Waterwatch Field Manual

A complete manual for on-site monitoring of water quality and waterway health

Waterwatch New South Wales
Communities Caring for Catchments

NSW Government
Environment, Climate Change & Water

Australian Government

Acknowledgements

This *Waterwatch Field Manual* and the accompanying *Senior Waterwatch Teachers' Guide* and *Community/Land Manager Waterwatch Guide* have been developed by Waterwatch coordinators in collaboration with Waterwatch partners. Many thanks to the following people for contributing their resources and expertise:

- Ingrid Berthold, Hunter–Central Rivers CMA Waterwatch Coordinator
- Bruce Chessman, Department of Environment, Climate Change and Water, Principal Research Scientist
- Robert Clegg, Australian Government Indigenous Land Management Facilitator, 2008
- Amanda Gregory, Department of Education and Training and Hunter–Central Rivers CMA Waterwatch Coordinator
- Colin Mondy for sharing his extensive expertise as Waterwatch Coordinator (retired)
- Stuart Naylor, Sydney Catchment Authority, Riparian Program Coordinator.
- Beryl Newman, Department of Environment, Climate Change and Water, Waterwatch Coordinator
- Carla Sbrocchi, Northern Rivers CMA Waterwatch Officer
- Jane Smith, Community Environment Network
- Samantha Willis, Central Coast Waterwatch Coordinator

NSW Waterwatch gratefully acknowledges the permission granted to use materials from a variety of sources including:

- Streamwatch, Sydney Water and Sydney Catchment Authority for information from the *Streamwatch Manuals*
- Waterwatch Australia Steering Committee for material from the *Waterwatch Australia national technical manual* modules
- Michael Cassidy for information from the *Waterwatch Tasmania reference manual: a guide for community water quality monitoring groups in Tasmania*
- NSW Department of Primary Industries for a variety of information including that contained in the publication *Physical Property Planning*
- Land and Water Australia for water quality, riparian assessments and illustrations
- Department of Natural Resources (WA), *Ribbons of Blue: in and out of the classroom*
- Department of Natural Resources and Water, Queensland, for their *Community waterway monitoring manual*
- Geoffrey Simpson (Aboriginal Project Officer, Murrumbidgee CMA) for his cultural landscape assessment.

NSW Waterwatch acknowledges the support provided in the development of this valuable resource by NSW Catchment Management Authorities, the Waterwatch network across New South Wales and Australia, and the community involved in Waterwatch.

Published by: Department of Environment, Climate Change and Water NSW
59–61 Goulburn Street, PO Box A290, Sydney South 1232
Ph: (02) 9995 5000 (switchboard)
Ph: 131 555 (environment information and publications requests)
Ph: 1300 361 967 (national parks, climate change and energy efficiency information and publications requests)
Fax: (02) 9995 5999 TTY: (02) 9211 4723
Email: info@environment.nsw.gov.au Website: www.environment.nsw.gov.au
ISBN 978 1 74232 372 5 DECCW 2009/497

How to use this manual

This *Waterwatch Field Manual* and the accompanying *Senior Waterwatch Teachers' Guide* and *Community/Land Manager Waterwatch Guide* have been designed to provide a complete guide to designing and implementing the Waterwatch program in New South Wales, within secondary schools or in the community, respectively.

The methods and procedures described combine best practice and scientific rigour with straight-forward instructions, to ensure participants gain maximum benefit while also contributing high quality data to the Waterwatch database. Such data becomes a valuable tool for natural resource managers to use in catchment planning.

This field manual provides background information and detailed instructions, to make visits to your chosen site a valuable learning experience resulting in high quality data. This resource is intended for use on-site and will guide students or other community members as they develop essential scientific skills and gain a first-hand insight into their local catchment.

The manual is divided into numbered sections:

Section 1: Assessing your site
Section 2: Waterwatch equipment
Section 3: Testing water quality in the field
Section 4: Procedure sheets
Section 5: Material safety data sheets
Section 6: Interpreting your results
Section 7: Habitat: the aquatic zone
Section 8: Habitat: the riparian zone
Section 9: Habitat: riparian condition assessment
Section 10: Water bug (macroinvertebrate) survey
Section 11: Human impacts on waterways

This field manual is to be used in conjunction with either the *Senior Waterwatch Teachers' Guide* or the *Community/Land Manager Waterwatch Guide*.

Waterwatch offers a way for students and other interested community members to get involved in monitoring the health of their environment and to take part in managing some of the problems.

Congratulations on your involvement in Waterwatch!

Disclaimer

The Department of Environment, Climate Change and Water advises that those who participate in Waterwatch do so at their own risk. No responsibility or liability is accepted for any injury, loss or damage, however caused, arising from any participant's involvement in the organisation, conduct or participation in Waterwatch.

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SECTION 1

Assessing your site

Site assessment is the preliminary work done to obtain an overview of your proposed site prior to beginning any water quality testing.

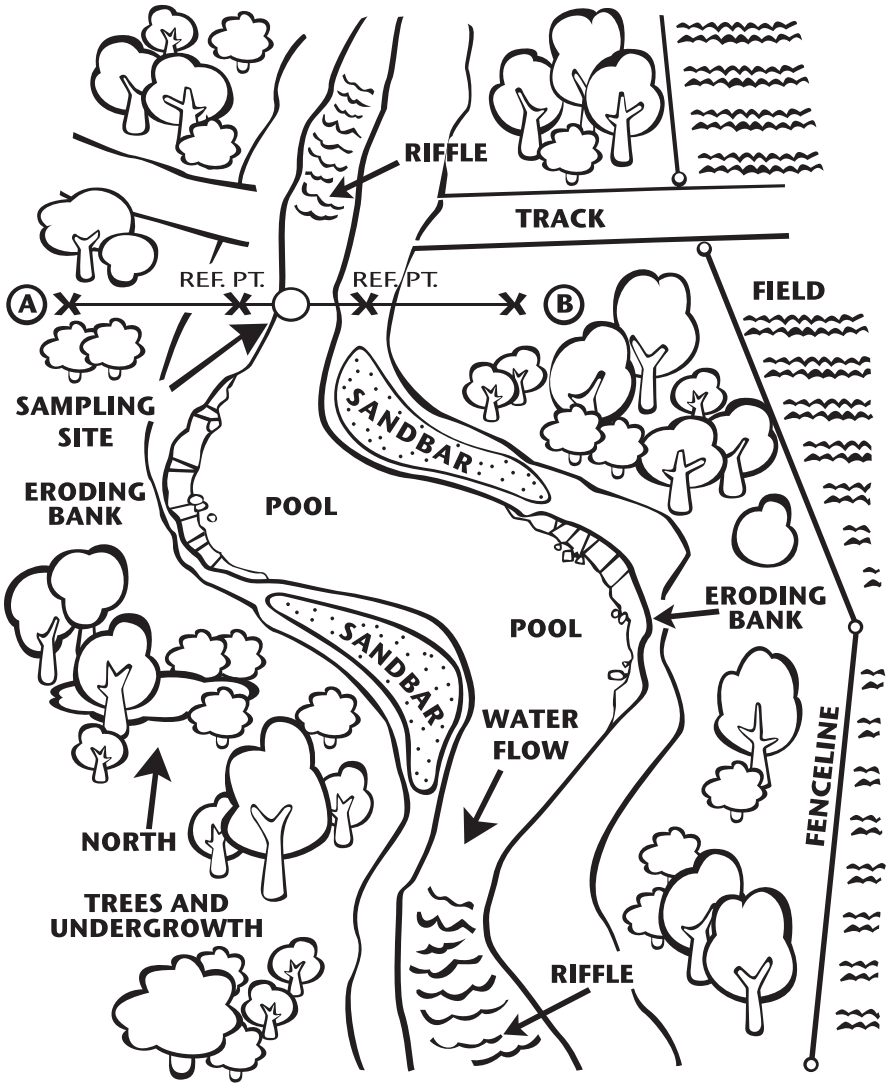
This section of the field manual contains a series of work sheets, checklists and some background information to assist participants to do a detailed assessment of the site they have chosen for investigation.

1.1 Draw a bird's eye view map of your site

A bird's eye view map provides an aerial view of your site. This type of map provides a clear visual description of the features of your site. You can use online map sites to help with aerial views or satellite imagery.

Draw the shape of a section of your waterway (at least 100 metres) and mark on your testing site. Draw and label the features such as large logs and branches within the stream, overhanging vegetation, aquatic vegetation and any other key features at the site.

Mark on the location where photographs were taken and water quality tests conducted (see Section 1.2). Your map may look something like the sketch below.



1.2 Photopoints of your site

Photos provide a visual record of your site over time and provide evidence of change. Find a location at the site where you can take photographs to compare changes over time. You must be able to find the same position to take photos at this site twice per year. It needs to be a recognisable feature that will provide a reference point for the centre of the photo.

At your site, make sure you photograph:

- ☐ upstream and downstream in different seasons, to record natural and human changes over time
- ☐ close ups, to show more detail about special features at your site.

Some possible photos:

- ☐ upstream/downstream
- ☐ distant view
- ☐ left bank/right bank
- ☐ human changes/natural changes
- ☐ potential pollution sources.

Record relevant details for each photo, including time, date and location.

Storing photographs

- ☐ As you take photos place them alongside their matching photos taken at different times and add the photo date to the file names.
- ☐ If taking digital photos, use the highest resolution possible. This will allow you to use your photographs for reports or for promotion of your activities.
- ☐ Record where your photographs are stored.

Site name: ..... Date: .....

Description of your photopoint:

.....

.....

.....

Record the file reference for each photo:

Date	Location	Photo description	Photo file reference

1.3 Line drawing work sheet

A line drawing is a sketch (or picture) of the key features at your site drawn in the field or from a photo. A line drawing is done in pencil and requires the following steps:

1. Draw a frame approximately ½ page in landscape or portrait depending on the area to be drawn.
2. Mark on the skyline.
3. Draw in the main features.
4. Draw in the detail in the foreground.
5. Add the background detail.
6. Label the main features.
7. Write a title (top of drawing) or caption (at the bottom of the drawing).
8. Record the date.

Line drawing at my site

Site name: ..... Date: .....

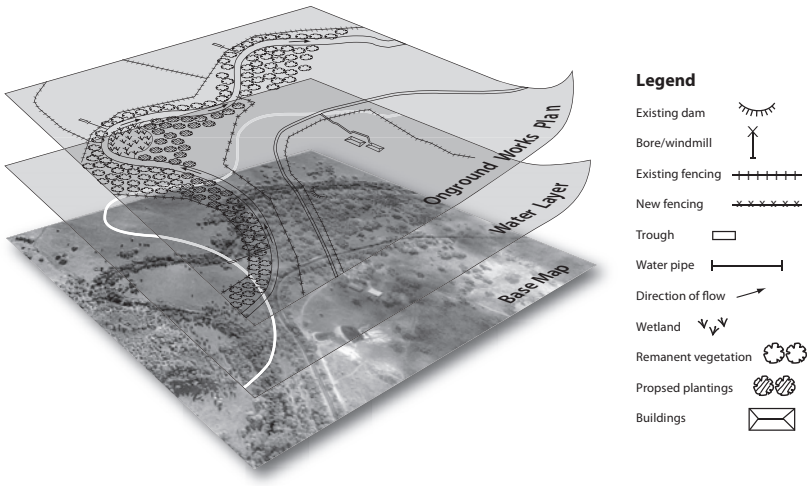
1.4 Aerial photographs and topographic maps

Aerial photographs and satellite images

Aerial photographs provide detailed information about landscape features across a small area. An aerial photograph is useful for:

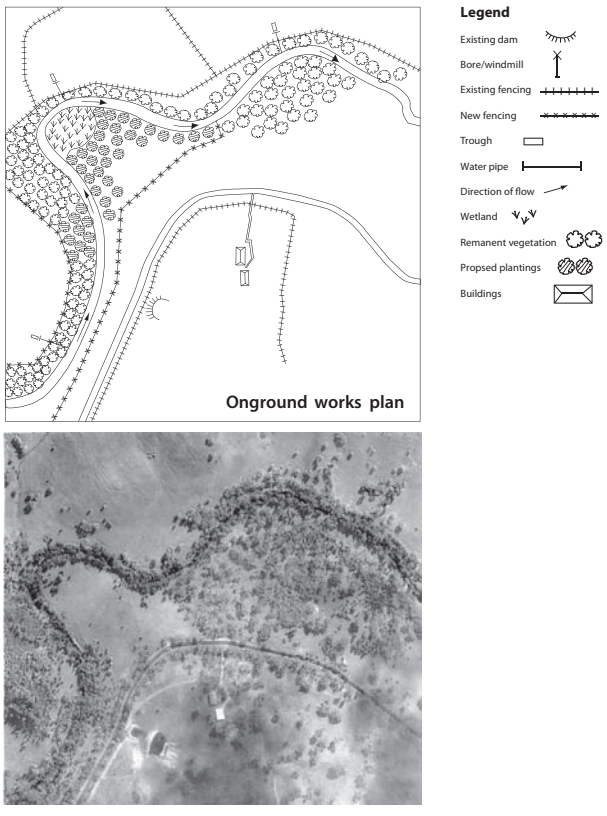
1. mapping catchment boundaries and the key sites for on-ground works
2. property planning where the features of a farm are identified as layers above the aerial photograph which is the base map.

Note: Some aerial photography can be downloaded from *Google Earth*.



Topographic maps

Topographic maps provide information about the shape of the landscape using contour lines and other natural landscape features such as vegetation and waterways. They also show infrastructure such as roads and buildings. These maps assist to locate sites by grid references. They provide detailed sub-catchment information that may be useful in designing a monitoring plan.



1.5 Locating your waterway: NSW catchments work sheet

In New South Wales, Catchment Management Authorities (CMAs) assist in the management of natural resources within catchments. The CMA regions are defined by the major catchment areas of NSW.

1. Locate and name your CMA region on the map of NSW.
2. On the large map of NSW locate and name your town.
3. What are the main rivers in your area?
4. Use a catchment map to help you to draw in your own catchment on the larger map.

Note: Your CMA may be able to provide a catchment map.

1.6 Mapping your local catchment work sheet

Mapping your local catchment is important. It provides a picture of the shape and size of your catchment, where your rivers go and what people do in the catchment.

How to develop a catchment map

It is best to use a topographic map but a road map or street directory may provide the basis for your local catchment map.

Before developing your catchment map consider the following:

Catchment and topography

- Where does the stream begin? What is its length? Where does it flow and where does it finish?
- Where are the catchment and sub-catchment boundaries?
- What are the local land forms, e.g. location of plateaus, escarpments, flood plains?
- How does the character of the river change along the catchment?

Land uses and values

- Where are the population centres in the catchment?
- How is the land used in the catchment?
- What are the current uses of the stream, e.g. fishing, swimming, drinking water supply, stock drinking, irrigation, aquaculture, industry, scenic value, protection of aquatic ecosystems?
- Have there been historical land-use changes in the catchment?
- What biological values are known, e.g. rare or threatened species and plant communities?
- What cultural heritage values are known, especially Aboriginal values?
- Past land use practices:
  - Was riparian vegetation cleared?
  - Were streams 'cleaned out', e.g. removal of coarse woody debris?
  - Were swamps in the catchment drained?
  - Has ploughing and/or road building triggered erosion?

On the map:

- Name the main rivers and creeks in your local catchment.
- Mark on the catchment boundary. This will be determined by the ridge line (highest point), above the smallest streams that drain into your river or creek.
- Complete the land-use survey on the following page and then locate the major land uses on your map.
- Develop a key for land use in the catchment.
- Mark on significant features that may impact on the quantity or quality of water in the catchment (e.g. dams, sewage treatment plant, etc.).
- Add the map essentials – a scale, direction (north), title or caption.

Take a waterway walk

Enlarge the section of your catchment map to include your testing site and approximately 1 kilometre upstream and downstream of the site.

Take a walk near your creek/river/estuary to discover the unmarked features that may be influencing water quality in this area. For example, stormwater drains, disturbed areas, unusual water colour, smells, etc.

Mark these on your enlarged map.

The information you have discovered will assist you to understand the issues that may affect water quality at your site.

Bird's eye view map

Refer to the beginning of this section to draw a bird's eye view map of your site.
Include:

- natural features at the site
- roads and fences
- buildings
- the direction of stream flow, indicated with an arrow.

Include the map essentials – direction, scale, frame and title.

1.7 Catchment survey work sheet

Make a photocopy of this sheet for each sub-catchment to be surveyed. At the end of your survey, collate information from each sub-catchment to get an overview of the whole catchment.

Note: These survey sheets work best when used for small catchments or sub-catchments.

Background information

Date: ..... Time: .....

Name of group: .....

Sub-catchment: .....

Nearest town or suburb: .....

Specific land uses identified

(tick as many as apply)

	Streamside	Within 1 km of stream	Within catchment
Agriculture			
Cattle crossing	☐	☐	☐
Streamside fencing	☐	☐	☐
Cropping	☐	☐	☐
Grazing	☐	☐	☐
Feed lot	☐	☐	☐
Dairying	☐	☐	☐
Orchard	☐	☐	☐
Other agricultural land	☐	☐	☐
Forestry	☐	☐	☐
Other (name)	☐	☐	☐
Recreation			
Swimming/fishing	☐	☐	☐
Power boating	☐	☐	☐
Picnic area	☐	☐	☐
Camp-ground	☐	☐	☐
Golfing	☐	☐	☐
Other (name)	☐	☐	☐

Catchment survey work sheet continued

Specific land uses identified

(tick as many as apply)

	Streamside	Within 1 km of stream	Within catchment
Other land uses			
Abandoned mine	☐	☐	☐
Landfill site	☐	☐	☐
Mines/quarry/gravel pits	☐	☐	☐
Aboriginal cultural heritage site	☐	☐	☐
Other (name)	☐	☐	☐
Built environment			
Urban residential	☐	☐	☐
Rural residential	☐	☐	☐
Industry (factories)	☐	☐	☐
Commercial, e.g. shops	☐	☐	☐
Schools	☐	☐	☐
Park/gardens	☐	☐	☐
Roads or bridges	☐	☐	☐
Sewage treatment plant	☐	☐	☐
Water treatment plant	☐	☐	☐
Petrol stations/car repair workshops	☐	☐	☐
Construction underway			
Housing development	☐	☐	☐
Commercial building	☐	☐	☐
Road or bridge repair	☐	☐	☐
Bush, forests, reserves			
Bushland area	☐	☐	☐
Water supply catchment	☐	☐	☐
Forestry area			
Crown land	☐	☐	☐
River reserve	☐	☐	☐
Wetlands	☐	☐	☐
Healthy native riparian vegetation	☐	☐	☐
Exotic riparian vegetation	☐	☐	☐
Other (name)	☐	☐	☐

1.8 Site assessment for the online database

Site name: .....

Date: ..... Time: .....

Fill in the information below and tick the boxes to provide an assessment of your monitoring site. If any parts of the assessment are **not** relevant to your site, write N/A.

