved oxygen BOD</td>
<td>G</td>
<td>Analyse immediately or “Fix” sample</td>
<td>8 hours</td>
</tr>
<tr>
<td>Nitrogen</td>
<td>P,G</td>
<td>RASAP or refrigerate</td>
<td>48 hours</td>
</tr>
<tr>
<td>Phosphorus</td>
<td>G - rinsed with 1:1 HNO₃</td>
<td>Refrigerate</td>
<td>48 hours</td>
</tr>
</tbody>
</table>

G = glass, P = plastic (polythene or equivalent)

*Always ensure that the test kit chemicals have not exceeded their “use-by-date” before attempting to undertake chemical analyses.*
What do the results mean?

Compare your results to the reference values provided on the decision support sheets in book 4, the ‘Field Handbook’ and values for typical Ruckland streams, provided in book 6 ‘Fact Sheets’.

You can convert clarity tube distances into turbidity readings using the graph provided in the Appendix to this book. Similarly dissolved oxygen readings can be converted into saturation using the graph provided in the Appendix.

Thorough analysis of the information you have collected is best undertaken by using book 5 ‘Data Management and Interpretation’.

Temperature test

Pollution clues and supporting tests

Temperature variations and range are important aspects of water quality: water temperature is unlikely to increase by more than 3°C in a day.

Unshaded Ruckland urban streams have been found to range between 10°C or less in winter up to 26°C in summer. By comparison urban streams with shading vegetation range between 8.5-21.5 °C

Check DO which decreases as water temperature increases.

Take a series of readings throughout the day.

Equipment

- An alcohol filled graduated thermometer in a protective casing.

Avoiding errors

- Do not collect the sample from the edge of the stream.
- Check the air temperature as a rough guide to water temperature readings.
- Use the same thermometer each time to take the readings.
- Repeat the reading a few times, returning the thermometer to the sample water between readings.
- To compare with previous results collect at the same time each day.
Measuring water temperature

1. Place the thermometer in the shade near the stream for several minutes to stabilise the temperature.
2. Read the graduations and note down the air temperature on the record sheet.
3. Carefully collect a bucket of water from the stream.
4. Immerse the lower part of the thermometer in the bucket for 1-2 minutes then remove and quickly read the graduations and note down the water temperature on the record sheet.
5. Rinse off the thermometer and place it back in the kit.
6. Compare the values recorded to the decision support table to determine if a water quality problem exists.

Most aquatic animals will thrive in naturally cool water

Effects of higher temperatures

- Warmer water will stress most aquatic animals and then they become vulnerable to disease and other problems.
- 'Good' water bugs aren't generally found in water warmer than 19 degrees. Temperatures above 21 degrees will kill many water bugs.
- Higher temperatures help plants grow more vigorously. Algal blooms can be started and the water then becomes eutrophic.
- Higher temperatures mean less oxygen in the water. Unfortunately, as the temperature increases the water bugs become more active which increases their need for oxygen.
Water clarity (turbidity) test

Equipment

Water clarity is measured in a water clarity tube.

Avoiding errors

- Make sure you do not disturb bottom sediment when collecting your sample.
- Keep the viewing window free of scratches that might distort readings.
- Check that the viewing window is clear of condensation.
- Take clarity readings in diffuse sunlight or the shade but avoid patchy light with shadows. If it is impossible to avoid bright sunlight, make sure that you work with the tube at right angles to the sun.
- You should not take readings in very low light conditions.

Note: The most accurate readings will be those up to around two-thirds of the tube length.

Pollution clues and supporting tests

The appearance of the water determined by the SoSmart check will be a useful supportive test for the clarity reading. Often you will know that the clarity is poor just from your observations without taking a measurement.

Urbanised Auckland stream turbidity ranges between 4-85 NTU, which equates to clarity tube readings of less than 10cm through to 70cm respectively.

Catchments where earthworks are occurring can have turbidity values up to 550 NTU.

Effects of murky water

- aquatic animals suffer from blocked gills
- habitats are silted up making them unsuitable for many animals
- fish egg hatching and larval development is interfered with
- water heats up because murky water absorbs more sunlight
- reduced light means that less photosynthesis occurs in plants, so less oxygen is produced
- animals that rely on vision to catch prey are less successful (and hungry)
1. Collect a bucket of water from the main flow of the stream, upstream of any disturbance made by your group.

2. Fill the tube completely then place the black disk magnet inside and stopper the end securely.

3. Support/hold the tube horizontally and move the black disk to the plugged end of the tube.

4. View through the clear end with your eye close to the viewing window, like looking through a telescope.

5. Move the black disk toward you using the magnet until it is clearly visible.

6. Move the black disk carefully back away from you until it just disappears from view. Using the graduations marked on the tube, read off the distance from the clear viewing end to the disk.

7. Repeat steps 3-6 to obtain at least two further readings, shaking the tube before each to ensure any sediment remains in suspension. Get another group member to take a reading if possible.

8. Use the average clarity reading for recording on your record sheet.

9. Using the water clarity vs turbidity conversion table on p50 of this book, convert your result to NTU’s.

10. Compare the values recorded to the decision support table to determine if a water quality problem exists.

11. Ensure you collect the black disc magnet from the tube as you tip the water out.
Nitrate and nitrite test

Equipment
Hach nitrate dip strip indicator papers and colour chart.

Avoiding errors
- Test as soon as possible after collecting the sample from the stream before biological activity in the sample converts nitrogen to another form.
- Measure at the same time of the day to avoid any variations which may be influenced by plant activity.
- Check the ‘dip strip’ use-by date on the container to ensure that they do not need to be refreshed.
- Ensure that more than one group member does the colour match as colour blindness or age will affect the ability of some members to provide accurate results.

Pollution clues and supporting tests
Unpolluted waters generally have a nitrate level of less than 2 mg/L although in some horticultural areas the levels can be much higher (above 10 mg/L). Nitrite levels are generally very low.

A phosphate test may be useful to determine whether the source of the problem is likely to be from a wastewater discharge or not.