Water body type

Tick the box of the water body type in the table below that best describes your site:

Freshwater rivers and streams	Tick
Upland river >150 metres	☐
Lowland river <150 metres	☐

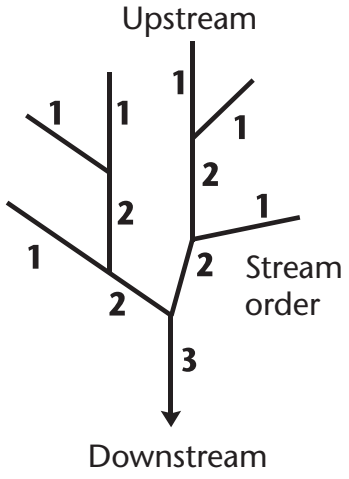
Estuary/marine	Tick
Estuary	☐
Coastal stream (tidal)	☐

Standing water (fresh)	Tick
Lakes/reservoirs	☐
Dam	☐
Wetland	☐

Site study area

For rivers and creeks **only**

Length of reach or bank: assessed ..... metres



Tick the box that best describes your site:

Stream order: 1st or 2nd order (mainly small non-perennial streams) ☐

≥ 3rd order (mainly larger perennial streams) ☐

Upstream and downstream site comparison

Tick the box that best describes your site compared to upstream and downstream sites.

Up-stream (tick)	Features compared to my site	Down-stream (tick)
☐	Similar to my site	☐
☐	More disturbed	☐
☐	Less disturbed	☐
☐	Undisturbed (natural) area	☐
☐	Wetland	☐

Does the current health of this site impact on sites lower in the catchment?

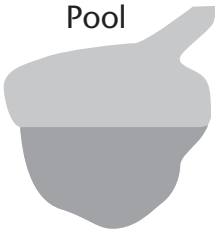
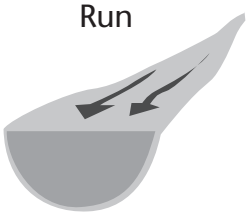
☐ Yes ☐ No ☐ Unknown

Does the current health of upstream sites impact on this site?

☐ Yes ☐ No ☐ Unknown

In-stream features

Natural

	Tick
Pool 	☐
Riffle 	☐
Run 	☐
Other (name)	☐

Artificial

	Tick
None	☐
Dam/weir upstream	☐
Water diversion upstream	☐
Weir pool at site	☐
Constructed wetland at site	☐
Other structures that affect flow (name)	☐

Banks

Looking downstream

Bank height

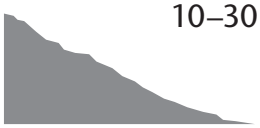
Left bank (tick)	Height in metres	Right bank (tick)
☐	0–2	☐
☐	2–5	☐
☐	>5	☐

Bank shape

The banks at the site have the following shape. (Tick the shape that best matches your left and right bank.)

Left bank (tick)	Slope	Right bank (tick)
☐	Concave 	☐
☐	Convex 	☐
☐	Stepped 	☐
☐	Wide lower bench 	☐
☐	Undercut 	☐
☐	Artificial banks	☐

The slope of the bank can be described as:

Left bank (tick)	Slope	Right bank (tick)
☐	80–90 deg.  **Vertical**	☐
☐	60–80 deg.  **Steep**	☐
☐	30–60 deg.  **Moderate**	☐
☐	10–30 deg.  **Low**	☐
☐	<10 deg.  **Flat**	☐

Bank erosion

Looking downstream

Tick the box that best describes erosion at the site.

Left bank (tick)	Erosion	Right bank (tick)
☐	Severe	☐
☐	Moderate	☐
☐	Little or no erosion	☐

Bank stability factors

Looking downstream

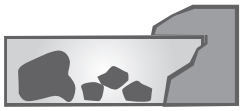
Tick the boxes that best describe factors affecting bank stability at the site.

Bank stability factors	Tick
None	☐
Stock access/crossing	☐
Vehicle tracks	☐
Roads/jetty/bridges	☐
Cleared vegetation	☐
Gravel and sand extraction	☐
Mining	☐
Unfenced riverbanks	☐
Pipes/drains	☐
Other (name)	☐

Erosion control structures at the site

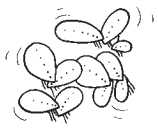
Structures at the site	Tick if visible
None	☐
Fences	☐
Concrete lined channel	☐
Concrete/rock wall/basket	☐
Logs strapped to banks	☐
Breakwater	☐
Other (name)	☐

In-stream habitats

In-stream habitat	Tick if present
Leaves and twigs 	☐
Logs/branches 	☐
Tree roots 	☐
Water plants 	☐
Silt/sand	☐
Stones/pebbles 	☐
Human made structures 	☐

Aquatic plants

Identify the form of the aquatic plants by ticking the boxes if present.

Habitat and form	Tick if present	Plant name
Free-floating 	☐	
Floating but attached 	☐	
Submerged (not feathery) 	☐	
Submerged and emergent (feathery) 	☐	
Emergent (narrow leaf) 	☐	
Emergent (broad leaf) 	☐	

Riparian vegetation along banks

For your site, tick the box that best describes vegetation along the banks.

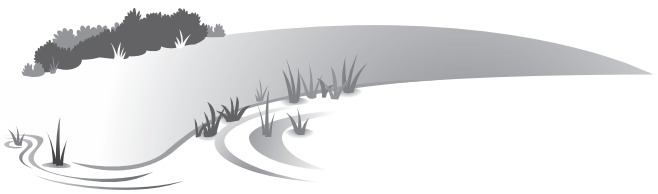
Features of riparian vegetation	Left bank (tick)	Right bank (tick)
Wide corridor of mainly undisturbed native vegetation 	☐	☐
Well vegetated with native and/or introduced species 	☐	☐
Narrow corridor of native and/or introduced species 	☐	☐
Clumps of native and/or introduced species 	☐	☐
Little or no riparian vegetation 	☐	☐

Local land use

Tick land use	Land use	Per cent
☐	Cropping	
☐	Grazing	
☐	Urban	
☐	Industrial	
☐	Mining	
☐	Fishing	
☐	Recreation	
☐	Native bushland, reserves or wetlands	
☐	Other (name)	

Significant Aboriginal landscape features

See Section 1-10 and 1-11.



1.9 Wetland site summary checklist

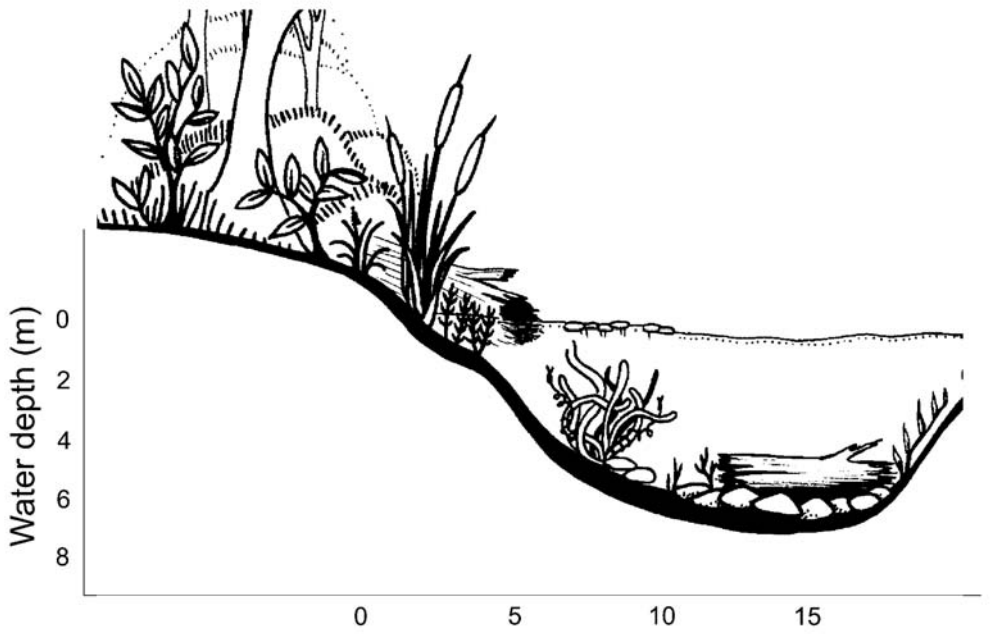
Lakes, billabongs and constructed wetlands (farm dams)

From your knowledge of the wetland you are monitoring, tick the description that best matches your wetland.

Feature	Description	Tick
Type of wetland	Natural	☐
	Modified natural wetland	☐
	Constructed urban (e.g. stormwater)	☐
	Constructed rural (e.g. farm dam)	☐
Shape	Round (e.g. lake)	☐
	Curved (e.g. billabong)	☐
	Other (name)	☐
Water source	River	☐
	Intermittent creek	☐
	Local runoff	☐
	Spring	☐
Water depth (when full)	Shallow <1 metre	☐
	Deep >1 metre	☐
Flooding patterns – frequency of fill	Every year	☐
	Every 2–3 years on average	☐
	Every 3 or more years average	☐
Timing of flooding	Spring	☐
	Summer	☐
	Autumn	☐
	Winter	☐
Drying patterns	Dries within 3 months of filling	☐
	Dries within 4–12 months of filling	☐
	Holds water for 1–2 years	☐
	Holds water for more than 2 years	☐

Feature	Description	Tick
Timing of drying	Spring	☐
	Summer	☐
	Autumn	☐
	Winter	☐
Aquatic plants	Present	☐
	Absent (omit next question)	☐
Types of aquatic plants	**Emergent plants** (plants have stems, flowers and most leaves above the water)	☐
	Submerged plants (plants have stems, flowers and leaves under the water)	☐
	Floating plants (plants that float on the water surface)	☐
	Fringing plants (reeds, rushes, sedges)	☐
Land use adjacent to the wetland	Agriculture	☐
	Urban	☐
	Recreation	☐
	Native bushland	☐
	Other (name)	☐

Adapted from *Wetlands Watch: a field guide for monitoring wetlands*

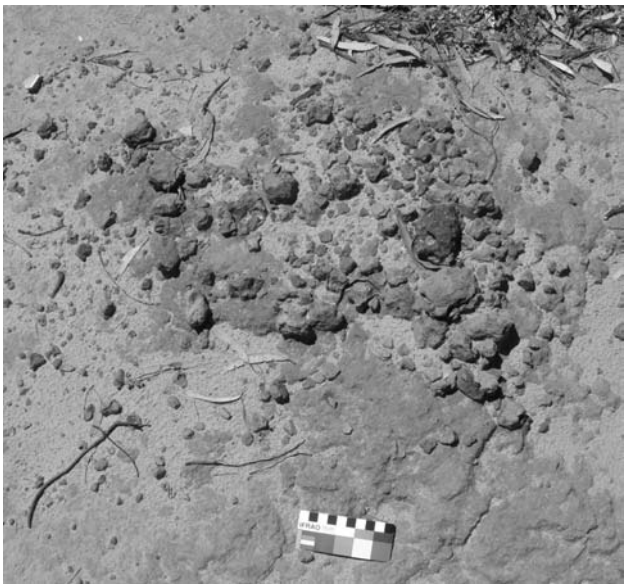


1.10 Landscape features of Aboriginal significance: background information

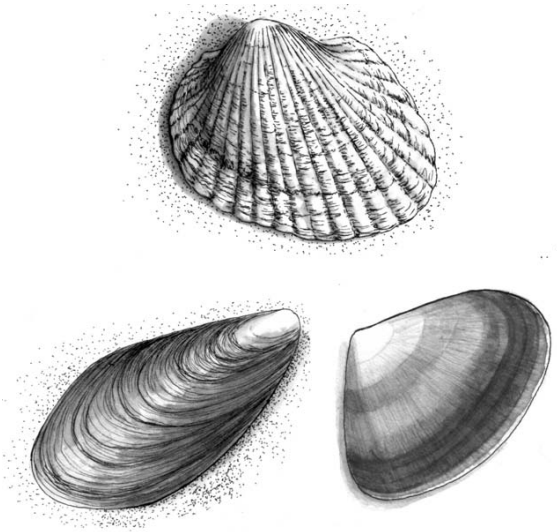
Waterways	Aboriginal significance
Stream, river, wetland or watercourse	Waterways are significant to Aboriginal people for fish, food (bush tucker), water, medicines, trade routes and storylines.
Hanging swamps	Hanging swamps are significant ceremonial areas (women).
Fish traps	Food – fishing.
Riverbanks	**Aboriginal significance**
Open campsites	These areas contain evidence such as food debris, charcoal and implements.
Shell middens	Shell middens provide an indication of aquatic environments used by Aboriginal people and seasonal and annual patterns of use. Middens are found along the coast and near inland waterways and billabongs. Burial sites may be contained within middens.
Stone artefact scatters	These may identify camping, trade or food preparation sites. When many artefacts are present at one site it may be a rock quarry (extraction of stone) or knapping site (making tools).
Axe grinding grooves	Sharpening and shaping of stone implements.
Riparian vegetation	**Aboriginal significance**
Riparian vegetation	Riparian vegetation provided food (bush tucker), medicines, habitat for animals, or indicators of seasonal arrival of plants/ insects/animals.
Riparian vegetation	Bark was used for making coolamons, canoes or humpies. Coolamons were used to carry food, water, children or tools and may also have been used as a flotation device.
Scar trees	The presence of canoe scars indicates the use of the area for fishing, trade or river crossing during floods.
Carved trees	Carved trees indicate significant sites such as initiation grounds, boundary markers or grave sites.

Landscape features	Aboriginal significance
Rocky or sandy hills, mountains, claypans, or rock shelters including caves and sandstone overhangs	Outcrops of rock such as sandstone or granite overhangs may form cave-type shelters. They may contain Aboriginal rock art sites, camp sites, ceremonial sites, burial grounds or other significant Aboriginal sites.
Elevated sites with long sight lines	These areas provide vantage points to observe landscape features and the movement of animals and people. They can be important for protection of men's and women's sites, greetings and may also be spiritual sites.
Large rock outcrops surrounded by flat plains	

Scar tree, Toorale Station.
Photo: S. Brown/DECCW



Hearth, Toorale Station.
Photo: S. Brown/DECCW



1.11 Landscape features of Aboriginal significance checklist

Rivers, lakes, wetlands and billabongs are significant sites for Aboriginal people. The following checklist will provide an indication of whether your site contains landscape features of significance to Aboriginal people.

Complete the checklist by ticking the features observed at your site.

Site name				
Date of assessment				
Features	**Indicators**	**Aboriginal significance**	**Yes (tick)**	**No (tick)**
WATERWAYS				
Stream, river, wetland or watercourse	Is the site a perennial river, creek, lake, wetland or watercourse?	Waterways are significant for fish, food (bush tucker), water, medicines, trade routes and storylines.	☐	☐
	Is the site a non-perennial river, creek, lake, wetland or watercourse?		☐	☐
	Is the area a hanging swamp?	Hanging swamps are significant ceremonial areas (women).	☐	☐
Fish traps	Are there stones in-stream (when flowing), arranged in a design (circle, semicircle or square)?	Food – fishing	☐	☐
RIVERBANKS				
Open campsites	Is there any evidence of stone scatters, bones, burnt clay nodules or charcoal in a selected area?	These areas contain evidence such as food debris, charcoal and implements.	☐	☐

Features	Indicators	Aboriginal significance	Yes (tick)	No (tick)
RIVERBANKS				
Shell middens	Is there evidence of an accumulation of shellfish, bones, stone tools, and charcoal from campfires in a selected area?	Shell middens provide an indication of aquatic environments used by Aboriginal people and seasonal and annual patterns of use. Middens are found along the coast and near inland waterways and billabongs. Burial sites may be contained within middens.	☐	☐
Stone artefact scatters	Are there implements, e.g. stone axe heads, grinding stones or cutting blades?	These may identify camping, trade or food preparation sites. When many artefacts are present at one site it may be a rock quarry (extraction of stone) or knapping site (making tools).	☐	☐
	Are there angular stones or debris from the manufacture of implements?		☐	☐
Axe grinding grooves	Are there axe grinding grooves apparent, especially in sandstone areas close to water?	Sharpening and shaping of stone implements.	☐	☐
Other	Are there any other indicators of Aboriginal use of the area? Please state:		☐	☐
RIPARIAN VEGETATION				
Riparian vegetation	Is the riparian area relatively undisturbed, especially near wetlands? It is important to protect riparian vegetation.	Riparian vegetation provided food (bush tucker), medicines, habitat for animals or indicators of seasonal arrival of plants/ insects/animals.	☐	☐

Features	Indicators	Aboriginal significance	Yes (tick)	No (tick)
RIPARIAN VEGETATION				
Scar trees	Are there large mature trees that may have potential to be scar trees (especially if located close to wetlands)?	Bark was used for making coolamons, canoes or humpies. Coolamons were used to carry food, water, children or tools and may also have been used as a flotation device.	☐	☐
		The presence of canoe scars indicates the use of the area for fishing, trade or river crossing during floods.	☐	☐
Carved trees	Are there trees that have geometric patterns or designs carved into the trunk?	Carved trees indicate significant sites such as initiation grounds, boundary markers or grave sites.	☐	☐
LANDSCAPE FEATURES				
Topography	Does the landscape surrounding your site contain any of the following landforms: rocky or sandy hills, mountains, claypans, or rock shelters including caves or sandstone overhangs?	Outcrops of rock such as sandstone or granite overhangs may form cave-type shelters. They may contain Aboriginal rock art sites, camp sites, ceremonial sites, burial grounds or other significant Aboriginal sites.	☐	☐
	Are there elevated sites with long sight lines?	These areas provide vantage points to observe landscape features and the movement of animals and people. They can be important for protection of men's and women's sites, greetings and may also be spiritual sites.	☐	☐
	Are there any large rock outcrops surrounded by flat plains?		☐	☐

(Adapted from a checklist developed by Geoffrey Simpson, Murrumbidgee CMA.)

Additional information:

Additional information about your site and the traditional owners of the land may be obtained by contacting:

- National Parks and Wildlife Service: Site officer
- Local Aboriginal Land Council: Cultural Heritage Officer
- Catchment Management Authority: Aboriginal Extension Officer.

SECTION 2

Waterwatch equipment

There are a number of specialised pieces of equipment for measuring the various parameters used to evaluate the health of a waterway. These are all provided in the Waterwatch kit available from your local Waterwatch Coordinator.

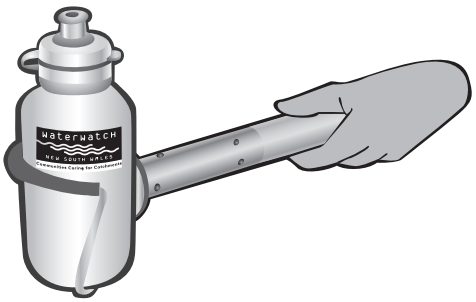
Waterwatch uses standardised equipment. It has been selected for precision and accuracy, so that your monitoring will produce the best results. Only data collected using this equipment can be entered on the Waterwatch online database.

This section of the manual lists equipment needed to conduct water quality tests and how to care for it. Detailed instructions are provided for calibrating specialised devices such as electrical conductivity (EC) meters and how to convert between various units of measurement.

2.1 Equipment list and Senior Waterwatch kits

Measuring equipment

Test	Equipment	Measurement units
Turbity (clarity of water)	Turbidity tube	Nephlometric turbidity unit (NTU)
Salinity (surface and groundwater)	Electrical conductivity (EC) meter	EC units – µS/cm, mS/cm
Temperature (air and water)	Thermometer	°Celsius
pH (acidity/alkalinity)	pH papers	0 – 7 – 14 pH scale
Available phosphate Dissolved oxygen	Colorimeter Smart, Smart 2, Phosphate (PO4) Dissolved oxygen	milligrams/L (mg/L) = ppm milligrams/L (mg/L) = ppm
Available phosphate	Colorimeter DC1200 Phosphate (PO4)	milligrams/L (mg/L) = ppm
Dissolved oxygen	Dissolved oxygen titration	milligrams/L (mg/L) = ppm
E. *coli* (total coliforms)	Petrifilm E. *coli* plates	CFU/100 mL



Accessories

As well as the equipment to test the water quality parameters, Waterwatch groups will require the following additional equipment and consumables. It is recommended that Waterwatch groups purchase an extra box to hold the following accessories (if not in the kit):

- Water sampling bottles
- Extension pole and water sampler
- Small specimen containers
- Wash bottle
- Latex gloves
- Paper towel
- Deionised water
- Calibration liquid
- Liquid waste container
- Dry waste bag.

Senior Waterwatch kits

Community groups and secondary schools use Waterwatch kits fitted with a colorimeter for measuring available phosphate and dissolved oxygen (Smart and Smart 2 kits).

The Phosphate DC1200 colorimeter kit contains the same equipment as the Smart and Smart 2 kits, but the colorimeter only measures phosphate and groups are required to measure oxygen by titration.

Waterwatch colorimeter kit contents:

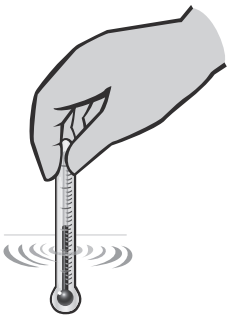
- Colorimeter and 4 test tubes
- Dissolved oxygen kit
- Phosphate kit – low range
- Filter paper holder and filter papers
- EC meter
- Calibration liquid
- Thermometer (armoured)
- Tape measure and stopwatch
- pH papers
- Turbidity tube.

Note: DC1200 requires equipment for dissolved oxygen titration.

2.2 Caring for your equipment

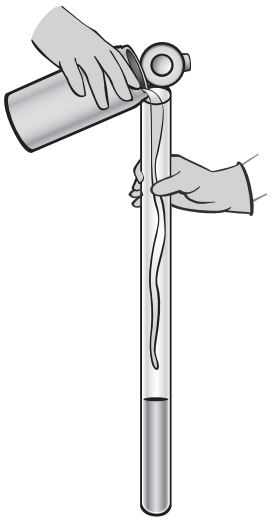
Thermometer

- Store the thermometer in a cool place.
- If the blue alcohol liquid in the tube develops bubbles or separates, run gradually warmer water along the tube until the bubbles disappear or the liquid rejoins.



Turbidity tubes

- Turbidity tubes should be kept clean. Rinse after use, and wash periodically in warm soapy water.
- Apply petroleum jelly lightly to the join occasionally for ease of assembly.



pH papers

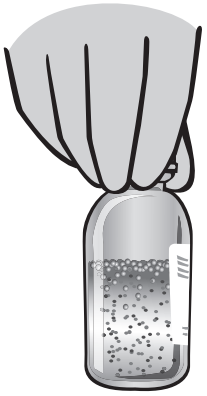
- Dispose of pH papers into a solid waste container. Never leave the papers on the bank of the creek or river.

Electrical conductivity (EC) meters

- Keep the meter in a cool place and replace batteries regularly as flat batteries will produce inaccurate results.
- Immerse only the probes of the meter in the water and rinse them with deionised water after use.
- Calibrate the meter regularly for accurate results.

i **Equipment tip:** **Never leave your meter in the car as it can overheat and destroy the internal thermometer.**

Bottles and tubes



- Wash all equipment with deionised water after use.
- Turn the bottles and tubes while rinsing to ensure all surfaces are washed.
- Dry the outside of containers with paper towel – do not dry the inside of the bottles and tubes.
- After each bottle or jar has been used, replace its lid and return it to its specific place in the kit. This avoids lids going on the wrong bottles and contaminating the contents.
- Always hold colorimeter bottles by the neck to avoid putting finger marks on the glass, as this will affect the results.
- Do not use cracked or scratched colorimeter bottles.

Reagent droppers

- Hold the droppers vertically above the test bottles, not at an angle. The droppers have been specifically calibrated to deliver exact amounts of solution when they are held vertically. If they are held at an angle, they will deliver less solution.
- Do not allow the droppers to touch the test bottles or tubes – this will contaminate the droppers.

Spoons

- Do not wet the spoons or allow them to touch the inside of the test bottle or tube.
- Dry the spoons thoroughly if they do become wet.

Colorimetric tubes

Routine maintenance

- The tubes are made of special crystalline glass which allows light to pass through without being refracted. Any lint or scratches on the glass will reduce the accuracy of the results. Always dry the glass thoroughly to remove moisture and chemicals before placing it in the colorimetric chamber.

- Do not allow moisture to enter the colorimeter chamber as it may cause the colorimeter to corrode and malfunction.
- Return colorimetric tubes to their designated place in the kit.

Periodic maintenance (by an adult or supervising teacher)

> ***i*** **Safety tip: Latex gloves and safety glasses are required.**

Colorimetric bottles should be cleaned periodically in an acid wash by the following method:

1. Rinse each bottle with phosphate-free detergent.

2. Rinse 3 times with tap water.

3. Fill the colorimetric tube with hydrochloric acid (36%) and invert several times.

4. Leave the acid in the tube for approximately 20–30 minutes.

5. Pour the acid into the liquid waste container.

6. Rinse 3 times with deionised water.

7. Allow to dry, check cleanliness, replace the lid and return the tubes to the kit.

2.3 Measuring electrical conductivity

Salinity is measured using an EC meter. EC meters measure electrical conductivity in µS/cm or mS/cm (= dS/m). A dual range meter measures both µS/cm and mS/cm.

EC meters

Type of meter	Range	Unit of measurement	Comments
*ECScan Low *ECScan High	0-19999 0-19.99	microsiemens/cm (µS/cm) millisiemens/cm (mS/cm)	No conversion required Convert mS/cm to µS/cm X1000
EC Scan Dual Range	0-19999	microsiemens/cm (µS/cm) – low range millisiemens/cm (mS/cm) – high range	Measurement changes from µS/cm to mS/cm as higher levels of salt are recorded Convert mS/cm to µS/cm

Converting units of measurement

Measurement tip: 1 mS/cm = 1000 µS/cm = 640 mg/L = 640 ppm

Electrical conductivity (EC)

1000 µS/cm = 1 mS/cm = 1 dS/m

To convert µS/cm to mS/cm or dS/m, **divide** by 1000

To convert mS/cm or dS/m to µS/cm, **multiply** by 1000

2.4 Using your EC meter

What does the EC meter measure?

As salts conduct electricity, electrical conductivity (EC) can be used to estimate the amount of salt in a water sample or soil/water solution. EC readings increase as salinity levels increase.

Waterwatch kits contain a low or high or dual range EC meter.

- **Low range** meters are for use in freshwater areas with low levels of salinity.
- **High range** meters are best for saline areas (and coastal streams) or for monitoring groundwater.
- **Dual range** meters are best when testing at sites where there are large differences in salinity, such as along a coastal stream influenced by tides or when measuring surface water and groundwater.

All meters have the following features:

- waterproof and float
- replaceable sensors
- push button calibration (and automatic calibration for dual range meters)
- automatic temperature compensation

Additional features of the ECtestr 11 and 11+ are:

- automatic and manual calibration
- ability to calibrate to different EC ranges
- temperature readings in °C or °F.

Specifications for the ECScan and ECTestr meters

Meter	Salinity measure	Range	Resolution	Calibration standard solutions
ECScan High	mS/cm	0–19.9	0.01 mS/cm	12.88 mS/cm
ECScan Low	µS/cm	0–1999	1.0 µS/cm	1413 µS/cm, 500 µS/cm
ECTestr 11 and 11+	mS/cm µS/cm	0–20 High 0–2000 Low	0.1 mS/cm 10.0 µS/cm	12.88 mS/cm 1413 µS/cm, 84 µS/cm

Note: Meters should be calibrated with a solution that is similar in EC to the water tested.
The dual range meter can be calibrated in both low and high ranges.

Key button functions on an EC meter

Button	Meter	Function
Hold	High/Low	Freezes the display in measurement mode Exits calibration mode without confirming calibration
	Testr 11; 11+	Freezes the display in measurement mode In hold mode, transfers to measurement mode Exits calibration mode without confirming calibration In range selection mode, selects a measurement range
INC/DEC	High/Low Testr 11; 11+	In measurement mode, enters calibration mode In calibration mode, adjusts the calibration manually
°C/°F	Testr 11; 11+	°C/°F key permits the selection of a monitoring range. Press and hold °C/°F key while turning off the meter. Turn the meter back on and release the °C/°F button. The meter goes to the range selection mode. Use the hold key to select the range.

Error messages

If the salinity is out of range, your meter display will show the letters OR (over range). To overcome this problem you need to dilute the sample.

Meter maintenance

Calibrate the meter before each use.

Rinse the electrodes with deionised water and dry in the air or by blowing on the probes, including between samples.

If the electrodes become green, dirty or rusty, soak them in methylated spirits for 10–15 minutes and then blow or wipe dry with a cotton bud.

Ensure the batteries are replaced regularly for accurate readings.

Calibration

What is calibration?

Calibration means adjusting the meter reading to ensure it conforms to a known salt solution. It is advisable to calibrate your meter with a solution similar to the EC of your waterway.

Calibration ensures the accuracy of data and should be done each time you use the equipment. Meters must be calibrated regularly to ensure consistent readings.

Calibration procedure

1. Use a standard calibration solution. This may be 500 µS/cm, 1413 µS/cm or 12.88 mS/cm.

2. The calibration liquid should be at **room temperature**.

3. Pour a small amount of calibration solution (about 2–3 cm) into a small clean container, such as the small beaker in your kit or a specimen jar.

4. Turn the conductivity meter on and place the electrodes in the solution.

5. Wait for the display to stabilise. If the meter does not read the same as the known calibration solution, you will need to calibrate.

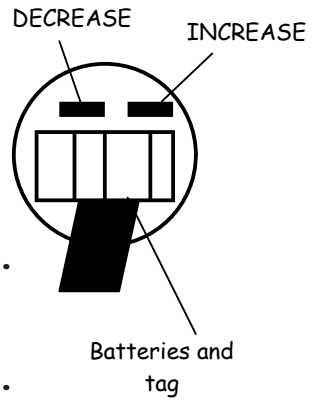
> **Testing tip: Calibration liquid must be made in a NATA accredited laboratory or purchased from a commercial supplier.**

Calibrating by site

It is best to calibrate in the EC range closest to that of your waterway. If more than one site is monitored and they are very different in EC (e.g. surface water and groundwater or freshwater stream and estuary), calibrate for each site with an appropriate standard.

Calibrating the ECScan low/high range meter

Procedure



1. Unscrew the top of the meter (battery compartment) and identify the white buttons (INC-DEC buttons).

2. Orientate the battery compartment as shown in the diagram.

3. Turn the meter on (by pressing the on/off button).

4. Dip the electrodes into the standard calibration solution and swirl the container, meter and solution.

5. Wait several seconds until the number stabilises.

6. Press the INC or DEC key to adjust the reading to match the calibration standard value.

7. After 3 seconds without pressing any buttons, the display will flash 3 times then show 'ENT'.

8. Turn off the meter. Dry the meter and replace the cap.

9. The meter is now calibrated.

> ***i*** **Testing tip: If the meter doesn't calibrate correctly the first time, just repeat the steps.**

> ***i*** **Testing tip: Discard the calibration solution after use. Never return it to the container.**

ECTestr 11 and 11+

The 11 and 11+ EC meters are dual range meters that permit the measurement of both high and low levels of salinity.

The meter uses different measurement units, depending on the salinity of the sample:

Selecting your measuring range

Selection	To be used when measuring water between:	Measuring range	Resolution
HI	2000 µS/cm and 20,000 µS/cm only	0–20 mS/cm (20,000 µS/cm)	0.1 mS/cm (100 µS/cm)
LO	0 and 2000 µS/cm only	0–2000 µS/cm	10 µS/cm
AUTO	0 and 20,000 µS/cm OR 0 and 20 mS/cm	0–2000 µS/cm 0–20 mS/cm (20,000 µS/cm)	10 µS/cm (LOW) 0.1 mS/cm (100 µS/cm) (HIGH)

How to select a measuring range

1. Ensure meter is switched off.

2. Hold the °C/°F button and then switch the meter on by pressing the ON/OFF button. Release the °C/°F button once the meter is turned on.

3. Select either HI, LO or AUTO by pressing the HOLD/ENT key to switch between each measuring range.

4. Once the correct range appears on the screen do not press again (if nothing is pressed for 5 seconds the meter will confirm the selection by displaying 'CO' and will then go into measurement mode).

The range does not need to be set again unless you wish to change the range.

Calibration of the ECTestr 11 and 11+

The dual range EC meter has automatic calibration.

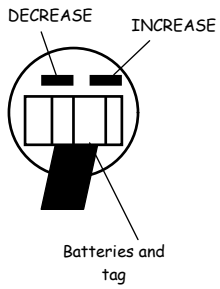
Meters should be calibrated across the entire measuring range over which they will be used. If meters are used in **both** high and low ranges, you will need to calibrate **both** ranges (i.e. multi point calibration). If you are using the meter in a single range, calibrate using the appropriate standard solution for that range (single point calibration). The following is a guide:

- HI – use single point calibration
- LO – use single point calibration
- AUTO – use multi point calibration.

Standard Solutions

Selected measuring range	Calibration standard
0–200 µS/cm	500 µS/cm
Low Range 0–2000 µS/cm	1413 µS/cm
High Range 0–20 mS/cm	12.88 mS/cm
AUTO	500 µS/cm; 1413 µS/cm; 12.88 mS/cm

How to change between single and multi point calibration

Procedure	Instructions on screen
1. Unscrew the top of the meter (battery compartment) and identify the white keys.	
2. Hold down one of the buttons on the right hand side, while pressing the ON/OFF button.	Press ON/OFF AND Press one of the white buttons (INC/DEC key) at the same time
3. The display will show 'A.CAL', and will flash 'YES'. Press the °C/°F button to skip.	YES A.CAL
4. The display will show '1PNT', and will flash 'YES' or 'NO'. Use either button under the cap to toggle between 'YES' and 'NO' and then press HOLD/ENT to make a selection. (Choose 'YES' if measuring in high or low range, or 'NO' if measuring in dual range.)	1PNT YES NO

Single point calibration (AUTO)

A standard solution appropriate to the range of measurement **must** be used.

1. Screw off the top cover and remove protective cap from the bottom.

2. Press the ON/OFF button to turn the meter on.

3. Press either button under the top cover.

4. Dip the electrodes into the standard calibration solution and swirl the container, meter and solution.

5. Wait until the top number stabilises (this is what the meter is reading **before** calibration).

6. Press HOLD/ENT. The meter will adjust to match the calibration standard value.

7. The meter shows 'CO' for 2 seconds and calibration is complete.

8. The upper display shows the conductivity reading and the lower display shows calibration standard values (see EC specifications) in 'AUTO'.

9. Rinse the probes in deionised water.

10. The meter is now calibrated in a single range (high or low).

11. The meter will now enter measurement mode.

> ***i*** **Testing tip: Discard the calibration solution after use. Never return it to the container.**

Multi point calibration (AUTO)

1. Screw off the top cover and remove protective cap from the bottom.

2. Press the ON/OFF button to turn the meter on.

3. Press either button under the top cover.

4. Pour **500** or **1413 µS/cm** calibration solution over the electrode.

5. Immerse the electrode in **500** or **1413 µS/cm** calibration solution.

6. Wait until the top number stabilises (this is what the meter is reading **before** calibration).

7. Press the HOLD/ENT button.

8. Pour deionised water over the electrode.

9. Pour **12.88 mS/cm** calibration solution over the electrode.

10. Immerse the electrode in **12.88 mS/cm** calibration solution.

11. Wait until the top number stabilises (this is what the meter is reading **before** calibration).

12. Press the HOLD/ENT button.

13. The meter will now enter measurement mode.

> *i* **Testing tip: Discard the calibration solution after use. Never return it to the container.**

Error messages

If during calibration the error message 'Er.O' appears, the calibration liquid is too hot or cold. Calibration liquid should be stored in the fridge but used at **room temperature**.

If during calibration the error message Er.1 appears, you have pressed the HOLD/ENT key before the tester has recognised the calibration standard.

> *i* **Equipment tip: To return your meter to the factory default settings press HOLD/ENT and switch on the tester using the ON/OFF key. Release the HOLD/ENT key.**

SECTION 3

Testing water quality in the field

The water quality tests included in the Waterwatch program will allow students and community members to measure and understand important catchment issues. Participants will learn about key water quality issues and the interaction between what happens on nearby land and the health of their waterway.

This section provides information on safety considerations for Waterwatch tests and how to ensure that the data collected is of the highest quality, plus methods for preserving samples and diluting them if needed.

Included in this section: *Page*

3.1 Occupational Health and Safety

It is important that you always follow the safety instructions when sampling water or carrying out the Waterwatch tests. There are safety tips throughout this manual. For more specific information on the chemicals supplied in your Waterwatch kit please refer to the material safety data sheets (MSDS) provided in Section 5 of this manual.

Always wear gloves when carrying out the Waterwatch tests.

- Gloves will help protect you when using chemicals for Waterwatch tests.
- Gloves will also help to avoid contamination of your water sample from substances you may have on your hands.
- Always remove your gloves after you have finished testing and dispose of them in your waste paper container. Wash your hands after testing.

Always work in pairs (with a buddy) at the water's edge.

To do this, one person collects the water sample while their buddy holds onto the back of their shirt. It is very important to look after each other in this way.

Always wear safety glasses when handling Waterwatch chemicals or when observing or assisting with testing.

Safety glasses will protect you from chemicals that could flick or spray into your eyes.

Hold all tubes and bottles over the liquid waste container while adding reagents.

- Pour water into the liquid waste container after each piece of equipment has been rinsed. Do not put solid waste into the liquid waste container.
- To dispose of liquid waste, dilute the contents of the liquid waste container with twice the volume of tap water and then flush the liquid waste down the sink.