Effects of high nitrate levels
- Nitrate is a plant nutrient and as such will cause an increase in the growth of algae (blooms) and other aquatic plants. Abundant growths may restrict flow and lead to increased water temperature and pH
- Depletion of DO as plant matter rots
- Drinking water containing excess nitrates can cause some health effects in infants

Effects of high nitrite levels
- Nitrite is a useful indicator of the proximity of a waste source as it is rapidly converted to nitrate under normal conditions
Measuring nitrogen levels

1. Remove a single ‘dip strip’ from the container without contaminating or wetting the other strips and replace the cap.

2. Dip the strip into the sample water for one second. Do not shake the strip.

3. Hold the strip level with pads facing up for 30 seconds.

4. Compare the NITRITE test pad to the colour chart on the bottle.

5. At 60 seconds compare the NITRATE pad (at the end of the strip) to the colour chart on the bottle.

6. Record these values on the record sheet.

7. Compare the values recorded to the decision support table to determine if a water quality problem exists.
Phosphate test

Equipment
Aquaspex phosphate kit.

Avoiding errors
- Test as soon as possible after collecting the sample from the stream.
- Ensure that more than one group member does the colour match.
- Make sure that the chemicals added to the water sample are carefully and thoroughly mixed at each step of the operation.
- Compare colours in diffuse daylight. Avoid direct sunlight and artificial light.

Pollution clues and supporting tests
Water in urbanised streams generally has a soluble phosphate level of around 0.1 mg/l. Both phosphorus and nitrate are required to stimulate excessive plant growth so problems are more likely where both are present.
- Test for nitrate levels and look for signs of excessive growth of algae and stream plants (periphyton and macrophytes).
- Also check water clarity (phosphorus can enter the water with soil particles), smell (sewage/animal wastes contain phosphorus) and look for evidence of detergent waste (foam).

Phosphate is a plant nutrient and is often a limiting factor for plant growth

Effects of high phosphate levels

- Increase in the growth of algae (blooms) and other aquatic plants. Abundant growths may choke the waterway, restricting flow and reducing penetration of light, leading to increased water temperature
- Depletion of DO due to night time respiration and as plant matter decomposes (and hence increased BOD)
- Eutrophication
- Altered habitat leading to reduced aquatic animal and plant diversity
1. Rinse both test tubes several times with the water to be tested. Then fill each to the 5ml mark.

2. Place one test tube in one of the holes in the foam block without a cap. This is the reference sample or control.

3. To the other test tube add 7 drops of reagent A, cap the test tube and mix by inverting several times.

4. To the same test tube, add 1 drop of reagent B, cap the test tube and shake vigorously to mix. (reagent B is a highly viscous liquid. Hold the bottle vertically and squeeze gently in order to produce a well developed droplet.)

5. Allow to stand for 5 minutes for full colour development. Remove the cap and place the test tube in the remaining hole of the foam block. This is the measurement sample.

6. Place the foam block on the colour comparator with the reference sample in the top position (ie. on same line as the coloured circles) and the measurement sample below on the white circles. Look from above and move the foam block across the colour fields until the colours of both solutions match.

7. Read the corresponding concentration of phosphorus in mg/l. In order to express result as phosphate, multiply by 3.07.

8. Compare the values recorded to the decision support table to determine if a water quality problem exists.

Note: The measuring range can be extended by taking a smaller water sample and diluting to 5ml. Example: Take a 1ml sample in each of the test tubes and fill both up to the 5ml mark with demineralised water. Proceed as per instructions from point 2. Then multiply the reading by 5 (mg/l).
Water pH test

Equipment
pH ‘dip strip’ indicator papers and colour chart.

Avoiding errors
- Test as soon as possible after collecting the sample from the stream before biological activity in the sample changes pH.
- Measure at the same time of the day to avoid any variations which may occur due to plant activity.
- Check the ‘dip strip’ use-by date on the container to ensure that they do not need to be replaced.
- Ensure that more than one group member does the colour match.

Pollution clues and supporting tests

The pH value in your stream should not show large changes over a period of weeks or months. Auckland urbanised streams generally range in pH between 6.8-8.3. You also would not expect to find big differences between different branches of the same stream.

Both human activities and local geology affect the pH of water.

pH readings of more than 8 are unusual in streams and may be caused by abundant plant growth in summer or discharges of lime from concrete laying activities. Check the streambed for a white coating that may indicate lime.

Swampy areas can result in slightly acidic discharges and these are often accompanied by iron staining in the water and the presence of a fine orange floc.

Effects of high or low pH
- Acid water causes fish and other aquatic organisms to suffer from skin irritations, tumours, ulcers and impaired gill functioning
- Alkaline water irritates sensitive membranes such as skin or gills
- Extremely high or low pH levels will lead to death of aquatic life
Measuring pH

1. Remove a single ‘dip strip’ from the container without contaminating or wetting the strips. Recap the container.

2. Dip the strip into the bucket of sample water for 1 second.

3. Wait for one minute for full colour saturation to develop.

4. Lay the dip strip alongside the colour indicator levels marked on the ‘dip strip’ container. Read off the best colour matches for pH and record these values on the record sheet.

5. Compare the values recorded to the decision support table to determine if a water quality problem exists.
Dissolved oxygen (DO) test

Equipment

- Hach Dissolved Oxygen Kit
- Thermometer

Avoiding errors

- Sample below the surface and well away from the edge of the waterway.
- Sample at about the same time of the day because light (photosynthesis) and temperature affect DO levels.
- Measure water temperature so that you calculate percentage saturation.
- Avoid bubbling air into the sample bottle when you collect it.

Pollution clues and supporting tests

A dissolved oxygen test tells us precisely how much oxygen is dissolved in water, but it does not indicate how much DO the water is capable of holding at that water temperature. The percentage saturation is a better measure of the availability of oxygen to aquatic organisms.

Scientific studies suggest that around 60% is the minimum saturation that will support a large and diverse fish population. The DO level in good fishing waters generally averages about 80-90%. When DO saturation drops below about 40%, even the hardy fish die.

Depressed DO levels may be found if the sample is collected in the early morning where there are a lot of stream plants due to night time respiration.

Effects of reduced levels of dissolved oxygen

- May result in loss of the more ‘sensitive’ species including mayflies, stoneflies and caddisflies
- At very low DO levels, only a very few hardy species such as sand fly larvae may be present, but these are often present in great abundance. Nuisance algae and anaerobic organisms (that live without oxygen) may also become more prolific
Measuring dissolved oxygen (DO)

1. To avoid contamination thoroughly rinse the plastic sample beaker with sample water. Do not return the rinse water to the sample as this will agitate the water and introduce oxygen into the sample - tip the rinse water away.