> ***i*** **Testing tip: Place all used gloves, paper towels, empty reagent packaging and any other rubbish from the day into a garbage bag or bin.**

3.2 Collecting quality data

Follow these tips to ensure the data you collect is of the highest quality.

Keep your equipment clean	Poorly cleaned equipment can lead to inaccurate results. Refer to Section 2.2 for specific instructions on cleaning and maintenance of equipment.
Follow the testing instructions closely	You are using scientific testing equipment. Following the procedures carefully will ensure the most accurate results.
Collecting your sample	Collect samples according to the instructions. Some tests, such as dissolved oxygen and faecal coliform tests, require special collection methods. Temperature should be tested at the waterway.
Record in the right units	Each test uses different measurement units. It is important that results are reported using the correct units. Some measures, such as electrical conductivity as a measure of salinity, require conversion.
Recording at the site	Record your results on the forms provided. This will ensure that all necessary information is recorded at the site. Never use scraps of paper.
Chemical storage	The storage and use of chemicals should be carefully monitored. Chemicals past their expiry date should not be used. Store your kit flat, right way up to prevent spills from loose caps or lids.
Correction factors	Some test results, e.g. faecal coliform, require multiplication depending on dilution factors used. Be careful to use the right dilution factors.
Calibration	Some meters require calibration to ensure they continue to record accurately, e.g. EC meters. Follow the instructions provided and calibrate prior to use.
Change the batteries	Equipment using batteries needs regular checking because low batteries can affect readings. Make sure you renew batteries at the recommended intervals.

3.3 Preserving samples

If you cannot do all the tests when the water sample is taken, some can be done later if samples are properly preserved.

Follow these guidelines for preserving samples:

Parameter	**Preservation method**	**Maximum holding time**	**Comments**
pH	Refrigeration	6 hours	Completely fill sample bottle and test as soon as possible
Conductivity	Refrigeration	30 days	Completely fill sample bottle
Turbidity	None required	24 hours	Preferably test on site
Dissolved oxygen and temperature	Not applicable	Not applicable	Must be analysed on site. *Alternatively can be fixed with 8 drops of reagents 1, 2 and 3 for brown solution. Then complete test later.*
Phosphates	Refrigeration Freezing	24 hours 30 days	Filter and fill on site. Freeze sample. Return to room temperature before testing.

Source: *Waterwatch Technical Manual,* Module 4, Physical and Chemical Parameters, p.5.

3.4 Measuring high salinity levels by diluting samples

When measuring salinity, some samples may exceed the limit of the available meter. An error message 'OR' will appear in the screen (over range). The sample will need to be diluted for your meter to measure the salinity.

Choose a dilution factor that will allow your meter to measure the result. For estuarine water a 1:5 dilution factor is recommended. For example, at 1:5 dilution: original sample = 10 mL, sample after dilution = 50 mL.

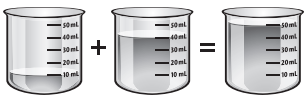
> ***i*** **Measurement tip: A 1:5 solution is 1 part sample water and 4 parts deionised water**

Instructions follow for diluting samples for use with either a TDS meter or a high range EC meter.

Diluting a sample for measurement of electrical conductivity (EC meter)

It is recommended that a high range EC meter is used when the salinity level is high.

1. Pour 10 mL of the sample into the 50 mL specimen tube or measuring cylinder and add 40 mL deionised water to make up to 50 mL.



> ***i*** **Testing tip: Using larger volumes of water (e.g. 20 mL sample water and 80 mL of deionised water) may reduce error caused by dilution.**

2. Rotate to mix thoroughly.

3. Pour the diluted sample into a clean specimen tube and mix thoroughly again by rotating.

4. Carry out the test using the EC meter.

5. Rinse the probes of the meter with deionised water and cap them.

6. Return the meter to the kit.

7. Calculate the result by multiplying the meter reading by the dilution factor of 5.

> ***i*** **Measurement tip: EC meter reading x 5 = EC result.**

8. Convert the result from millisiemens per centimetre (mS/cm) to microsiemens per centimetre (µS/cm) (for high range EC meters).

9. Record the result in the table on the result sheet.

Calculating the EC of the original sample

If the result is given in microsiemens per centimetre (µS/cm), multiply your result by the dilution factor (e.g. x5 for the above procedure).

Example 1: Diluting samples measured in microsiemens/centimetre (µS/cm)

Salinity result for diluted water sample = 600 µS/cm

Salinity of diluted water sample x dilution factor = 600 µS/cm x 5 = 3000 µS/cm

Salinity of original water sample = 3000 µS/cm

Number on screen	Dilution factor x 5	EC µS/cm
600	5	3000

If the result is given in mS/cm, multiply your result by the dilution factor, then multiply by 1000 to return the result to µS/cm.

Example 2: Diluting samples measured in millisiemens/cm (mS/cm) to microsiemens/cm (µS/cm)

Salinity of diluted water sample = 5.30 mS/cm

Salinity of diluted water sample x dilution factor = 5.30 mS/cm x 5 = 26.5 mS/cm

Salinity of original water sample = 26.5 mS/cm x 1000 (convert mS/cm to µS/cm)

Salinity of original water sample = 26,500 µS/cm

Number on screen	Dilution factor x 5	EC mS/cm	Convert to µS/cm x 1000	EC µS/cm
5.3	5	26.5	1000	26,500

Note: The dilution factor will depend on the salinity of the sample water and the type of meter you are operating. Low range meters measure in microsiemens per centimetre (µS/cm) and high range meters in millisiemens per centimetre (mS/cm). Remember to notice if the result has a decimal point and is given in mS/cm or as a whole number (µS/cm). Make sure you multiply the on-screen display by the dilution factor.

SECTION 4

Procedure sheets

The water quality tests included in the Waterwatch program will allow students and community members to measure and understand important catchment issues. Participants will learn about key water quality issues and the interaction between what happens on nearby land and the health of their waterway.

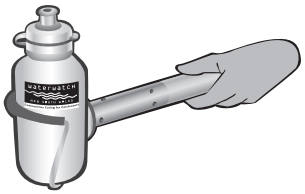
This section provides detailed procedure sheets, including how to collect a water sample and how to do each kind of test on it.

Note: Background information about the water quality tests is provided in Section 4 of the *Senior Waterwatch Teachers' Guide* and Section 4 of the *Community/Land Manager Waterwatch Guide*.

4.1 Collecting a surface water sample

Procedure

Equipment: Long arm sampler, snap adaptor and water sample bottle.



1. Use a long arm sampler so the water sample is taken away from the edge of the bank.

2. Rinse the collection bottle three times with water from downstream of the test site, i.e. with water from the same area. Empty the bottles on the bank to avoid stiring up the water you are about to test.

3. Extend the pole with the sample bottle in place. Make sure the pole is not too long and difficult to handle.

4. Have a buddy hold you while collecting the sample.

5. Turn the bottle top down over the water and submerge it about 20 cm or elbow depth if possible. In shallow water make sure you do not disturb the stream bed as this may discharge sediments that will contaminate your sample.

6. Once the bottle is under the water, turn it sideways, pointing upstream (into the direction of flow) and allow it to fill.

7. Turn the bottle upright and quickly bring it up out of the water to avoid surface scum contaminating the sample.

8. Use the **same** sample for all tests conducted at the site.

> ***i*** **Safety tip: To avoid electrocution never carry or lift the pole above your head! Always carry the extension pole horizontally and below shoulder level. This applies both on the way to the site and at the site.**

4.2 Measuring temperature

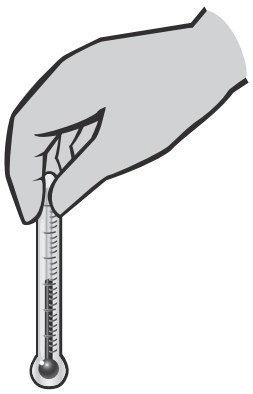
What is temperature?

Temperature is a measure of heat and cold. It is measured in degrees Celsius.

Procedure

Equipment: Thermometer, water sample bottle (optional).

Air Temperature



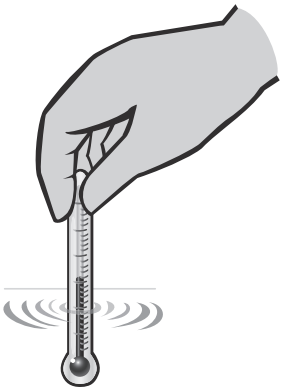
1. Measure the temperature in the shade, by holding the thermometer at waist height, by the top of the thermometer and in the shade of your body – or find a shaded place.

2. Wait for at least one minute before reading the thermometer.

3. Ask another person (if possible) to confirm your result.

4. Record your results for Air Temperature.

Water Temperature



1. Lower the thermometer into the water (in the creek or in a freshly collected sample).

2. Keep the thermometer in the water for 1 minute before taking the temperature reading.

3. Read the thermometer while it is still in the water.

4. Repeat the test to verify the results.

5. Rinse the thermometer with deionised water and put it back into the kit.

6. Record your result.

> ***i*** **Measurement tip: Temperature can also be measured from a sample bottle immediately after collection.**

What do the results mean?

There are no trigger values for temperature to apply a healthy or poor rating. The acceptable temperature range will depend specifically on your site, and any nearby features, such as large dams.

4.3 Measuring pH

What is pH?

pH is a measure of acidity and alkalinity measured on a scale of 0–14.

Procedure

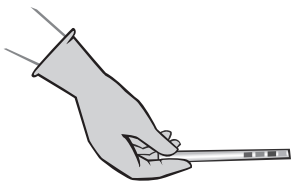
Equipment: pH papers, sample water specimen container.

1. Take the water sample you collected in Procedure 4.1.

2. Shake the sample.

3. Rinse out a specimen container with sample water 3 times.

4. Fill the container with sample water or test directly from the sample bottle.



5. Take a pH strip and dip all the coloured squares into the sample water. Make sure all colours are underwater.

6. Leave the strip in the water for **5 minutes**.

7. Remove the strip and match its colours against the colour chart to work out your pH.

8. If you cannot match the colours exactly you can estimate between two colours to 0.5 of a pH unit.

9. Pass the strip to others to verify the result.

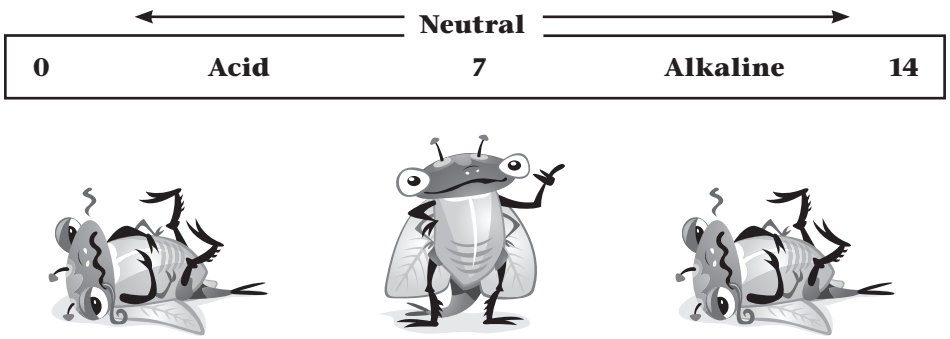
10. Dispose of the pH strip in the bin.

11. Record your result.

Additional laminated colour charts are available from the distributor.

What do the results mean?

pH



<6.5 poor | 6.5–8 healthy | >8 poor

Note: Sudden changes in pH are undesirable and may cause a loss of native plants and animals.

4.4 Measuring total dissolved solids

Make sure you use the procedure sheet that corresponds to the type of meter you have.

What is total dissolved solids (TDS)?

TDS is a measure of salinity. The TDS meter measures electrical conductivity and converts this measurement to an amount of salt in parts per million or milligrams per litre. The more salt in the water, the greater the current that will be transferred and the higher the TDS reading.

Procedure to measure total dissolved solids

Equipment: TDS meter, sample water specimen container, calibration liquid, deionised water.

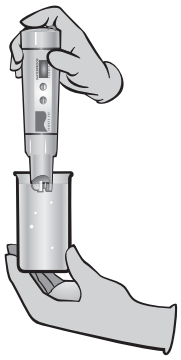
Equipment tip: Remember to calibrate your meter before testing!

1. Take the water sample you collected in Procedure 4.1.

2. Rinse out a small container with sample water at least twice.

3. Shake the water sample and pour some into the specimen container to a depth of about 3 centimetres.

4. Remove the cap from the meter and turn it on. Wait until a 0 appears.



5. Dip the meter into the specimen container of sample water so the probes are covered.

> ***i*** **Equipment tip: Do not put the meter deeper than the immersion line marked on the base. Do not rest the probes on the base of the container.**

6. Hold the meter in the sample water and rotate your wrist so that the sample water, container and meter move. Allow time for the number value to display and stabilise.

7. Read the TDS from the meter screen.

8. Identify the unit of measurement the meter is reading: parts per million for the lower range (low salinity) or parts per thousand for the higher range (high salinity).

9. Repeat the test to verify the result.

10. Turn the meter off and rinse the probes with deionised water.

11. Do not wipe the meter probes – blow on them or allow to air dry.

12. Replace the cap on the meter and put it back in the kit.

13. Record your result.

> ***i*** **Testing tip: Enter your TDS result as mg/L = ppm on the online database.**

What do the results mean?

Total dissolved solids

Healthy

Less than 200 mg/L

Fair – may affect river health

200–500 mg/L

Poor – river health at risk

Greater than 500 mg/L

Note: Average for all waterways; not specific to position in the catchment.
Adapted from *ANZECC Guidelines 2000*

4.5 Measuring electrical conductivity: low and high range meters

Make sure you use the procedure sheet that corresponds to the type of meter you have.

What is electrical conductivity (EC)?

Electrical conductivity (EC) is the amount of transfer of electricity through water and is a measure of salinity. The more salt in the water, the greater the current that will be transferred and the higher the EC.

Procedure to measure salinity: low and high range meters

For detailed instructions on setting up these meters, refer to Section 2.4.

Equipment: Electrical conductivity meter (low or high range), specimen container, sample water, calibration liquid, deionised water.

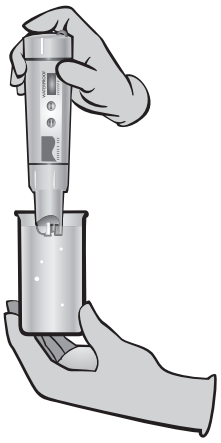
Equipment tip: Remember to calibrate your meter before testing!

1. Take the water sample you collected in Procedure 4.1.

2. Rinse out a small container with sample water at least twice.

3. Shake the water sample and pour some into the specimen container to a depth of about 3 centimetres.

4. Remove the cap from the meter and turn it on. Wait until a 00 appears.



5. Dip the meter into the specimen container of sample water so the probes are covered.

> ***i*** **Equipment tip: Do not put the meter deeper than the immersion line indicated by the shape of the base. Do not rest the probes on the base of the container.**

6. Hold the meter in the sample water and rotate your wrist so that the sample water, container and meter move. Allow time for the number value to display and stabilise.

7. Read the EC from the meter screen.

8. Identify the unit of measurement the meter is reading (µS/cm or mS/m).

9. Turn the meter off and rinse the probes with deionised water.

10. Repeat the test to verify the result.

11. Turn the meter off and rinse the probes with deionised water.

12. Do not wipe the meter probes – blow on them or allow to air dry.

13. Replace the cap on the meter and put it back in the kit.

14. Record your result.

> ***i*** **Measurement tip: Low range meters measure in microsiemens per centimetre (µS/cm). Enter this measure. High range meters measure in millisiemens per centimetre (mS/cm) also called decisiemens per metre (dS/m). Multiply the on-screen display by 1000 to convert to µS/cm.**

What do the results mean?

Electrical conductivity (EC)

Salinity is measured by electrical conductivity (EC). Increases in salinity can affect freshwater ecosystems.

Healthy

Less than 300 µS/cm

Less than 0.3 mS/cm

Fair – may affect river health

300 to 800 µS/cm

0.3 to 0.8 mS/cm

Poor – river health at risk

Greater than 800 µS/cm

Greater than 0.8 mS/cm

Note: Average for all waterways; not specific to position in the catchment.
Adapted from *ANZECC Guidelines 2000*

4.6 Measuring electrical conductivity: dual range meters

Make sure you use the procedure sheet that corresponds to the type of meter you have.

What is electrical conductivity (EC)?

Electrical conductivity (EC) is the amount of transfer of electricity through water and is a measure of salinity. The more salt in the water, the greater the current that will be transferred and the higher the EC.

Procedure to measure salinity: dual range meter (EC11 & EC11+)

Equipment tip: For detailed instructions on setting up this meter refer to Section 2.4.

Equipment: Electrical conductivity meter (dual range), specimen container, sample water, calibration liquid, deionised water.

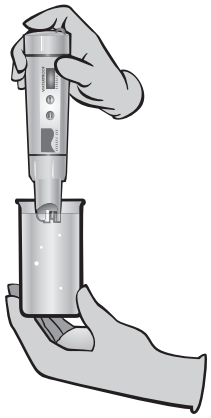
Equipment tip: Remember to calibrate your meter before testing!

1. Take the water sample you collected in Procedure 4.1.

2. Rinse out a small container with sample water at least twice.

3. Shake the water sample and pour some into the specimen container to a depth of around 3 centimetres.

4. Remove the cap from the meter and turn it on. Wait until a 0 appears.



5. Dip the meter into the specimen container of sample water so the probes are covered.

> ***i*** **Testing tip: Do not put the meter deeper than the immersion line marked on the base. Do not rest the probes on the base of the container.**

6. Hold the meter in the sample water and rotate your wrist so that the sample water, container and meter move. Allow time for the number value to display and stabilise. (The HOLD button can be pressed, so that a measurement can be read after taking the probe out of the solution.)

7. Read the EC from the meter screen. This meter will change from µS/cm to mS/cm depending on the salinity of the water being tested.

8. For dual range meters record the upper number as EC, noting whether it is µS/cm or mS/cm. Record the lower number as temperature.

> ***i*** **Measurement tip: mS/cm = dS/m = 1000 µS/cm**

9. Turn the meter off and rinse the probes with deionised water.

10. Repeat the test to verify the result.

11. Turn the meter off and rinse the probes with deionised water.

12. Do not wipe the meter probes – blow on them or allow to air dry.

13. Replace the cap on the meter and put it back in the kit.

14. Record your result in microsiemens per centimetre (µS/cm).

What do the results mean?

Electrical conductivity (EC)

Salinity is measured by electrical conductivity (EC). Increases in salinity can affect freshwater ecosystems.

Healthy

Less than 300 µS/cm

Less than 0.3 mS/cm

Fair – may affect river health

300 to 800 µS/cm

0.3 to 0.8 mS/cm

Poor – river health at risk

Greater than 800 µS/cm

Greater than 0.8 mS/cm

Note: Average for all waterways; not specific to position in the catchment.
Adapted from *ANZECC Guidelines 2000*

4.7 Measuring turbidity

What is turbidity?

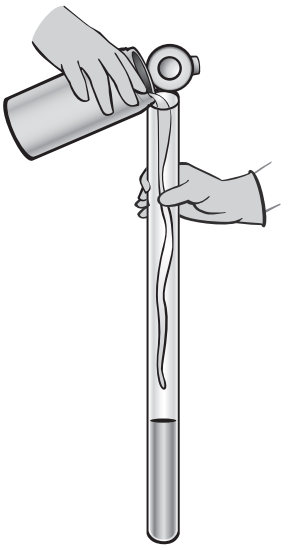
Turbidity measures the muddiness or cloudiness of the water. Suspended material such as clay, silt, sand or algae can increase the turbidity of water, affecting biodiversity, plant growth and other water uses. In general, the more suspended material there is in water, the higher the water's turbidity and the lower its clarity. This means that turbid water can have a range of colours from brown to yellow and green.

Procedure

Equipment: Turbidity tube, sample water.

1. Assemble the turbidity tube by sliding the two pieces together.

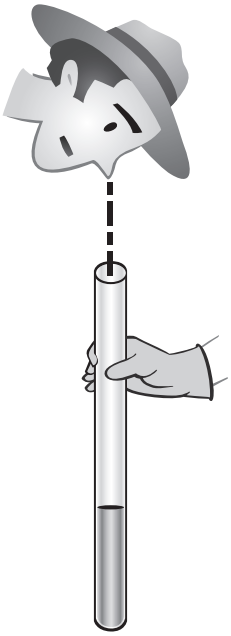
i **Testing tip: The turbidity test should be conducted in the shade**



2. One person holds the joined tube upright on the ground ready for pouring.

3. The other person shakes the water sample in the sample bottle and slowly pours it into the tube. Pour a little at a time and look down into the tube.

4. Stop pouring when the three distinct black lines at the bottom of the tube cannot be seen clearly (you may need to wait for the water to stop swirling to see whether the lines can be observed clearly).



5. Measure the turbidity by recording the last marked point **below** the level of the water. Do not estimate between the lines.

> ***i*** **Measurement tip: If you can still see the black lines when the water reaches the top of the tube, record the result as 7 NTU.**

6. Check the result by repeating the test.

7. Rinse the tube and place it back in the kit.

8. Record you result.

> ***i*** **Measurement tip: The turbidity tube has a non-linear scale so readings cannot be estimated between two numbers. Read the number immediately below the water level, e.g. correct reading is 15 when the water level is between 10 and 15.**

What do the results mean?

Turbidity

Increases in turbidity may cause a loss of plant and animal species.

Healthy
7 to 10 NTU

Fair – may affect river health
Greater than 10 to 30 NTU

Poor – river health at risk
Greater than 30 NTU

Note: Average for all waterways; not specific to position in the catchment.
Adapted from *ANZECC Guidelines 2000*

4.8 Measuring rate of flow

What is rate of flow?

The rate of flow is the speed or velocity of water movement. The rate of flow of water can be a very important influence on the environment of your stream, affecting the oxygen levels, the concentration of pollutants or salinity, and other environmental needs of living things.

Procedure to estimate the flow rate

Equipment: Stopwatch, sticks, tape measure.

1. Measure out 20 metres along the top of the stream bank (two natural markers such as trees can be used).

2. From the upstream marker, throw a stick or leaf into the water and start the stopwatch.

3. When the stick reaches the second marker, stop the stopwatch.

4. Use the time taken and the distance to calculate the flow.

5. Record your result.

Calculation of rate of flow

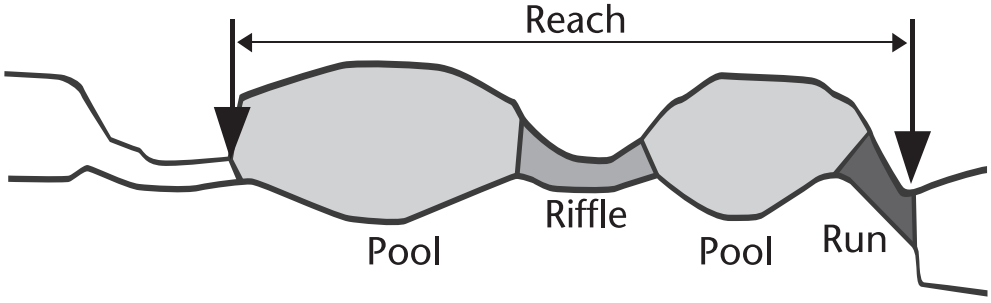
Measurement tip: Rate of flow = $\frac{\text{distance}}{\text{time}}$

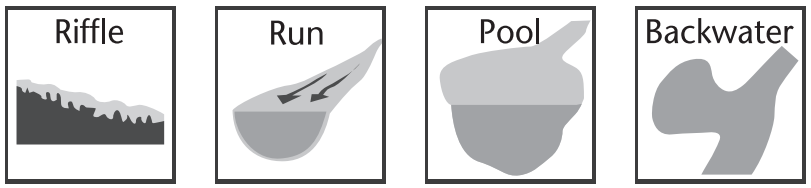
Rate of flow = distance/time, where distance is the distance between the start and finish points in metres (e.g. 20 metres) and time is the time taken for the stick to travel the distance in seconds. The unit of measurement for rate of flow is therefore metres per second (m/sec).

What do the results mean?

The rate of flow will change over time, and can be very different from one area to another. Knowing about rate of flow can help us understand what is happening with the amount of water running through your waterway.

Note: The above method gives only an indication of the speed of flow. Accurate measurement of flow involves measuring flow **volume** and requires a calculation of flow rate and stream cross-sectional area. This requires more specialised techniques.





4.9 Measuring available phosphate

What is phosphate?

Phosphorus (P) is a nutrient essential to the growth of plants and animals.

Waterwatch measures phosphates (PO_4).

Total phosphate is a measurement of all forms of phosphate compounds in a sample: orthophosphate (or available phosphate), condensed phosphates and organically bound phosphates.

Available phosphate is a measurement of the phosphate compounds that are soluble in water and therefore available to be absorbed by plants.

Risk assessment: chemical safety

Risks: chemicals used in the available phosphate test

- **Sulfuric acid** and **phosphate acid reagent** are strong acids and can cause severe burns.
- **Phosphate reducing reagent** is an irritant.

Minimising the risk

- Always wear gloves.
- Always work in pairs (with a buddy).
- Always wear safety glasses when handling chemicals.
- Hold all tubes and bottles over the liquid waste container while adding reagents.
- Recap and return reagents to kit after use.
- If any chemicals spill onto your clothes or yourself, wash them off immediately with water.

i **Safety tip: Keep the material safety data sheets (MSDS) provided in Section 5 handy for safety information on the chemicals used in this test. The first aid procedure for acid on skin is to flush it with plenty of water.**

Procedure to measure available phosphate

Equipment:

latex gloves

safety glasses

Waterwatch sample bottle (general water sample)

phosphate colorimeter tube

60 mL syringe and 0.45 micron filter paper and filter holder

liquid waste container

phosphate acid reagent and 1 mL syringe

phosphate reducing reagent and 0.1 g spoon

stopwatch

blank colorimeter bottle

deionised water

paper towel/lint-free cloth

colorimeter (Smart, Smart 2 or DC1200)

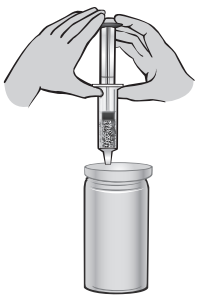
small beaker

Step 1: Filtering the sample

1. Shake the sample bottle well to mix.

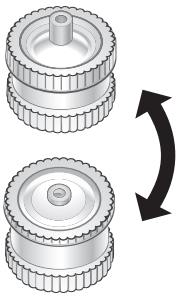
2. Remove the phosphate colorimeter tube and the 60 mL syringe from the kit.

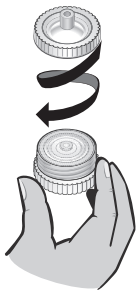
3. Rinse both twice with sample water over the liquid waste container.

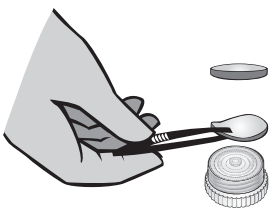


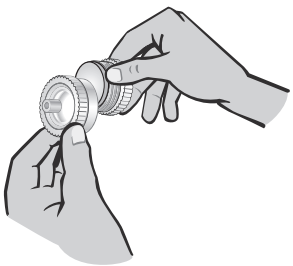
4. Using the 60 mL syringe, draw up approximately 40 mL of sample water.

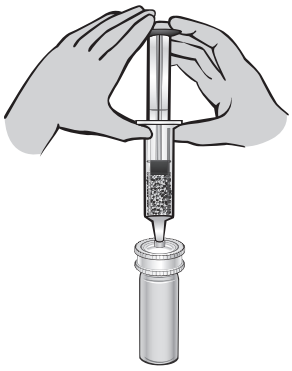
5. Insert a 0.45 micron filter paper into the filter holder.











6. Attach the filter holder to the syringe. Gently expel a small portion of the sample water through the filter into the liquid waste container before expelling a 10 mL portion into the phosphate colorimeter tube.

A plastic single use 0.45 micron filter can be used instead of the filter holder and paper filters.

7. Rinse the blank colorimeter tube with filtered sample water over the liquid waste container, then fill this colorimeter tube to the 10 mL mark with filtered sample water.

> ***i*** **Testing tip: If sample water is very turbid, filter it with pre-filter papers first to ensure enough sample water passes through the 0.45 micron filter paper.**
>
> **OR**
>
> **A pre-filter paper can be placed in front of the 0.45 micron filter in the filter holder so the sample water from the syringe passes through the pre-filter paper before the 0.45 filter paper.**

Dispose of used filter papers in the solid waste container and any excess sample water in the liquid waste container.

Step 2: Mixing the reagents

1. Remove the phosphate acid reagent and the 1 mL syringe from the kit.

Note: Make sure a safety plug has been inserted in the phosphate acid reagent bottle by the distributor.

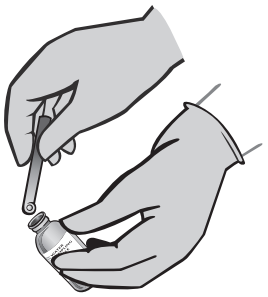
2. Draw the plunger back halfway and insert the tip of the syringe into the small hole in the top of the bottle. Push the plunger in to expel the air into the bottle. This avoids a vacuum being created in the bottle as the liquid is withdrawn.

3. Turn the bottle and syringe upside-down and while supporting both, slowly pull back on the plunger until the black stopper is aligned with the 1 mL line. If bubbles form on the black stopper, push the plunger in and redraw the phosphate acid reagent.

> ***i*** **Testing tip: This may have to be done several times to eliminate the bubbles.**

4. Turn the bottle upright and carefully remove the syringe by twisting from its base. Add 1 mL of phosphate acid reagent to the phosphate colorimeter tube.

5. Recap the tube and invert several times to mix.



6. Remove the phosphate reducing reagent and the 0.1 g spoon from the kit. Add one level spoonful of phosphate reducing reagent to the phosphate colorimeter tube.

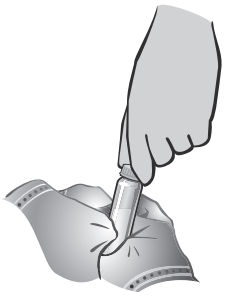
7. Do not wet the spoon. If the spoon does get wet, rinse with deionised water and wipe with a paper towel. After use, wipe the spoon with a dry paper towel. Dispose of the waste paper in the solid waste container.

8. Recap and invert several times until the crystals are dissolved.

9. Time the reaction for 5 minutes with the stopwatch.

> ***i*** **Testing tip: Have the stopwatch ready to time the reaction as once the phosphate reducing reagent has dissolved, the reaction will take exactly 5 minutes.**

10. Clean the blank colorimeter tube with lint-free cloth or paper towel to remove all smudge marks and fingerprints.



11. While waiting for the reaction, follow the appropriate instructions for your colorimeter.

Step 3: Reading the results

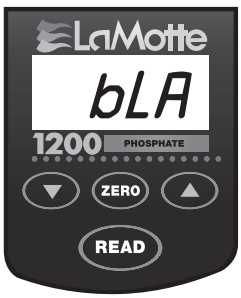
DC1200 phosphate colorimeter

1. Press the READ button for 2 seconds to turn on the colorimeter.

2. Clean and dry the colorimeter tube containing the blank.

3. Lift the lid and insert the colorimeter tube with filtered sample water, filled to the line. This is the blank.

4. Make sure that the tube has the vertical line to the front and is lined up with the arrow on the meter.



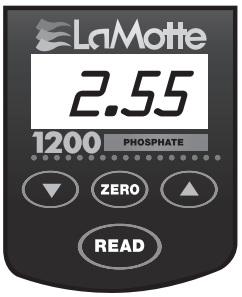
5. Press and hold down the ZERO button until the letters BLA appear. This is the blank sample and the screen should read 0.00.

6. Take out the blank sample and place it back in the kit.

7. Clean the phosphate colorimeter tube with lint free cloth or paper towel to remove all smudge marks and fingerprints.

8. Place the colorimeter tube containing the phosphate sample into the colorimetric chamber, align the vertical line with the arrow and close the lid. It will be filled to the line on the bottle.

9. Press and release the READ button.



10. The number displayed is the available phosphate reading (mg/L).

11. Record the result.

12. Remove the bottle from the colorimeter and empty the contents of both colorimeter tubes into the waste container.

Measurement tip: The result will appear in milligrams per litre (mg/L) or parts per million (ppm).

Equipment tip:
Remember to:

- **turn the colorimeter off by holding down the OFF button on the DC1200**
- **rinse the colorimeter tubes twice with deionised water over the liquid waste container**
- **clean the syringes**
- **wipe the spoon with paper towel**
- **return all equipment to the kit after use.**

Smart colorimeter

1. Press USE button to turn the colorimeter on.

2. Scroll to and select ALL TESTS from TESTING MENU.

3. Scroll to and select 33 PHOSPHATE-L from menu.

4. Clean and dry the colorimeter tube containing the blank.

5. Insert blank colorimeter tube into colorimeter chamber, cover with black cap and select SCAN BLANK.

6. Remove blank tube from colorimeter.

7. Clean the phosphate colorimeter tube with a lint-free cloth or paper towel to remove all smudge marks and fingerprints.

8. Insert phosphate colorimeter tube into colorimeter chamber and cover with black cap.

9. At the end of the 5 minute reaction time select SCAN SAMPLE.

10. Record the result.

> ***i*** **Measurement tip: The result will appear in milligrams per litre (mg/L) or parts per million (ppm).**
>
> **ppm = mg/L**

Equipment tip:
Remember to:

- **turn the colorimeter off by pressing the OFF button 3 times for Smart**
- **rinse the colorimeter tubes twice with deionised water over the liquid waste container**
- **clean the syringes**
- **wipe the spoon with paper towel**
- **return all equipment to the kit after use.**

Smart 2 colorimeter

1. Press and hold ON button until colorimeter turns on.

2. Press ENTER to start.

3. Press ENTER to select TESTING MENU.

4. Select ALL TESTS from testing menu.

5. Scroll to and select 78 PHOSPHATE-L from menu.

6. Clean and dry the colorimeter tube containing the blank.

7. Insert the tube containing the blank into colorimeter, close lid and select SCAN BLANK.

8. Remove blank tube from colorimeter.

9. Clean the phosphate colorimeter tube with lint-free cloth or paper towel to remove all smudge marks and fingerprints.

10. Insert phosphate colorimeter tube into colorimeter chamber and close lid.

11. At the end of the 5 minute reaction time select SCAN SAMPLE.

12. Record the result.

> ***i*** **Measurement tip: The result will appear in milligrams per litre (mg/L) or parts per million (ppm).**
>
> **mg/L = ppm**

> ***i*** **Equipment tip:**
> **Remember to:**
> - **turn the colorimeter off by pressing the OFF button once for Smart 2**
> - **rinse the colorimeter tubes twice with deionised water over the liquid waste container**
> - **clean the syringes**
> - **wipe the spoon with paper towel**
> - **return all equipment to the kit after use.**

Cleaning syringes

1. Pour a small amount of deionised water into the small beaker.

2. Draw water into the syringe and expel into the beaker. Repeat.

3. Dry the outside of the syringe with paper towel.

4. Rinse the beaker twice with deionised water over the liquid waste container.

Bottle cleaning

1. Wash glassware three times with deionised water.

2. Turn glassware as rinsing occurs to make sure all surfaces are cleaned.

3. Your glassware will require periodic cleaning in acid wash (see Section 2.2).

What do the results mean?

Available phosphates:

Healthy
Less than 0.06 mg/L

Fair – may affect river health
0.06–0.3 mg/L

Poor – river health at risk
Greater than 0.3 mg/L

Note: Average for all waterways; not specific to position in the catchment – see Section 6 for more information.
Adapted from *ANZECC Guidelines 2000*

Summary: available phosphate test procedure: DC1200

Step 1: Filter the phosphate sample

1. **Shake** the sample bottle well to mix.
2. Using the 60 mL syringe, **draw up** approximately 40 mL of sample water.
3. **Insert** a 0.45 micron filter paper into the filter holder.
4. **Attach** the filter holder to the syringe.
5. **Filter** 10 mL portion into two colorimeter tubes – one for the blank and one the sample.

Step 2: Mixing the reagents

1. **Add** 1 mL of phosphate acid reagent to the colorimeter tube containing filtered sample water (to the line marked).
2. **Recap** and **invert** to mix
3. **Add** 1 flat spoonful (0.1 g) of phosphate reducing reagent.
4. **Shake** until dissolved.
5. **Stand** tube for 5 minutes (a blue colour will develop if phosphate is present).

Step 3: Reading the result

1. **Press** the READ button for 2 seconds to turn on the colorimeter.
2. **Lift** the lid and **insert** the colorimeter tube with filtered sample water filled to the line. Make sure the bottle's vertical line is to the front.
3. **Make sure the bottle is wiped dry and is free of fingerprints.**
4. **Press** and **hold down** the ZERO button until the letters "BLA" appear. For the blank sample the screen should read 0.00.
5. **Place** the colorimeter tube containing the phosphate sample into the colorimetric chamber and close the lid.
6. **Make sure the tube is wiped dry and is free of fingerprints.**
7. **Press** and **release** the READ button.
8. The number displayed is the available phosphate reading (mg/L).
9. **Record** the result.

Summary: available phosphate test procedure: Smart colorimeter

Step 1: Filter the phosphate sample

1. **Shake** the sample bottle well to mix.
2. **Using** the 60 mL syringe, draw up approximately 40 mL of sample water.
4. **Insert** a 0.45 micron filter paper into the filter holder.
5. **Attach** the filter holder to the syringe.
6. **Filter** 10 mL portion into two colorimeter tubes – one for the blank and one the sample.