2. Carefully fill the sample beaker with sample water taking care not to agitate the water and introduce oxygen into the sample.

3. Carefully pour 10 ml of the sample water into the zeroing vial, screw on the lid and place into the left top opening of the colour comparator.

4. Place the plastic ampoule opener into the plastic beaker.

5. Take an ampoule containing the powdered DO chemical and place tip down into the slot of the opener in the plastic beaker containing the sample water.

6. Push down gently on the ampoule to break the tip and allow the vacuum to suck up the correct amount of sample water.

7. Fill the blue rubber ampoule cap with sample water and place it under the broken tip of the ampoule.

8. Shake the ampoule for around 30 seconds to ensure that the powder and sample water are fully mixed and wait 2 minutes for the colour to develop.

9. Shake the ampoule again and then place upright in the right top opening of the colour comparator.

10. Direct the comparator toward a light source and look through the openings in the front rotating the disk until a best match is established.

11. Record the value on your record sheet and compare the value to the decision support table to determine if a water quality problem exists.

Note: You can convert DO to % saturation using the method provided in the Appendix.

Collect all your waste in a container to dispose of appropriately once you leave the sampling site.
Biochemical oxygen demand (BOD) test

Equipment

- BOD bottle - 300ml glass bottle with a ground glass stopper
- Hach Dissolved Oxygen Kit (or other equipment for measuring DO levels)
- A black plastic bag with twist tie to seal the top

Pollution clues and supporting tests

Unpolluted natural waters will have a BOD of 2 mg/L or less. Polluted stormwater runoff from urbanised areas may have a BOD reading of up to 15 mg/L. Increases in BOD result from increasing amounts of organic material in the water, and indicates the potential for depletion of dissolved oxygen.

- Look for sources of organic matter, such as excessive algal growths and stream plants (macrophyte), garden/plant waste and other debris.
- Sewage and some industrial wastes may also be evident in your visual/smells test.

Avoiding errors

- Collect your sample carefully, as outlined in the DO test.
- Make sure there are no air bubbles in the BOD bottle.
- Keep the temperature relatively constant. A light bulb will provide sufficient warmth.

Effects of high BOD

- When the oxygen use or demand by micro-organisms or chemical breakdown is high there is little available oxygen for other larger aquatic organisms

Measuring biochemical oxygen demand (BOD)

Sample collection

1. Collect the sample from an area that is representative of the stream, (i.e. not below a waterfall or from stagnant pools or backwaters).

2. The BOD water sample must be taken at the same time and place as the DO sample.
Measurement

1. Rinse the BOD bottle with stream water before collecting the sample.
2. Submerge the bottle to near the top of the neck in the sample water and carefully tilt the bottle to one side allowing the sample water to gently trickle in with the minimum of disturbance to avoid introducing more oxygen.
3. When the bottle is completely underwater (and full) stand it upright and allow any trapped air bubbles to escape.
4. Tap the sides of the bottle to dislodge any air bubbles clinging on.
5. Insert the glass stopper and remove the bottle from the water.
6. Twist the glass stopper several millimetres in a clockwise direction to ‘lock’ it in place. Create a seal at the neck of the bottle by squirting distilled water around the stopper.
7. Label your bottle with the date, time and place of collection, and the mg/L of dissolved oxygen measured on day 1.
8. Place the sample bottle in the black plastic bag and store for 5 days at about 20°C.
9. After 5 days, carefully and slowly fill the DO sample beaker from the BOD bottle and carry out the DO test. The result is the DO level on day 5.
10. Subtract the amount of DO on day 5 from the amount of DO in the sample on day 1.

An example calculation can be found in the appendix.

Total coliform and E.coli test

Total coliforms and E.coli are indicator micro-organisms that may provide a measure of faecal contamination from sewage or animal dung in stormwater runoff. Coliforms and E.coli are normally not pathogenic (illness causing) but are naturally found in the gut of warm blooded animals and therefore their presence could be associated with pathogenic micro-organisms. However high levels of coliforms can be present naturally in runoff from the bacteria naturally present in our environment.

Bacteriological sampling is not part of the Wai Care standard set of parameters and is only available where specialised equipment is available, such as some school laboratories or the North Shore City Council Bacteriological Laboratory.

Equipment

Samples must be collected in sterile watertight containers of at least 100mL volume. The samples are grown using the colilert system in a laboratory.
Sample collection

1. Label the bottle with the site number, date, and time.
2. Remove the cap from the bottle just before sampling. Avoid touching the inside of the bottle or the cap. If you accidentally touch the inside of the bottle, use another one or triple rinse the bottle with sample water.
3. Try not to disturb bottom sediment when collecting your sample. Do not analyse a water sample that has sediment from bottom disturbance.
4. Stand facing upstream. Collect the water sample on your upstream side, in front of you. You may also tape your bottle to an extension pole to sample from deeper water or collect a bucket of water and sample from that. If using a bucket ensure it is carefully rinsed with stream water before sampling.
5. Hold the bottle near its base and immerse (sink) it, mouth first into the stream or collected water.
6. Do not fill the bottle completely (so that the sample can be shaken just before analysis). Recap the bottle carefully, remembering not to touch the inside.
7. Fill in the bottle number and/or site number on your field data sheet. This is important because it tells the lab co-ordinator which bottle goes with which site and enables you to recognise your site information.
8. If the samples are to be analysed in the lab, place them in a cooler for transport to the lab.

Avoiding errors

- Make sure the sample container is sterilised either by autoclaving or in a pressure cooker at 121°C for 15 min or boiling for 30 min.
- It is very important that the entire process is carried out in sterile conditions. If the sample comes into contact with skin or a non-sterile container, your sample may be contaminated and your results could be distorted.
- Keep the samples chilled with ice packs and process them within six hours, or twelve hours if kept in a fridge. If the samples are not chilled bacterial growth can occur distorting the bacterial levels in your sample.
- Make sure the reagent packs are not past their use-by date. If so, let your Wai Care co-ordinator know. It can take a couple of days to replace water and reagent packs.
- Make sure that as few people as possible are involved with taking, transporting and processing the samples. This reduces the chance of contamination.