Step 2: Mixing the reagents

1. **Add** 1 mL of phosphate acid reagent to the colorimeter tube containing filtered sample water (to the line marked).
2. **Recap** and **invert** to mix.
3. **Add** 1 flat spoonful (0.1 g) of phosphate reducing reagent.
4. **Shake** until dissolved.
5. **Stand tube** for 5 minutes (a blue colour will develop if phosphate is present).

Step 3: Reading the results

1. **Press** USE button to turn the colorimeter on.
2. **Scroll** to and select ALL TESTS from TESTING MENU.
3. **Scroll** to and select 33 PHOSPHATE-L from menu.
4. **Clean** and **dry** the colorimeter tube containing the blank.
5. **Insert** blank colorimeter tube into colorimeter chamber, cover with black cap and select SCAN BLANK.
6. **Remove** blank tube from colorimeter.
7. **Clean** the phosphate colorimeter tube with a lint-free cloth or paper towel to remove all smudge marks and fingerprints.
8. **Insert** phosphate colorimeter tube into colorimeter chamber and cover with black cap.
9. At the end of the 5 minute reaction time **select** SCAN SAMPLE.
10. **Record** the results.

Summary: available phosphate test procedure: Smart 2 colorimeter

Step 1: Filter the phosphate sample

1. **Shake** the sample bottle well to mix.
2. **Using** the 60 mL syringe, draw up approximately 40 mL of sample water.
3. **Insert** a 0.45 micron filter paper into the filter holder.
4. **Attach** the filter holder to the syringe.
5. **Filter** 10 mL portion into two colorimeter tubes – one for the blank and one for the sample.

Step 2: Mixing the reagents

1. **Add** 1 mL of phosphate acid reagent to the colorimeter tube containing filtered sample water (to the line marked).
2. **Recap** and **invert** to mix
3. **Add** 1 flat spoonful (0.1 g) of **phosphate reducing reagent.**
4. **Shake** until dissolved.
5. **Stand** tube for 5 minutes (a blue colour will develop if phosphate is present).

Note: Make sure the colorimeter bottles are clean and dry before placing in the colorimeter.

Step 3: Reading the results

1. **Press** and hold ON button until colorimeter turns on.
2. **Press** ENTER to start.
3. **Press** ENTER to select TESTING MENU.
4. **Select** ALL TESTS from testing menu.
5. **Scroll** to and select 78 PHOSPHATE-L from menu.
6. **Clean** and **dry** the blank tube and **insert** it into colorimeter, close lid and select SCAN BLANK.
7. **Remove** blank tube from colorimeter.
8. **Clean** and **dry** the phosphate colorimeter tube with lint-free cloth or paper towel to remove all smudge marks and fingerprints.
9. **Insert** phosphate colorimeter tube into colorimeter chamber and close lid.
10. At the end of the 5 minute reaction time **select** SCAN SAMPLE.
11. **Record** the result.

The result will appear in milligrams per litre (mg/L) or parts per million (ppm). ppm = mg/L.

4.10 Measuring dissolved oxygen: Smart colorimeter method

What is dissolved oxygen?

Dissolved oxygen (DO) is the volume of oxygen that is contained in water.

Risk assessment: chemical safety

Risks: chemicals used in the tests

- **Reagent 1**: Manganous sulfate can irritate eyes and skin.
- **Reagent 2**: Alkaline potassium iodide azide can cause severe burns and is poisonous if swallowed.
- **Reagent 3**: Sulfuric acid will cause severe burns, ingestion may be fatal, and inhalation can cause coughing and chest problems.

Minimising the risks

- Always wear gloves.
- Always work in pairs (with a buddy).
- Always wear safety glasses when handling chemicals.
- Hold all tubes and bottles over the liquid waste container while adding reagents.
- Recap and return reagents to the kit after use.
- Place the liquid waste bottle, paper towels and a squirt bottle of deionised water nearby when testing.
- If any chemicals spill onto your clothes or yourself, wash them off immediately with water.

i **Safety tip: Keep the material safety data sheets (MSDS) provided in Section 5 handy for safety information on the chemicals used in this test. The first aid procedure for acid on skin is to flush it with plenty of water**

Procedure to measure dissolved oxygen

Equipment:

latex gloves

safety glasses

DO sample bottle (not the DO colorimeter tube)

DO reagent No. 1 (manganous sulfate solution)

DO reagent No. 2 (alkaline potassium iodide azide)

DO reagent No. 3 (sulfuric acid)

liquid waste container

paper towel / lint-free cloth

deionised water

DO colorimeter tube

blank colorimeter tube

Smart colorimeter, Smart 2 colorimeter or Titration kit

cooler bag

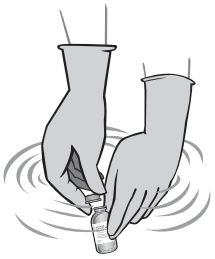
Step 1: Collecting a dissolved oxygen sample

1. Take the temperature reading (refer to temperature test instructions in Section 4.2) at the same place where the DO sample was taken, immediately after the DO sample is collected.

2. Rinse the appropriate DO bottle or tube twice with sample water and recap the sample bottle. Always pour the rinse water downstream of where the water sample is to be taken.

3. Turn the sample bottle on its side and lower it into the water until the sample bottle is fully immersed.

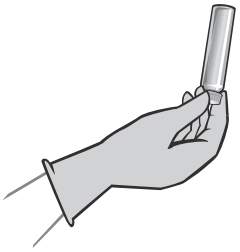
4. Unscrew the lid of the sample bottle, allowing the water to enter.



5. Turn the sample bottle upright, while still under the water, to allow it to fill completely and release all the trapped air.

6. Recap the bottle while it is immersed under the water.

7. Remove the bottle from the water and turn it upside down to check that no bubbles have been trapped inside.



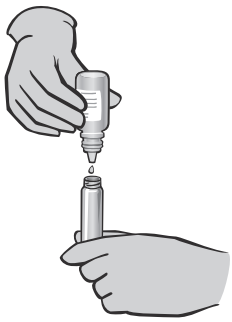
8. If bubbles are present, follow steps 3 to 7 until the bottle is filled completely and no air bubbles are present.

> ***i*** **Measurement tip: The percentage saturation of oxygen in the water can be calculated later using the dissolved oxygen results in mg/L and the temperature result.**

Step 2: Mixing the reagents

> **Note: Steps 1–6 below should be performed immediately after taking the dissolved oxygen sample.**

1. Hold the DO sample bottle which has been filled with sample water above the liquid waste container and carefully remove the lid.



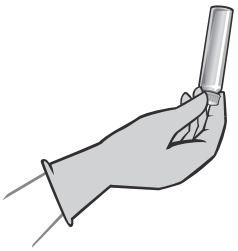
2. Add 2 drops of DO reagent No. 1 (manganous sulfate solution) to the sample water.

> ***i*** **Testing tip: Reagent bottles must be held vertically upside down or standard drops will not be delivered.**

3. Add 2 drops of DO reagent No. 2 (alkaline potassium iodide azide) to the sample water using the same method as for DO reagent No. 1.

4. Recap the DO sample bottle and wipe the bottle dry with paper towel.

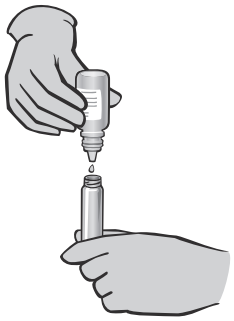
5. Invert the DO sample bottle several times to mix the solution – a yellow or brown precipitate will appear.



6. Stand the DO sample bottle and wait until the precipitate has settled to at least halfway down the bottle. This will take 5 or more minutes if the water is saline. Place the DO sample bottle in the cooler bag for preservation until you are ready to continue the test.

> ***i*** **Testing tip: The browner the solution the more oxygen is present.**

7. Hold the DO sample bottle over the liquid waste container and carefully uncap it.



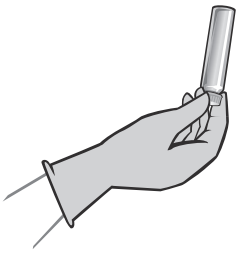
8. Add 2 drops of DO reagent No. 3 (sulfuric acid) to the sample. Recap the DO sample bottle and wipe it dry with paper towel. Invert the DO sample bottle for several minutes until the precipitate has completely dissolved.

> ***i*** **Safety tip: The sulfuric acid is very strong (1:1 solution) and can cause severe burns.**

9. Rinse the 10 mL DO colorimeter tube with treated sample over the liquid waste container and then fill to the 10 mL mark with treated sample.

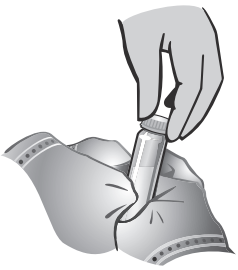
> ***i*** **Testing tip: The solution should not contain any specks, which would reduce the accuracy of your result. If there are specks in your sample, stand the bottle until they settle.**

10. Invert the DO sample bottle for several minutes to dissolve the precipitate.



11. Recap both bottles and stand on a stable surface.

12. Rinse the 10 mL blank colorimeter bottle with filtered sample water over the liquid waste container (see Section 4.9, Step 1 for filtering sample water). Fill the colorimeter tube to the 10 mL mark with filtered sample water. This is the blank.



13. Clean and dry the blank colorimeter tube with a lint-free cloth to remove all smudge marks and fingerprints.

14. Place the blank colorimeter tube into the colorimeter and cover with the black cap. Follow the relevant Smart colorimeter instructions (below).

Step 3: Reading the results

> **Measurement tip: The result will appear in milligrams per litre (mg/L) or parts per million (ppm).**
> **mg/L = ppm**

Smart colorimeter

1. Press the USE button to turn the colorimeter on.

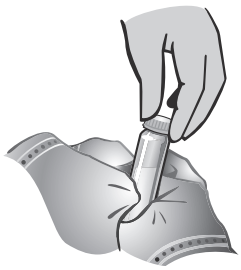
2. Scroll to and select ALL TESTS from TESTING MENU.

3. Scroll to and select 27 OXYGEN from menu.

4. Clean the blank DO colorimeter tube with paper towel to remove all smudge marks and fingerprints.

5. Insert blank colorimeter tube into colorimeter chamber, cover with black cap and select SCAN BLANK.

6. Remove blank tube from colorimeter.



7. Clean the sample DO colorimeter tube with paper towel to remove all smudge marks and fingerprints.

8. Insert DO colorimeter tube into colorimeter chamber and cover with black cap.

9. Select SCAN SAMPLE.

10. Record your result.

Smart 2 colorimeter

1. Press and hold the ON button until colorimeter turns on.

2. Press ENTER to start.

3. Press ENTER to select TESTING MENU.

4. Select ALL TESTS from testing menu.

5. Scroll to and select 39 DO from menu.

6. Clean the blank DO colorimeter tube with paper towel to remove all smudge marks and fingerprints.

7. Insert blank colorimeter tube into colorimeter, close lid and select SCAN BLANK.

8. Remove blank colorimeter tube from colorimeter.

9. Clean the sample DO colorimeter tube with paper towel to remove all smudge marks and fingerprints.

10. Insert DO colorimeter tube into colorimeter chamber and close lid.

11. Select SCAN SAMPLE.

12. Record your result.

Equipment tip:
Remember to:

- **turn the colorimeter off by pressing the OFF button (3 times for Smart and once for Smart 2)**
- **rinse colorimeter tubes twice with deionised water over the liquid waste container**
- **return all equipment to the kit after use.**

Step 4: Calculation of percentage saturation dissolved oxygen

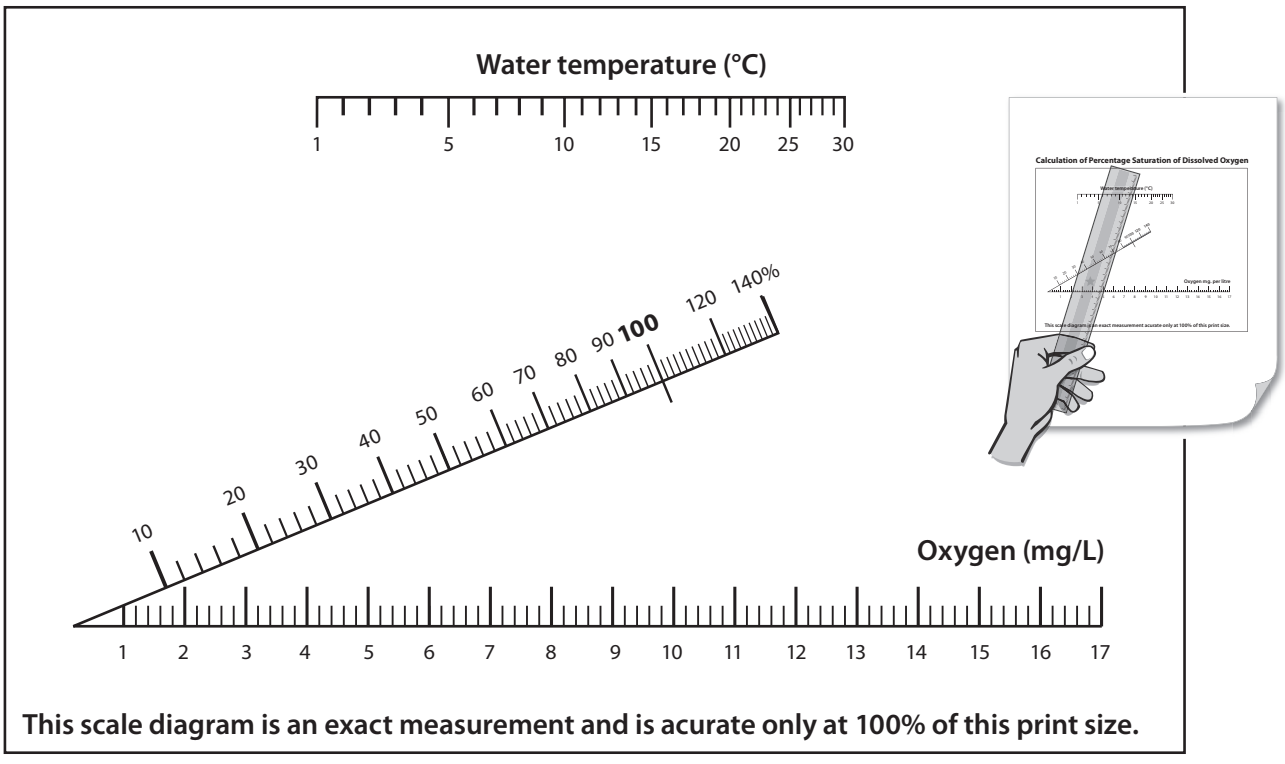
1. Plot temperature on the upper scale (water temperature °C).

2. Plot oxygen concentration on the lower scale (oxygen mg/l).

3. Hold the rule between the two points so that it crosses the middle scale as shown.

4. The point where the ruler crosses the middle scale (%) is the percentage saturation of DO.

Calculation of percentage saturation of dissolved oxygen



What do the results mean?

Dissolved oxygen

Very low – may kill organisms that rely on oxygen in the water

Less than 2 mg/L

Low – may cause stress to aquatic organisms

Less than 5–6 mg/L

% saturation

Healthy

90–110%

Poor – river health at risk

Less than 90% or greater than 110%

Note: Average for all waterways; not specific to position in the catchment.
Adapted from *ANZECC Guidelines 2000*

Summary of dissolved oxygen test: Smart and Smart 2 colorimeters

Step 1: Mixing the reagents into the tubular DO sample bottle

1. Add 2 drops of reagent 1 (manganous sulfate)
2. Add 2 drops of reagent 2 (alkaline potassium iodide azide).
3. Recap and invert several times.
4. Stand bottle and wait for precipitate to fall below shoulder of bottle.
5. Add 2 drops of reagent 3 (sulfuric acid)
6. Recap and invert bottle continuously until precipitate dissolves.

Step 3: Reading the results Smart colorimeter

1. Add 10 mL of filtered sample water to 10 mL colorimeter tube. This is the 'blank'.
2. Press USE button to turn the colorimeter on.
3. Scroll to and select ALL TESTS from TESTING MENU.
4. Scroll to and select 27 OXYGEN from menu.
5. Clean and dry colorimeter tube containing the blank to remove fingerprints.
6. Insert blank colorimeter tube into colorimeter chamber, cover with black cap and select SCAN BLANK.
7. Remove blank tube from colorimeter.
8. Clean the DO colorimeter tube with paper towel to remove all smudge marks and fingerprints.
9. Insert DO colorimeter tube into colorimeter chamber and cover with black cap.
10. Select SCAN SAMPLE.
11. Record the results in mg/L.

Step 2: Mixing the reagents into the 10 mL colorimeter tube:

1. Pour 10 mL of yellow brown iodide solution from the dissolved oxygen bottle.
2. Remove the blank from the colorimeter.
3. Insert sample bottle into the colorimeter chamber.
4. Cover tube with black plastic.
5. Press the button to SCAN sample.
6. Record the result. ppm is equivalent to mg/L.
7. Press OFF button twice to exit.

Step 3: Reading the results Smart 2 colorimeter

1. Add 10 mL of filtered sample water to 10 mL colorimeter tube. This is the 'blank'.
2. Press and hold ON button until colorimeter turns on.
3. Press ENTER to start.
4. Press ENTER to select TESTING MENU.
5. Select ALL TESTS from testing menu.
6. Scroll to and select 39 DO from menu.
7. Clean and dry colorimeter tube containing the blank to remove fingerprints.
8. Insert blank tube into colorimeter, close lid and select SCAN BLANK.
9. Remove blank tube from colorimeter.
10. Clean the DO colorimeter tube with paper towel to remove all smudge marks and fingerprints.
11. Insert DO colorimeter tube into colorimeter chamber and close lid.
12. Select SCAN SAMPLE.
13. Record the results in mg/L.

4.11 Measuring dissolved oxygen: modified Winkler titration method

This method is used for kits with the DC1200 colorimeter. The DO equipment is in a small box marked Dissolved Oxygen Titration.

What is DO?

Dissolved oxygen (DO) is the volume of oxygen that is contained in water.

Risk assessment: chemical safety

Risks: chemicals used in the tests

- **Reagent 1**: Manganous sulfate can irritate eyes and skin.
- **Reagent 2**: Alkaline potassium iodide azide can cause severe burns and is poisonous if swallowed.
- **Reagent 3**: Sulfuric acid will cause severe burns, ingestion may be fatal, and inhalation can cause coughing and chest problems.

Minimising the risks

- Always wear gloves.
- Always work in pairs (with a buddy).
- Always wear safety glasses when handling chemicals.
- Hold all tubes and bottles over the liquid waste container while adding reagents.
- Recap and return reagents to the kit after use.
- Place the liquid waste bottle, paper towels and a squirt bottle of deionised water nearby when testing.
- If any chemicals spill onto your clothes or yourself, wash them off immediately with water.

Safety tip: Keep the material safety data sheets (MSDS) provided in Section 5 handy for safety information on the chemicals used in this test. The first aid procedure for acid on skin is to flush it with plenty of water.

Procedure to measure dissolved oxygen (modified Winkler titration method)

Equipment:

latex gloves

safety glasses

DO sample bottle (not the smaller DO sample tube)

liquid waste container

DO reagent No. 1 (manganous sulfate solution)

DO reagent No. 2 (alkaline potassium iodide azide)

DO reagent No. 3 (sulfuric acid)

paper towel/lint-free cloth

20 mL vial with hole in lid and titrator syringe

sodium thiosulfate and syringe

starch indicator

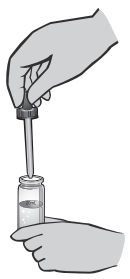
deionised water

Step 1: Collecting a dissolved oxygen sample – refer to Section 4.10, Step 1.

Step 2: Mixing the reagents

i **Testing tip: Steps 1–6 should be performed immediately after taking the dissolved oxygen sample (i.e. Step 1 above).**

1. Hold the large DO sample bottle above the liquid waste container and carefully remove the lid.



2. Add 8 drops of DO reagent No. 1 (manganous sulfate solution) to the sample water.

i **Testing tip: Reagent bottles must be held vertically upside down for standard drops.**

3. Add 8 drops of DO reagent No. 2 (alkaline potassium iodide azide) to the sample water, using the same method as reagent 1.

4. Recap the DO sample bottle and wipe the bottle dry with paper towel.

5. Invert the DO sample bottle several times to mix the solution – a yellow brown precipitate will appear.

6. Stand the DO sample bottle and wait until the precipitate has settled to at least halfway down the bottle.

> *i* **Testing tip: This will take 5 or more minutes if the water is saline.**

7. Hold the DO sample bottle over the liquid waste container and carefully uncap it.

8. Add 8 drops of DO reagent No. 3 (sulfuric acid) to the sample water.

> *i* **Safety tip: The sulfuric acid is very strong (1:1 solution) and can cause severe burns.**

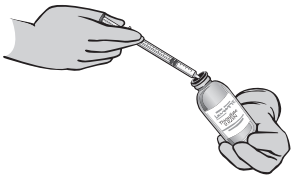
9. Recap the DO sample bottle and wipe the bottle dry with paper towel. Invert the DO sample bottle for several minutes until the precipitate has completely dissolved.

> *i* **Testing tip: All brown flakes must be dissolved. If the water has a high DO level this may take several minutes. If brown flakes remain after 5 minutes, add 4 further drops of sulfuric acid and continue mixing. If brown flakes still remain, let the bottle stand to settle the precipitate, then pour off the clear coloured solution.**

Step 3: Titration

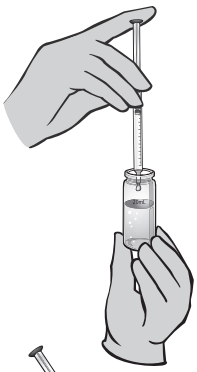
1. Hold the 20 mL vial over the liquid waste container and fill to the 20 mL mark with treated sample. Recap the vial.

2. Remove the sodium thiosulfate and titrator syringe from the kit.



3. Draw the plunger back halfway and insert the tip of the titrator syringe into the small hole in the top of the sodium thiosulfate bottle. Push the plunger in to expel the air into the bottle. This avoids a vacuum being created in the bottle as the liquid is withdrawn.

4. Turn the bottle and titrator syringe upside down and while supporting both, slowly pull back on the plunger until the black stopper is aligned with the zero line. If bubbles form on the stopper, push the plunger in and redraw sodium thiosulfate. This may have to be done several times to eliminate the bubbles.

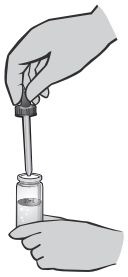


5. Turn the bottle upright and carefully remove the titrator syringe by pulling from its base. Insert the titrator syringe into the hole in the lid of the 20 mL vial.

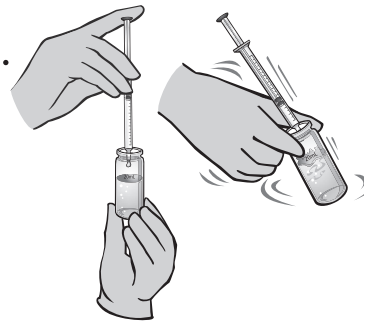
6. Add one drop of sodium thiosulfate at a time by depressing the plunger and gently swirling between each drop.

7. Continue adding single drops of sodium thiosulfate, swirling a few times in between each drop, until the liquid changes to straw colour (pale yellow).

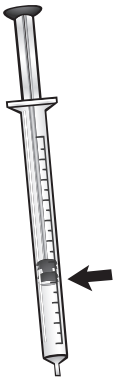
8. Remove the cap from the titrator leaving the syringe in place and add 8 drops of starch indicator to the liquid in the vial. The solution will turn deep blue. The starch indicator is added to make the titration endpoint easier to see. Recap the vial.



9. Continue adding drops of thiosulfate, swirling between each drop, until the blue turns from blue to clear with one drop. This is the endpoint of the reaction.



10. Record the number of units of sodium thiosulfate used from the titration syringe. This is equivalent to the mg/L of DO in the sample water.



11. Calculate the percentage saturation of DO by following the instructions given in Step 4 and record this result.

i **Testing tip: Occasionally the sample may require more than 10 units of sodium thiosulfate to reach the endpoint.**

Before refilling the titrator syringe, rinse the tip with deionised water and dry it with paper towel.

Five units of sodium thiosulfate will be more than is required to finish the titration.

i **Equipment tip:**
Remember to:

- **rinse the vial and its cap twice with deionised water over the liquid waste container**
- **clean the titrator syringe**
- **return all equipment to the kit after use.**

Cleaning the titrator syringe

1. Pour a small amount of deionised water into the small beaker.

2. Draw water into the syringe and expel into the beaker twice.

3. Dry the outside of the syringe with paper towel.

4. Rinse the beaker twice with deionised water over the liquid waste container.

Step 4: Calculation of percentage saturation of DO

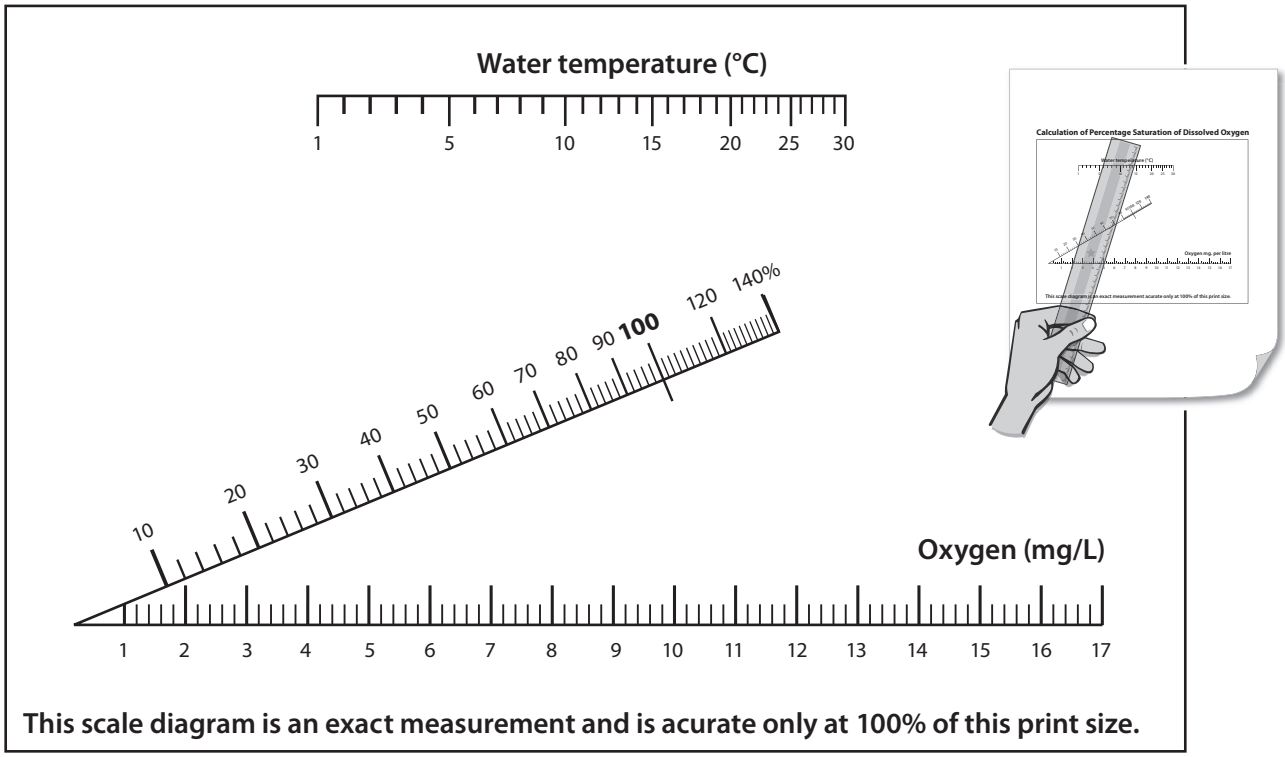
1. Plot temperature on the upper scale (water temperature °C).

2. Plot oxygen concentration on the lower scale (oxygen mg/L).

3. Hold the ruler between the two points.

4. The point where the ruler crosses the middle scale (%) is the percentage saturation of DO.

Calculation of percentage saturation of dissolved oxygen



This scale diagram is an exact measurement and is acurate only at 100% of this print size.

What do the results mean?

Dissolved oxygen

Very low – may kill organisms that rely on oxygen in the water

Less than 2 mg/L

Low – may cause stress to aquatic organisms

Less than 5–6 mg/L

% saturation

Healthy

90–110%

Poor – river health at risk

Less than 90% or greater than 110%

Note: Average for all waterways; not specific to position in the catchment.
Adapted from *ANZECC Guidelines 2000*

4.12 Measuring E. coli

Measuring E. *coli* provides an alternative measure of faecal contamination. The method is quick and relatively inexpensive and can be conducted with a high degree of accuracy by Waterwatch groups. This method provides an early warning of faecal contamination and if colonies exceed the limit for primary contact, groups may wish to undertake further tests using the filtration method, or send samples to a laboratory.

Note: this test is optional.

What is E. coli ?

E. *coli* is the abbreviated name of the bacterium *Escherichia* (genus) *coli* (species).

E. *coli* is the primary bacterium in the intestinal tract of warm-blooded animals. This makes it a good indicator of faecal contamination.

Why test faecal bacteria?

Faecal coliforms are used as an indicator for assessing risk to human health.

E. *coli* is the most common bacterium present in the intestinal tract and faeces of mammals, birds and humans. Its presence in freshwater environments may be due to wastewater contamination, runoff from agriculture, waterbird and livestock defecation and stormwater contamination.

Risk assessment

Risks identified

- E. *coli* may be harmful.

Minimising the risks

- Always wear gloves.
- Always work in pairs (with a buddy).
- Ensure gel is set before placing in incubator to limit spillage.
- Dispose of used Petrifilm E. *coli* plates in a biological hazard waste container or wrap in a plastic bag before disposal.

Safety tip: Petrifilm is not considered to be hazardous. It does not result in exposure to hazardous chemicals under normal conditions. A material safety data sheet (MSDS) is available from the manufacturer if required.

Collecting an E. coli sample

To minimise the chance of contaminating the sample, take your sample for E. *coli* testing before collecting sample water for other tests. You will need a sterile Schott bottle to collect your E. *coli* sample.

Step 1: Sterilisation

As this test is a microbiological test, the bottles must be sterilised before each use. They can be sterilised by one of two methods:

- boiling for 20 minutes
- autoclaving (preferred method).

To sterilise equipment by boiling fill a large clean pot with water and bring to boil on the stove. Place equipment in boiling water for at least 20 minutes.

Step 2: Collecting the sample

> ***i*** **Testing tip: Do not rinse or remove the lid of the sample bottle before doing this test as this may contaminate the sterile bottle.**

1. Turn the sample bottle on its side and lower it into the water until it is fully immersed. The lid on the bottle should not be removed until it is below the surface of the water to prevent surface scum entering the bottle.

2. Slowly unscrew the lid, allowing water to enter. Fill the bottle to three-quarters full so that there is an air gap. The air gap will let you mix your sample when shaking before doing the test later.

3. Recap the bottle while it is still underwater and remove from the water.

4. Place the sample into the cooler bag for preservation until you are ready to start the test.

> ***i*** **Testing tip: The sample should be tested and incubated as soon as possible and within 24 hours of collection.**

The Petrifilm method

The Petrifilm method has been specifically designed for volunteer water monitoring purposes. The method is user friendly, relatively inexpensive and easily quantifies and identifies total coliforms and E. *coli*.

Equipment:

gloves

sterile Schott bottle or pre-sterilised disposable container

Petrifilm E. *coli* plates

disposable sterile plastic pipette or syringe

small glass (heat resistant) beaker

incubator

Test preparation

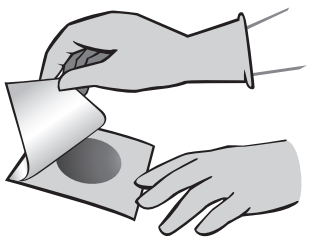
- Store unused Petrifilm E. *coli* plates in a sealed plastic container in the freezer.
- Collect your sample in a sterile Schott bottle or sterile disposable container.
- Turn on the incubator and ensure it has reached 44°C ± 1°C.
- Perform the test on a flat surface.

Method

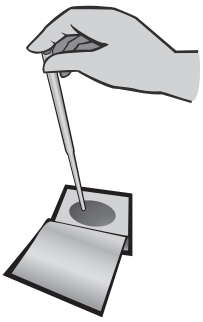
1. Shake sample bottle vigorously.

2. Using 1 mL disposable plastic pipette or sterile syringe, draw up 1 mL of sample water.

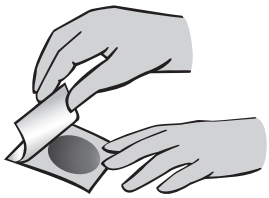
3. Lift up the top clear film of the Petrifilm E. *coli* plate.



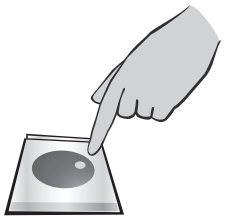
4. Hold pipette upright and dispense the 1 mL sample onto the centre (pink circle) of the bottom film.



5. Roll the top film back onto the sample quickly and gently to minimise bubble formation.



6. Allow the gel to set.



7. Place in an incubator clear side up.

Setting the incubator

1. The incubator needs to be turned on and brought up to 44°C ±1°C.

2. A small beaker of water should be placed into the incubator to act as a humidifier.

3. It is important that the incubator maintains this temperature for 24 hours ±2hr as fluctuations in temperature, or temperatures outside of this range, may inhibit the growth of E.*coli*.

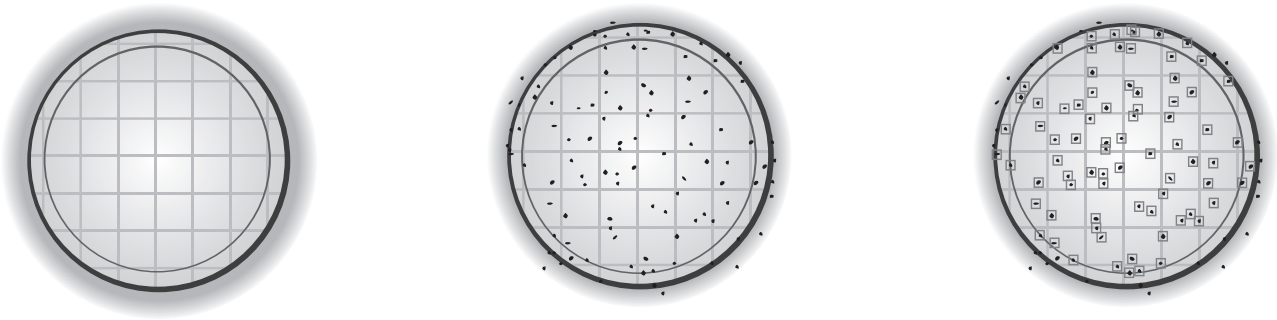
Interpreting your results

The Petrifilm E. *coli* plates contain violet red bile (VRB) nutrients and glucuronidase indicators to stain E. *coli* colonies blue. Most E. *coli* colonies will also produce gas or bubbles.

Coliform colonies produce acids, which activate the pH indicators in the Petrifilm E. *coli* plate medium and will appear red.

Do not include colonies that grow on the foam barrier.

> ***i*** **Measurement tip: Blue colonies + gas = E. *coli***
>
> **Red colonies and blue colonies + gas = total faecal coliforms**



Recording your results

E. *coli* counts are reported as colony forming units per 100 mL of water (CFU/100 mL).

Petrifilm E. *coli* plates can only accommodate a sample volume of 1mL. This means that the resulting number of colonies needs to be multiplied by 100.

> ***i*** **Measurement tip: E.g. 11 E. *coli* (blue) colonies on a Petrifilm E. *coli* plate = 11 x 100 = 1100 CFU/100 mL**
>
> **E. *coli* (blue + red colours) x 100 = CFU/100 mL**

What do the results mean?

E. *coli* provides a measure of faecal contamination. Compare your results to the ANZECC guidelines for faecal coliforms.

E. *coli* as an indicator of faecal contamination
Stressor: increase to indicator (+)
Effect: loss of native plants and animals, health risks
Trigger value: all parts of the catchment **Primary contact:** <150 CFU/100 mL (direct contact, e.g. swimming) **Secondary contact:** <1000 CFU/100 mL (indirect contact, e.g. boating)

4.13 Measuring faecal coliforms

What are faecal coliforms?

Faecal coliforms are naturally occurring bacteria found in the intestines of all warm-blooded animals (including humans) and birds.

Risk assessment

Risks identified

- Sterilising equipment: risk of burns.
- Flaming forceps: risk of burns.
- Faecal colonies may be harmful.

Minimising the risks

- Always wear gloves. One exception to this is that when flaming forceps, gloves should not be warn, as they may easily catch alight or melt.
- Always work in pairs (with a buddy).

- Take care when sterilising equipment – wait until equipment has cooled before handling.
- Take care when flaming forceps and ensure that the alcohol bottle is covered before lighting forceps.

- Take care when connecting the pipettes to the pump as pipettes are easily broken.
- Dispose of used petri dishes in the biological hazard waste or alternatively boil before disposal in the bin.

Collecting a faecal coliform sample

Refer to the procedure for collecting an E. *coli* sample in Section 4.12.