Effects of high coliform and E.coli test

- Contaminated water can be a health risk and cause disease for humans and other animals
- Uses up dissolved oxygen in stream and therefore decreases plant and animal numbers
### Water quality data sheet

<table>
<thead>
<tr>
<th>Test</th>
<th>Units</th>
</tr>
</thead>
<tbody>
<tr>
<td>Air temp</td>
<td>°C</td>
</tr>
<tr>
<td>Water temp</td>
<td>°C</td>
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<td>cm</td>
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<td>Turbidity</td>
<td>NTU</td>
</tr>
<tr>
<td>pH</td>
<td>1-14</td>
</tr>
<tr>
<td>Dissolved Oxygen</td>
<td>mg/L</td>
</tr>
<tr>
<td></td>
<td>% sat</td>
</tr>
<tr>
<td>BOD</td>
<td>mg/L</td>
</tr>
<tr>
<td>Nitrate</td>
<td>mg/L</td>
</tr>
<tr>
<td>Nitrite</td>
<td>mg/L</td>
</tr>
<tr>
<td>Soluble phosphorus</td>
<td>mg/L</td>
</tr>
<tr>
<td>Total coliforms</td>
<td>No./100ml</td>
</tr>
<tr>
<td><em>E. coli</em></td>
<td>No./100ml</td>
</tr>
<tr>
<td>Stream flow</td>
<td>m³/sec</td>
</tr>
</tbody>
</table>
Stream flow

a. Float method

Equipment
- Tape measure
- Stopwatch
- Orange, tennis ball or film canister (part filled with water)

Avoiding errors
- Select relatively straight lengths of the stream
- If you get results that vary widely repeat the measurement/s
- Avoid using a stick or leaf, particularly if its windy

b. Head Rod method

Equipment
- Tape measure
- Head rod (1m stainless steel ruler or wooden ruler with bevelled edge)

Avoiding errors
- Ensure the base of the ruler is placed on the stream bed, and not on top of a large rock etc
- Be careful not to bury the end of the ruler, or shift its position, when it is rotated

Pollution clues and supporting tests

The amount of sediment a stream carries depends on flow rate. If it is safe measure water clarity and flow rate after a rainstorm. You may be able to construct a simple graduated marker set in the stream bed or bank that will show you what the flow rate is just by recording the water height off the gauge.
a. Measuring stream velocity using a float

1. Choose a section of stream that is fairly straight, free of snags and relatively fast flowing.
2. Use the tape to measure out 10m of the stream.
3. Drop the orange into the middle of the stream, a few metres upstream of the 'start' point (to allow the orange to build up speed).
4. As the orange crosses the start line begin timing.
5. Stop timing when the orange reaches the end of the measured length.
6. Repeat the measurement up to three times, particularly if the first two times are different. Write the results into the record sheets.
7. Calculate the average corrected velocity using the guidelines in the flow record sheet.

b. Measuring stream velocity with a head rod

1. Place a tape measure across the waterway and secure it on both sides.
2. Measure the depth of the stream with the thin edge of the ruler facing into the flow. Record.
3. Rotate the ruler 90° so that the flat side faces the flow, creating a standing wave or 'head'. Calculate the height of the head and record on the flow record sheet.
4. Repeat these measurements at 5 to 10 approximately equal intervals across the stream, recording the position of each depth/flow measurement on the tape measure. Finish measuring before you reach the edge, where the flow is minimal.

Effects of large flow variations

- Higher than normal flow velocities may disrupt communities of aquatic organisms, flush away algae and aquatic plants, and increase sediment levels
- Lower than normal flows can increase temperatures, lower oxygen levels, concentrate nutrients and increase algae and plant growth
Stream Flow Record Sheet - Float Method

Site/sample name: ____________________________ Date: ____________________________ (NB: all measurements in metres*)

**Step 1**
**Calculate the average depth**

Depths across stream: 
- \( d^* \) ______ m at _____ m
- \( d^* \) ______ m at _____ m
- \( d^* \) ______ m at _____ m
- \( d^* \) ______ m at _____ m
- \( d^* \) ______ m at _____ m
- \( d^* \) ______ m at _____ m
- \( d^* \) ______ m at _____ m
- \( d^* \) ______ m at _____ m
- \( d^* \) ______ m at _____ m
- \( d^{10} \) ______ m at _____ m

Distance across: 
- _____ m
- _____ m
- _____ m
- _____ m
- _____ m
- _____ m
- _____ m
- _____ m
- _____ m
- _____ m

Average depth = total depth
\[ \text{Average depth} = \frac{\text{total depth}}{\text{number of depths}} \]

**Average depth = _____ m**

**Step 2**
**Measure the stream width = ___m**

**Step 3**
**Calculate the area of the cross section**

Average velocity = \( \frac{\text{distance travelled (m) \times correction factor}}{\text{average time}} \)

Area = average depthxwidth = _____ m\(^2\)

**Average velocity = _____ ms\(^{-1}\)**

**Step 4**
**Calculate the average float time**

Measure the time taken for the float to travel your measured distance (e.g., 5m or 10m)

Float distance = ___ m

- \( t_1 \) ______ seconds
- \( t_2 \) ______ seconds
- \( t_3 \) ______ seconds
- \( t_4 \) ______ seconds
- \( t_5 \) ______ seconds

Total time = ______ seconds

**Average time (t) = _____ seconds**

**Step 5**
**Calculate the average velocity of the flow**

Average velocity = \( \frac{\text{distance travelled (m) \times correction factor}}{\text{average time}} \)

Average depth = total depth
\[ \text{Average depth} = \frac{\text{total depth}}{\text{number of depths}} \]

**Flow = velocity _____ ms\(^{-1}\) \times area _____ m\(^2\) = _____ m^3s^{-1}**
Step 1
Calculate the average depth

Depths across stream

<table>
<thead>
<tr>
<th>d1</th>
<th>d2</th>
<th>d3</th>
<th>d4</th>
<th>d5</th>
</tr>
</thead>
<tbody>
<tr>
<td>m</td>
<td>m</td>
<td>m</td>
<td>m</td>
<td>m</td>
</tr>
</tbody>
</table>

Distance across

<table>
<thead>
<tr>
<th>at 1</th>
<th>at 2</th>
<th>at 3</th>
<th>at 4</th>
<th>at 5</th>
</tr>
</thead>
<tbody>
<tr>
<td>m</td>
<td>m</td>
<td>m</td>
<td>m</td>
<td>m</td>
</tr>
</tbody>
</table>

Average depth = total depth
number of depths

Average depth = ____ m

Step 2
Measure the stream width = ___ m

Step 3
Calculate the area of the cross section

Area = average depth x width = ____ m²

Step 4
Calculate the velocity head

Height against flat edge

h1  ____ m  ____ m
h2  ____ m  ____ m
h3  ____ m  ____ m
h4  ____ m  ____ m
h5  ____ m  ____ m
h6  ____ m  ____ m
h7  ____ m  ____ m
h8  ____ m  ____ m
h9  ____ m  ____ m
h10 ____ m  ____ m

Head heights

Average head height = total head heights
total number of heights

Average head height (h) = ____ m

* Flat edge height (h) minus knife edge height (d)

Step 5
Calculate the velocity of the flow

Velocity (v) = \( \sqrt{2gh} \), where g = 9.81 m/s²
or \( v = 4.45h \)

Average velocity = ____ m/s

Step 6 Calculate the flow rate of the Stream

Flow = velocity _____ m/s x area _____ m² = _____ m³/s

Average depth

<table>
<thead>
<tr>
<th>1</th>
<th>2</th>
<th>3</th>
<th>4</th>
<th>52</th>
<th>53</th>
<th>54</th>
</tr>
</thead>
<tbody>
<tr>
<td>m</td>
<td>m</td>
<td>m</td>
<td>m</td>
<td>0.01</td>
<td>0.02</td>
<td>0.03</td>
</tr>
</tbody>
</table>

*Examples 1 cm = 0.01
Stream shape (cross-sectional area)

Equipment
- Tape measure
- Metre ruler
- Pen and field record sheet/notebook

Avoiding errors
- The more depth measurements you take the more accurate your cross sections will be. Ten readings are usually adequate.
- Measuring width - measure only the width of the flowing water. Still water, at the very edges, does not 'count'.

Pollution clues and supporting tests
Measuring the cross section at the same point each time will enable you to detect changes in the shape of the stream channel caused by erosion or deposition.
- Other indicators of change include percentage of different substrate sizes and substrate embeddedness.
- Some changes may be easily observed, such as bank erosion and the formation of island bars.
- Obstructions to stream flow may create changes to the streambed and banks. Observe to see if the changes are serious enough to require the removal of the obstruction. Seek advice if it is large or deeply buried.

Effects of changing channel shape
- Changes in channel shape always has downstream consequences such as bank erosion, sediment deposition, alteration to channel shape and position, and habitat scouring or smothering.
1. Stretch the tape or string line across the width of the stream, at right angles to the stream bank.

2. Measure the maximum depth (*d1) plus at least four other depth measurements (d2 - d5), with the measurements spaced at approximately equal intervals across the channel.

3. Repeat this at a second point on the stream and record these results on the table on the record sheet.

4. Calculate the cross sectional area using the guidelines in the 'Stream Flow' record sheet.

5. From your recorded measurements draw the stream cross sections.
Macroinvertebrate sampling

Macroinvertebrates are animals that do not have backbones, but are mostly visible to the naked eye. *In this section, and in book 6, we have used the terms macroinvertebrates and 'bugs' interchangeably.*

The macroinvertebrate sampling methods described in this section use a kick net to gather samples from the range of habitats available at your site. Generally macroinvertebrates attach themselves to stable substrate which occurs in the flow conditions that suit them. Directions are given below for selecting the best sampling sites for the range of habitats you are likely to find at your sites.

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 required. 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.

It is particularly important that your survey procedures are consistent for these assessments, from sample collection to sample sorting and identification. Collecting your samples from a measured area of stream habitat is fundamental to this process; either the kicked area for riffles or the number of 'jabs' of the net for other habitat. Use the same techniques for collecting samples throughout the reach if possible.
Collecting your sample

Where to sample

Your sampling site is likely to have four or five habitats present, in various proportions, as shown below. It is best to concentrate sampling efforts on the most productive habitat available, yet sample others if they are present. This will improve your chances of collecting the greatest variety of organisms possible. Favourite bug colonisation places (habitats) are presented in order below:

1. **Riffle areas.** Shallow parts of the stream where water flows swiftly over a rough bed (such as cobbles) are generally well oxygenated and, therefore, prime habitats for bugs. This habitat is relatively uncommon in our low gradient streams.

2. **Vegetated bank margins.** Consist of overhanging bank vegetation and submerged root mats attached to banks, providing stable attachment sites for bugs to colonise. This is generally a highly productive habitat in a muddy-bottom stream.

3. **Snags and logs.** Consists of submerged wood, primarily dead trees, logs, branches, roots and leaf packs lodged between rocks or logs. This is also a very productive muddy-bottom stream habitat.

4. **Aquatic vegetation beds and decaying organic matter.** Consists of beds of submerged, green/leafy plants that are attached to the stream bottom. This habitat can be as productive as vegetated bank margins, and snags and logs.

5. **Silt/sand/gravel substrate.** This is the least productive of the stream habitats and in most cases should be avoided. Sorting through a muddy sample is time consuming and usually quite unrewarding.

Habitats found in low gradient streams. Most macroinvertebrates are usually found in riffle, vegetated habitats and snags and logs.
Planning your sampling effort

The objective is to collect a combined sample from the variety of habitats at your site, concentrating on the most productive available.

Not all habitats are present in all streams or present in significant amounts so the first task is to decide how you will divide your sampling effort:

- **If all four habitats are present** in plentiful amounts, aim to collect half your sample from the riffle area, quarter from the vegetated banks, and the rest from the other two habitats - snags/logs and aquatic vegetation. Avoid sampling from silt/s and gravel substrate.

- **If three habitats are present** in plentiful amounts and riffle is absent, jab the vegetated banks 10 times and divide the remaining 10 jabs among the remaining 2 habitats.

- **If only two habitats are present** in plentiful amounts, jab the more productive habitat 15 times and the other 5 times.

- **If some habitats are plentiful and others are sparse**, sample the sparse habitats to the extent possible, even if you can take only one or two jabs. Take the remaining jabs from the plentiful habitat(s). This rule also applies if you cannot reach a habitat because of unsafe stream conditions. Jab a total of 20 times.

Note on the field record sheet how many jabs you took in each habitat. This information will be needed next time, to ensure you collect samples in exactly the same way each time at your site(s).

Also note the proportion of each type of habitat in your stream reach.

Equipment

- Kick net (with a mesh size of 0.8mm)
- Bucket (2 if one is needed to sieve debris and mud)
- Gloves (and/or brush for removing bugs from substrate)
- Spray water bottle
- Sorting tray
- Suitable footwear

Where to start

Approach each sample point from downstream, and sample as you walk upstream.

How to collect your sample

a. **To sample a riffle area**

1. Select a 0.5 x 0.5m riffle area for your sample.

2. Place the net in the water at the downstream edge of the sample area, making sure the bottom of the net fits tightly against the stream-bed.
3. Pick up any large rocks in the sampling area and brush or rub the sides and lower surface directly in front of the net.

4. Check that all clinging macroinvertebrates have been dislodged then place each cleaned rock outside of the sampling area. (After sampling is completed, rocks can be returned to the place they came from).

5. Use your foot to dislodge the upper layer of cobble or gravel, starting at the edge nearest the net. Be sure to disturb the first few centimetres of stream sediment to dislodge burrowing organisms.

6. As a guide, disturb the sampling area for about 1 minute, or until the area is thoroughly worked over.

b. To sample vegetated bank margins
jab the net vigorously against vegetation and roots along the bank. Move the net against the current with an upward motion to make sure you net most of the bugs you dislodge. The entire jab motion should occur underwater.

c. To sample snags and logs, hold the net with one hand under the section of submerged wood you are sampling. With the other hand (which should be gloved), rub about 25 cm x 25 cm of area on the snag or log. Scoop organisms, bark, twigs, or other organic matter you dislodge into your net. Each combination of log rubbing and net scooping is one jab.

Branches and sticks can be held directly over the bucket and bugs removed by pouring water over the bits of wood while gently brushing the surface.

d. To sample aquatic vegetation beds, jab vigorously, with an upward motion, against or through the plant bed. The entire jab motion should occur underwater.
Identifying and sorting bugs using the bug box

Equipment

- Sorting tray
- Bug handling tools (bug suckers and brushes)
- Bug box
- Hand lens and key/s to identify bugs

How to collect your sample

1. Pour some or all of the contents of the bucket into the sorting tray as a thin layer, so the bottom is still visible. (a method for 'cleaning-up' the sample if it is full of debris is included in the Appendix.

2. Part-fill the bug box with clean stream water.

3. Use bug suckers or brushes to pick through the leaf litter and organic material looking for anything that swims, crawls, or is hiding in the debris. Look carefully; many of these creatures are quite small and may not move until touched.

4. Use the pictures on the bug box (or suitable key) to identify your bugs.
5. Sort organisms into the bug box compartments, using the lid pictures to help you to place these in the right place.

Note: Size and colouration alone are not valid criteria for distinguishing different types, or taxa. While some species are smaller than others, individuals within a species may vary in size depending on age.

6. Be thorough as you search and try your best to find as many different groups as you can. Keep going until you have sorted at least 100 bugs.

7. Check that there are no strays, all bugs should be in their proper place.

Have someone check your sorting before you tally your sample.

8. Use the biological assessment sheet to record the numbers of bugs you have identified.

9. State where you collected your sample on the record sheet, or show the position on the stream reach map.

10. Calculate biological indices that suit the goals of your programme.

Avoiding errors

- Some bugs are very small and it is easy to miss some, or they are placed in the wrong cells in the tray. Have someone check your sorting.

- To be confident of making valid comparisons between sites it is essential that you sample the same combination of habitat types at each. For example, if you have sampled a riffle area and bank vegetation at site 1, then for valid comparison you must also sample the same combination at other sites.

The chemical and physical tests described above can be used to define the life support capacity of a stream.

If habitat quality is not a limiting factor then chemical/physical factors may be responsible for reducing the size of the population, eliminating sensitive species and/or reducing diversity.

What to do with your data

Macroinvertebrates are sorted in a bug box and grouped into three colour coded categories based on their ability to tolerate pollution.

The ratings are:
- green for low tolerance;
- yellow for moderate tolerance;
- red for high tolerance;

More complicated water quality indices can then be calculated from the bug box tallies (see the macroinvertebrate record sheet) using the formula provided in the following section on analysing macroinvertebrate results.
### Macroinvertebrate record sheet

**Site/sample name** | **Date**
--- | ---

**Sorting:**

Sort the bugs into the positions shown on the lid of the bug box.

Use the booklet “Auckland Stream Invertebrates - examples of the good, the bad and the ugly” to help you identify the bugs.

<table>
<thead>
<tr>
<th>Group</th>
<th>Count</th>
</tr>
</thead>
<tbody>
<tr>
<td>Mayflies (Ephemeroptera)</td>
<td></td>
</tr>
<tr>
<td>Stoneflies (Plecoptera)</td>
<td></td>
</tr>
<tr>
<td>Caddisflies (Trichoptera)</td>
<td></td>
</tr>
<tr>
<td>Beetles (Coleoptera)</td>
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</tr>
<tr>
<td>Dobsonflies (Megaloptera)</td>
<td></td>
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<tr>
<td>Damselflies (Odonata)</td>
<td></td>
</tr>
<tr>
<td>Boatman / Backswimmers (Hemiptera)</td>
<td></td>
</tr>
<tr>
<td>Shrimp (Crustacea)</td>
<td></td>
</tr>
<tr>
<td>Other Crustacea (Isopods/Copepods)</td>
<td></td>
</tr>
<tr>
<td>Mussels (Bivalvia)</td>
<td></td>
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<tr>
<td>Snails (Gastropoda)</td>
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<tr>
<td>Leeches (Hirudinea)</td>
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<tr>
<td>Flatworms (Tricladiida)</td>
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<tr>
<td>Worms (Oligochaeta)</td>
<td></td>
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<tr>
<td>Trueflies/Bloodworms (Diptera)</td>
<td></td>
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</tbody>
</table>
There are a number of indices that can be used to assess the health of a particular stream. They are relatively simple to calculate and each one gives a slightly different perspective on the ‘health’ of your stream. By calculating them all you can gain a more comprehensive understanding of your stream and what might be affecting it.

**Tolerance - measures of sensitivity to impacts**

Macroinvertebrates can be grouped according to their tolerance for pollution. At the simplest level a stream quality assessment can be made by looking at the way the bugs are distributed in the bug box.

If most of the sample occupies the ‘sensitive’ (green) part of the box then it can be assumed that stream condition - a product of water and habitat quality - is reasonable. A balanced stream community would have representatives spread throughout the box with the greatest number in the medium sensitivity portion. A stream with compromised physical or chemical characteristics will have most bugs in the red compartments or worse still none at all.

Bug sensitivity to pollution varies considerably within orders, so this method provides only a very coarse level of pollution detection.

**Taxa richness - a measure of diversity**

Count the number of taxa represented in the whole sample and record this value on the data sheet. Richness numbers usually increase with increasing sample size. 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 taxa, tells you important information about the diversity 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 better information. The greater the taxa richness, the higher the diversity of your bug population.

In general, streams with a higher diversity of bugs are considered healthier than those with a lower diversity. However, a moderate amount of organic pollution (discharges from the sewer system or contaminated stormwater) sometimes causes an increase in taxa richness, especially in streams that are naturally low in taxa and number of individuals (such as muddy bottomed streams).

1. Taxonomy can be to any level (order, family or genus/species), but should be consistent among samples. Wai Care indicies require samples to be sorted to the order level of classification.
### Composition - measures of dominance

#### A. Percentage dominance

Percentage dominance is calculated using the following formula:

\[
D = \frac{\text{Number of the most abundant taxa present in a sample}}{\text{Total number of individuals collected in a sample}} \times 100
\]

Percentage dominance is used to provide an estimate of the diversity patterns in the community. Generally, a community that is dominated by a single group of bugs, or taxon, is indicative of sites with large disturbances (e.g., regular floods, pollution or high water temperatures), while low percentage dominance indicates low disturbance and better quality.

#### B. Percent EPT

Add together the number of mayflies, stoneflies and caddisflies in the sample (i.e. total EPT) and calculate percent EPT by the following formula.

This index is based on the percentage of pollution 'sensitive' types represented 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.

\[
\text{Percent EPT} = \frac{\text{Number of EPT in sample}}{\text{Total number of bugs in sample}} \times 100
\]
Habitat assessments

The following descriptions will assist you to investigate the opportunities for different types of stream life to colonise and live successfully in your stream. It will help you calculate a rating for the stream habitat for your study area and indicate what you can expect to live there. Consider both sides of the stream over the entire length of the study area when making your assessment. Score stream habitat from 8 (excellent) to 0 (poor), and the stream bank features from 4 to 1 as shown below.

1 Instream cover and surfaces to colonise.

*Does the stream provide ‘cover’ and places to colonise for stream life?*

Fish and other aquatic organisms require stable natural structures such as snags, logs, vegetation, rock or cobble areas where they can shelter from predators and swift currents, find food and reproduce.

Habitat quality is greater if there is a range of stable substrates for different types of stream life (e.g. fine woody debris, submerged logs, leaf packs and rocks).

Large aquatic plants (macrophytes) and undercut banks may also be very important to the fish in your stream, particularly if other forms of cover or refuge are not abundant.

A lower variety and abundance of cover decreases the potential for recovery following disturbance.

<table>
<thead>
<tr>
<th>Excellent</th>
<th>Good</th>
<th>Fair</th>
<th>Poor</th>
</tr>
</thead>
<tbody>
<tr>
<td>&gt;50% of streambed a good stable mix of habitat types. Many plants overhanging and some instream</td>
<td>30-50% of streambed a good stable mix of habitat types. Some plants overhanging and/or instream</td>
<td>10-30% of streambed a good stable mix of habitat types. Signs of frequent bed disturbance. Few plants overhanging stream</td>
<td>&lt;10% stable habitat. Bed unstable or absent. Few overhanging plants</td>
</tr>
<tr>
<td>8 7</td>
<td>6 5</td>
<td>4 3</td>
<td>2 1 0</td>
</tr>
</tbody>
</table>
2. Pool variability

Are there different types of pools present in the stream?

- A stream with many pool types (and bends) will support a wide variety of aquatic species.
- The 4 basic types of pools are large-shallow, large-deep, small-deep, and small-shallow. If a pool is more than half the width of the stream in length or width it is considered to be large, and deep if it is twice as deep as most of the rest of the stream. A shallow pool is less than 1 times deeper than the most of the rest of the stream.
- Pools are important resting and feeding sites for fish.

<table>
<thead>
<tr>
<th>Excellent</th>
<th>Good</th>
<th>Fair</th>
<th>Poor</th>
</tr>
</thead>
<tbody>
<tr>
<td>Even mix of large-shallow, large-deep, small-deep and small-shallow pools present</td>
<td>Majority of pools large-deep, very few shallow</td>
<td>More shallow pools than deep</td>
<td>Most pools small-shallow or pools absent</td>
</tr>
<tr>
<td>8 7</td>
<td>6 5</td>
<td>4 3</td>
<td>2 1 0</td>
</tr>
</tbody>
</table>

3. Sediment deposition

Is sediment being deposited in the stream?

- Estimate the amount of sediment that has accumulated in pools and the changes that have occurred to the stream bottom as a result of deposition.
- Sediment deposition may result in the filling of runs and pools, or cause the formation of islands or bars.
- Usually deposition is evident in areas that are obstructed by natural or manmade debris and areas where the stream flow decreases, such as bends.
- High levels of sediment deposition are symptoms of an unstable and continually changing environment that becomes unsuitable for many organisms.
4. Bank erosion and stability

How stable are both left and right banks (looking upstream) of the stream?

Stream banks naturally erode, particularly on bends. However, changes in adjacent land areas can cause stream to become unstable, resulting in continuous erosion along its channel. Bank erosion introduces sediment into the stream, smothering instream habitat.

Changes include increased run-off from hard (imperious) surfaces and piped tributaries, or direct interference such as straightening or channelling the stream.

Steep banks are generally more likely to collapse and suffer from erosion than are gently sloping banks.

<table>
<thead>
<tr>
<th>Excellent</th>
<th>Good</th>
<th>Fair</th>
<th>Poor</th>
</tr>
</thead>
<tbody>
<tr>
<td>Stream bank is stable with little evidence of erosion. No evidence of human changes</td>
<td>Stream bank is stable with some evidence of old but stabilised erosion. No evidence of human changes</td>
<td>Stream bank is unstable and obvious examples of erosion. Human changes to stream have used natural materials</td>
<td>Stream bank is very unstable and actively eroding, or human changes have used concrete, wood or rock baskets</td>
</tr>
<tr>
<td>Left Bank</td>
<td>4</td>
<td>3</td>
<td>2</td>
</tr>
<tr>
<td>Right Bank</td>
<td>4</td>
<td>3</td>
<td>2</td>
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</table>
5. Bank protection

Are plants protecting the stream banks?

- The soil on banks is held in place by plant roots.
- Deep root systems offer more bank protection than shallow root systems.
- The more diverse the plant community on the banks the better. Young plants, which grow and reproduce rapidly, are better than old plants.
- The depth of plant root systems becomes more important as height and slope of the stream bank increases.

<table>
<thead>
<tr>
<th>Excellent</th>
<th>Good</th>
<th>Fair</th>
<th>Poor</th>
</tr>
</thead>
<tbody>
<tr>
<td>Banks and stream margin has good coverage with a range of larger native plant types with little evidence of human interference</td>
<td>More than half the bank covered with different types of native plants. Growth affected by some human activity (e.g. mowing, herbicide)</td>
<td>Half to a quarter of bank covered with different types of plants (including exotics); marked human activities</td>
<td>Less than one quarter of bank covered with larger plants. Plant growth severely affected by human activities</td>
</tr>
<tr>
<td>Left Bank</td>
<td>4</td>
<td>3</td>
<td>2</td>
</tr>
<tr>
<td>Right Bank</td>
<td>4</td>
<td>3</td>
<td>2</td>
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6. Riparian zone

Is the stream vulnerable to runoff from human disturbance of the stream bank?

- The effectiveness of the riparian zone is diminished as vegetation is removed, and the width reduced.
- Healthy vegetation from the edge of the stream bank out through the riparian zone can serve as a buffer to pollutants entering the stream from runoff, control erosion, provide habitat and contribute nutrients (in the form of plant matter such as leaves and twigs) to the stream.
- Grasses and small shrubs next to the stream will provide escape cover or refuge for fish.
- Urban streams often have little or no riparian vegetation. Residential developments, parking areas, shopping centres, roads and access ways are often squeezed in near streams creating narrow riparian zones.
Habitat assessment

Equipment

- Habitat assessment record sheets
- Guide to habitat assessment scoring (if required)

Measurement

1. Use the descriptions on the habitat record sheet to help you search for the key scoring features for each habitat factor.

2. Work your way across the rankings (excellent through to poor) until you find the one that best describes the features of your site.

3. Make sure the features you select for your score represent the entire sampling reach. A few small exceptions to the overall condition are usually not included, but may be noted at the bottom of the record sheet.

4. Each bank is evaluated separately and the cumulative score (right and left) is used for this parameter.

Avoiding errors

- Habitat assessments are often better done in pairs - different people place different emphases on the different features.

- After scoring refer to previous scores for the same site.

- Take a photo of the site and ask for independent evaluation.

The variety and number of living things in your stream is limited by the quality of the habitat. Both stream and riparian habitat influence the structure and function of stream life, setting the basic template within which biological communities develop.
<table>
<thead>
<tr>
<th>Habitat factors</th>
<th>Date</th>
<th>Date</th>
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</thead>
<tbody>
<tr>
<td>1. Instream cover and surfaces to colonise.</td>
<td>Score</td>
<td></td>
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<tr>
<td>2. Pool variability</td>
<td>Score</td>
<td></td>
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<tr>
<td>3. Sediment deposition</td>
<td>Score</td>
<td></td>
</tr>
<tr>
<td>4. Bank erosion and stability</td>
<td>Left Bank Score</td>
<td></td>
</tr>
<tr>
<td>Bank erosion and stability</td>
<td>Right Bank Score</td>
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<tr>
<td>5. Bank protection</td>
<td>Left Bank Score</td>
<td></td>
</tr>
<tr>
<td>Bank protection</td>
<td>Right Bank Score</td>
<td></td>
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<tr>
<td>6. Riparian zone width</td>
<td>Left Bank Score</td>
<td></td>
</tr>
<tr>
<td>Riparian zone width</td>
<td>Right Bank Score</td>
<td></td>
</tr>
<tr>
<td>Total scores</td>
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Has instream habitat been otherwise greatly altered? How?
<table>
<thead>
<tr>
<th>Date</th>
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Water clarity vs turbidity

Water clarity is related to turbidity but the values you measure with the turbidity tube are not exactly the same. This graph can be used to approximate turbidity values from water clarity tube readings.

Compare your results to the water quality decision support table in book 4 to determine if a water quality problem exists.
Percentage saturation of oxygen

Note: A dissolved oxygen test tells us precisely how much oxygen is dissolved in water, but it does not indicate how much DO the water is capable of holding at the test temperature of the water. The percentage saturation is a better measure of the availability of oxygen to aquatic organisms.

Calculation of results

1. Plot temperature on upper scale.

2. Plot oxygen concentrate on lower scale.

3. Hold a ruler between the two points.

4. The point where the ruler crosses the middle scale is the % saturation.

5. Record this result on your test results worksheet.

Example BOD calculation:

DO level on day 1 = 8 mg/L
DO level on day 5 = 5 mg/L
BOD level = DO on day 1 - DO on day 5
= 8 mg/L - 5 mg/L
= 3 mg/L
Cleaning the mud and debris from your sample

If your sample is full of debris and mud follow this simple procedure:

1. Remove large pieces of debris (leaves, twigs, and rocks) from the sample. Carefully remove the debris one piece at a time. While holding the material over the bucket, use the forceps, spray bottle, and your hands to pick, rub, and rinse the leaves, twigs, and rocks to remove any attached organisms. Use your magnifying lens and brush to find and remove small organisms clinging to the debris. When you are satisfied that the material is clean, discard it back into the stream.

2. Place a large piece of sieve cloth over the top of your second bucket, and push down in the centre to create a slight depression. The sieve cloth can be secured at the top to prevent it collapsing.

3. Swirl the remaining contents of the bucket to lift the bugs and debris off the bottom then carefully pour through the sieve. The sieve will catch the bugs and some of the fine debris.

4. If the sieve still contains lots of mud pour some clean water over the sample. This should wash the fine particles of mud through the sieve, leaving a reasonably clean sample behind.

5. Remove the sieve and wash the bugs into the sorting tray using your spray bottle. Be sure to remove all the clinging bugs from the sieve.
Design and illustrations: Wazoo Design
Cover photo: Rob L. Suisted, Nature’s Pic Images

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