Faecal coliform test

Equipment:

latex gloves

aluminium foil

petri dish with absorbent pad

mFC broth ampoule/s

permanent marker pen

filter paper

liquid waste container

alcohol

forceps

long-handled lighter

0.45 micron filter paper

sterile Schott bottle (faecal coliform sample bottle)

peptone water (PW)

sterile filtration unit (top filter chamber, filter support plate, bottom receiving chamber)

three-way valve

syringe

pipette pump/s

sterile pipettes

paper towel

incubator

Step 1: Sterilisation

As this test is a microbiological test, equipment must be sterilised before each use.

All equipment should be pulled apart (e.g. remove lids, etc.) before sterilising. The following equipment needs to be sterilised each time:

- top filtration chamber/s
- filtration plate/s
- Schott bottle/s (faecal coliform sample bottle)
- pipettes (0.1 mL, 1 mL and 10 mL).

The equipment can be sterilised by one of two methods:

- boiling for 20 minutes
- autoclaving (preferred method).

To sterilise equipment by boiling fill a large clean pot with water and bring to boil on the stove. Place equipment in boiling water for at least 20 minutes.

> ***i*** **Testing tip: Pipettes may be too long to fit in a pot. If this is the case place them in the boiling water for 10 minutes then carefully remove and place the other end of the pipettes in the water for a further 10 minutes.**

Wrap pipettes and the filtration chamber and plate in aluminium foil to keep them sterile.

Schott bottles should have their lids fastened and be labelled or marked as sterile.

Step 2: Setting the incubator

The incubator needs to be turned on and brought up to 44.5°C ±0.2°C before the filtering process begins. It is important that the incubator maintains this temperature for 24 hours as fluctuations in temperature, or temperatures outside of this range, may inhibit the growth of faecal coliforms. Incubation of petri dishes at this temperature inhibits the growth of background bacteria.

Step 3: Selecting the sample dilution

Faecal coliforms are measured by the number of colony-forming units (CFU) or colonies per 100 mL of water. A 1 mL volume of sample should always be filtered. A 1 mL volume will allow sufficient growth to indicate a situation which may require reporting to your regional coordinator. To calculate your result per 100 mL the number of colonies counted on your 1 mL plate should be multiplied by 100.

If you suspect the water you are sampling is very clean, 100 mL can be filtered. Below is a guide to the dilutions you should choose in addition to your 1 mL.

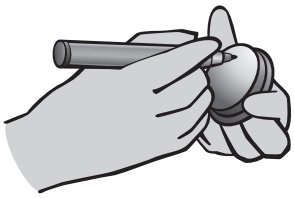
Faecal coliforms are reported per 100 mL of water so a result of zero (0 CFU/100 mL) will only be obtained if 100 mL of sample has been filtered. The table below shows how to record your results when no colonies are grown on the plate at various dilutions.

Testing tip: **An ideal sample will produce 20 to 80 faecal coliform colonies and not more than 200 colonies on a petri dish. You should try to estimate the likelihood of high numbers of faecal coliforms and pick a dilution that is likely to produce 20–80 colonies. You may need to try a few dilutions to increase the chance of finding the correct dilution. A rough guide for estimating which dilutions to try is:**

- **sewer overflow suspected or pollution visible (>10 000 CFU/100 mL): filter 0.1 mL**
- **after rain or some pollution suspected (>1000 CFU/100 mL): filter 10 mL**
- **faecal coliforms likely in low numbers (<1000 CFU/100 mL): filter 100 mL**

Volume filtered	Colonies counted on plate	Results should be recorded as:
0.1 mL	0	<1000 CFU/100 mL
1.0 mL	0	<100 CFU/100 mL
10 mL	0	<10 CFU/100 mL
100 mL	0	0 CFU/100 mL

Step 4: Preparing the petri dishes



1. Select the required number of petri dishes with absorbent pads and on the bottom of each, write:
- the volume of sample filtered (i.e. 0.1 mL, 1 mL, 10 mL or 100 mL)
- site name
- date
- time incubation is to start
- group name or number.

2. Invert an ampoule containing mFC broth several times to mix. Twist the cap of the ampoule, remove the lid from a petri dish and pour the entire 2 mL of mFC broth evenly over the absorbent pad. Replace the petri dish lid immediately.

3. Repeat for other petri dish/es.

Step 5: Setting up the filtration unit

1. Place the bottom receiving chamber on a stable, level surface and screw in the sterile top filter chamber with attached filter support plate.

> ***i*** **Equipment tip: Make sure that the vents are open on the top chamber, but closed with plastic caps on both the bottom receiving chamber and one of the sides on the filter support plate.**

2. Attach the three-way valve to the vent that is not capped on the filter support plate and then attach the syringe to the other end.

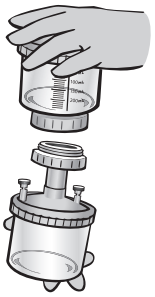
3. Remove the lid from the alcohol bottle and dip the tips of the forceps into the alcohol. Replace the lid of the alcohol bottle before the long-handled lighter is ignited.

> ***i*** **Equipment tip: The top filter chamber may be used to filter different volumes of the same sample. However, always filter the smallest volume first.**

4. Hold the forceps down and angled away from you and pass the tips fairly quickly through the top of the flame of the long-handled lighter about 7 or 8 times.

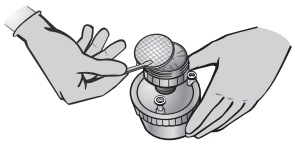
> ***i*** **Safety tip: Gloves should not be worn when flaming forceps as they may catch alight or melt. Keep your fingers away from the flame.**

5. Unscrew the top filter chamber from the filter support plate and add a few drops of peptone water to the plate.



6. Use the sterile forceps to remove the filter paper from its packet.

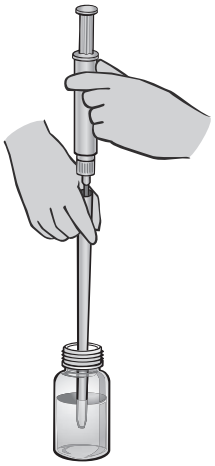
7. Place the filter paper on top of the filter support plate with the grid facing up.



8. Screw the top chamber back onto the filter support plate.

> ***i*** **Testing tip: Shake the faecal coliform sample bottle well by inverting several times before drawing off or pouring the desired volume.**

Using pipettes



The green pipette pump is for the 10 mL pipette.

The blue pump is for the 1 mL and 0.1 mL pipettes.

To filter 100 mL, pour sample directly into the top filter chamber using the graduated scale on the chamber to measure the volume. To draw up sample water using the pipette, roll the small wheel at the top of the pipette pump with your thumb. Be careful not to draw sample water up into the pipette pump. To release sample water from the pipette press the long button.

> ***i*** **Testing tip: It is good practice to filter at least two different sample volumes. Always filter the smallest volume first to prevent contamination of the equipment by the larger sample. Replace the lid on the sample bottle and re-shake the bottle before taking up each sample.**

9. After selecting the desired volume, remove the lid from the top filter paper and then follow the appropriate instructions:

For 0.1 mL, 1 mL and 10 mL volumes:

- Draw up the required amount of sample water in the pipette and expel it into the sterile peptone water (PW).
- Recap the peptone water bottle and shake before pouring the water (which now contains sample water) evenly onto the filter paper in the filter chamber.

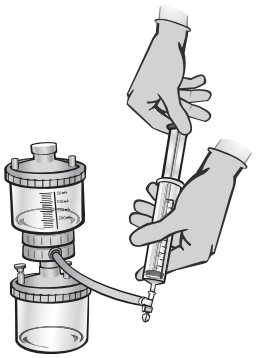
For a 100 mL volume:

- Pour sample water into the top filtration chamber up to the 100 mL mark.
- Pour a vial of peptone water onto the filter paper in the chamber to rinse.

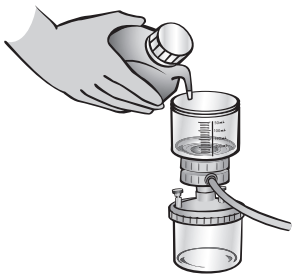
Step 6: Filtering the sample

1. Replace the lid on the top filter chamber.

2. Draw back and release the syringe several times to create a vacuum in the lower chamber. This will draw the water from the top chamber through the filter paper and into the bottom chamber. If the water does not start draining through after a few pumps, check that all sections are screwed together tightly and are sealed.



3. Remove the lid from the top filter chamber. Pour a vial of peptone water into the top chamber to rinse any remaining faecal coliforms from the inside of the chamber.

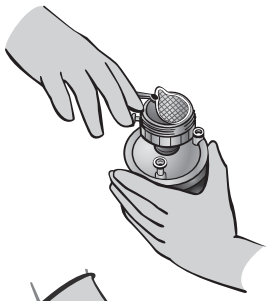


4. Replace the lid and draw this water through the filter paper using the syringe pump.

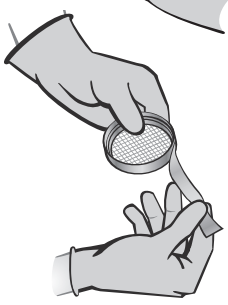
5. Remove one of the plastic caps on the bottom receiving chamber to release the vacuum and remove the syringe pump.

6. Dip the forceps in the alcohol again and flame them with the long-handled lighter following the same procedure used previously.

7. Remove the top filter chamber from the filter support plate and using the sterile forceps, carefully lift and take hold of the edge of the filter paper.



8. Transfer the filter paper onto the absorbent pad in the petri dish using a rolling motion to ensure that no air bubbles form. Replace the lid on the petri dish immediately.



i **Testing tip: The filter paper grid should be placed face up on the absorbent pad, making sure they are superimposed (in total contact).**

9. Place the petri dish/es upside down (grid facing down) in the incubator as soon as possible after filtering.

10. Incubate the petri dish/es for 24 hours ±2 hours at 44.5°C ±0.2°C.

11. After 24 hours remove the petri dish/es from the incubator and count the blue colonies within 20 minutes.

Step 7: Counting faecal coliform colonies

If more than one volume has been filtered, select the petri dish which has between 20 and 80 faecal coliform colonies present.

1. Colonies are seen as blue dots on the filter paper.

2. In some instances faecal coliform colonies may appear pink or red rather than blue on the mFC media. Count pink colonies as faecal coliforms but note that colonies were seen as pink.

3. Ignore any other colours such as cream or grey – they are not faecal coliforms.

4. Ignore any very tiny (pin point or smaller) blue dots as these are not viable organisms.

5. Use the grid pattern on the filter paper as an aid to counting the colonies by counting along the grid lines.

Step 8: Recording your results

> ***i*** **Measurement tip: Faecal coliforms are reported as colony forming units per 100 mL of water (CFU/100 mL)**

1. If 0.1 mL of sample water was filtered, multiply the resulting number of faecal coliform colonies by 1000. For example, 21 colonies on a 0.1 mL plate: 21 x 1000 = 21 000 CFU/100 mL.

2. If 1 mL of sample water was filtered, multiply the resulting number of colonies by100. For example, 21 colonies on a 1 mL plate: 21 x 100 = 2100 CFU/100 mL.

3. If 10 mL of sample water was filtered, multiply the resulting number of colonies by 10. For example, 21 colonies on a 10 mL plate: 21 x 10 = 210 CFU/100 mL.

4. If 100 mL of sample water was filtered, you do not need to multiply the resulting number of colonies at all. For example, 21 colonies on a 100 mL plate: 21 CFU/100 mL.

5. If more than one volume of sample water has been tested from the one sample bottle, do not average the results. Select the petri dish which has between 20 and 80 faecal coliform colonies present to determine the result.

6. If the filter paper is covered in colonies that have grown into each other or there are too many to count, record the result as TNTC – 'too numerous to count'.

> ***i*** **Equipment tip:**
> **Remember to:**
> - **Always sterilise equipment after use.**
> - **Dispose of used petri dishes in the biological hazard waste or boil petri dishes before placing in the bin.**
> - **Return all equipment to the kit after use.**

What do the results mean?

Faecal coliforms
Stressor: increase to indicator (+)
Effect: loss of native plants and animals, health risks
Trigger value: all parts of the catchment **Primary contact:** <150 CFU/100 mL **Secondary contact:** <1000 CFU/100 mL

SECTION 5

Material safety data sheets

Some of the chemicals used in Waterwatch water quality testing can be dangerous if correct procedures are not followed. The material safety data sheets (MSDS) provided in this section contain specific information about the chemicals supplied in your Waterwatch kit, including first aid and clean-up procedures in the event of an accident.

Included in this section:

5.1 Summary of MSDS sheet: available phosphate test

Phosphate acid reagent V-6282

Hazardous ingredients

Sulfuric acid [CAS# 7664-93-9]; ammonium molybdate [CAS# 12054-85-2]; antimony potassium tartrate [CAS# 28300-74-5].

Physical

Clear colourless liquid; soluble in water; slight odour.

Fire and explosion

Not a fire hazard; reacts exothermically and violently with water; reacts with metals to form flammable, explosive hydrogen gas.

Reactivity

Stable; avoid contact with water, metals, organic or combustible materials, and strong bases; decomposes to sulfur oxide (SOx) and hydrogen gas.

Health hazard

Toxic; primary routes of entry are skin and ingestion; corrosive to all body parts; ingestion may be fatal; inhalation causes coughing, chest pains, damage to lungs; not carcinogenic.

First aid

Eye contact

Immediately flush eyes with water for 15 minutes, get immediate medical attention.

Ingestion

Do not induce vomiting, rinse out mouth immediately, drink lots of water, call a physician immediately.

Inhalation

Remove to fresh air, give artificial respiration if needed, or give oxygen if breathing is difficult.

Skin contact

Immediately flush with water while removing affected clothing, rinse skin thoroughly for 15 minutes, consult a physician.

Spills and disposal

Wear gloves and eye protection; cover spills with sodium bicarbonate or soda ash/calcium carbonate mixture; carefully add water to form a slurry, avoiding heat, spattering and fumes; scoop up, add water and flush to drain. Dispose by stirring very slowly into a large volume of soda ash/slaked lime, pour neutralised solution plus water down drain; dispose according to local regulations.

Precautions

Handling

Use gloves, eye protection and a lab coat.

Hygiene

Avoid inhalation and contact with skin and clothing.

Storage

Store away from alkalis.

Phosphate reducing reagent V-6283

Hazardous ingredients

D(-)-isoascorbic acid [CAS# 89-65-6]; sucrose [CAS# 57-50-01].

Physical

White powder; soluble in water; no odour.

Fire and explosion

Extinguish with water spray; reacts with chemically active metals (Zn, Al, Mg, Cu) to form flammable, explosive hydrogen gas.

Reactivity

Stable; avoid heat and moisture; incompatible with strong oxidisers (nitric or sulfuric acid) and metals; decomposes to carbon oxides (COx).

Health hazard

Not toxic; primary routes of entry are skin, inhalation and ingestion; dust may irritate eyes, skin and respiratory tract; not carcinogenic.

First aid

Eye contact

Flush with water for 15 minutes.

Ingestion

Rinse mouth, drink plenty of water.

Inhalation

Remove to fresh air.

Skin contact

Rinse skin, wash with soap and water.

Spills and disposal

Sweep up (careful not to raise and breathe dust), dissolve in water and flush down drain with water. Dispose by dissolving in water, flushing down drain with water; dispose according to local regulations.

Precautions

Handling

Use gloves, eye protection and a lab coat.

Hygiene

Avoid inhalation and contact with skin.

5.2 Summary of MSDS sheet: dissolved oxygen test: colorimetric method

DO reagent No. 1 (manganous sulfate solution) 4167

Hazardous ingredients

Manganese sulfate monohydrate [CAS# 10034-96-5].

Physical

Clear pink liquid; soluble in water; no odour.

Fire and explosion

Not a fire hazard.

Reactivity

Stable.

Health hazard

Toxicity unknown (manganese could be a tumorigen, mutagen reproductive effector); primary routes of entry are ingestion and skin; harmful if swallowed; contact with skin or eyes causes irritation; not carcinogenic.

First aid

Eye contact

Flush eyes with water for 15 minutes, consult a physician.

Ingestion

Induce vomiting immediately, consult a physician.

Skin contact

Flush thoroughly with water, remove affected clothing, wash skin with soap and water, consult a physician.

Spills and disposal

Mop up spills carefully and hold for disposal.

Dispose small quantities by flushing with water down drain; dispose large quantities in containers according to regulations.

Precautions

Handling

Use gloves, eye protection and a lab coat.

Hygiene

Wash after handling.

DO reagent No. 2
(alkaline potassium iodide azide) 7166

Hazardous ingredients

Potassium hydroxide [CAS# 1310-58-3]; sodium azide [CAS# 26628-22-8]; potassium iodide [CAS# 7681-11-0]

Physical

Clear colourless liquid; soluble in water; no odour.

Fire and explosion

Not a fire hazard; can react with metals to produce explosive hydrogen/air mixture; violent exothermic reaction occurs with water (possibly hot enough to ignite combustibles).

Reactivity

Stable; avoid heat; avoid contact with strong acids and metals; decomposes to hazardous hydrogen gas.

Health hazard

Toxic; primary route of entry is the skin; causes severe burns; may be fatal if swallowed; not carcinogenic.

First aid

Eye contact

Immediately flush eyes with water for 15 minutes, get immediate medical attention.

Ingestion

Do not induce vomiting, rinse mouth immediately, drink lots of water, call a physician immediately.

Skin contact

Immediately flush with water while removing affected clothing, rinse skin thoroughly for 15 minutes, consult a physician.

Spills and disposal

Neutralise by slowly adding hydrochloric acid (6M or less) to pH 7. Flush small amounts (<25 mL) of neutralised waste with water to drain; dispose of large amounts as hazardous waste according to regulations (sodium azide reacts with metal to form explosive metal azides).

Precautions

Handling

Use gloves, eye protection and a lab coat.

Hygiene

Avoid contact with skin and clothing.

Storage

Store away from acids and metals.

DO reagent No. 3
(sulfuric acid 1:1) 6141

Hazardous ingredients

Sulfuric acid [CAS# 7664-93-9]

Physical

Colourless liquid; soluble in water; no odour.

Fire and explosion

Flammable; extinguish using dry chemical or CO_2, not water; wear breathing apparatus and protective equipment; reacts exothermically and violently with water; reacts with metals to form flammable, explosive hydrogen gas.

Reactivity

Stable; avoid contact with water, metals, organic or combustible materials, and strong bases; decomposes to sulfur oxides (SOx) and hydrogen gas.

Health hazard

Toxic; primary routes of entry are skin, inhalation and ingestion; corrosive to all body parts; ingestion may be fatal; inhalation causes coughing, chest pains, damage to lungs; not carcinogenic.

First aid

Eye contact

Immediately flush eyes with water for 15 minutes, and get immediate medical attention.

Ingestion

Do not induce vomiting, rinse mouth immediately, drink lots of water and call a physician immediately.

Inhalation

Remove to fresh air, give artificial respiration if needed, or give oxygen if breathing is difficult.

Skin contact

Immediately flush with water while removing affected clothing, rinse skin thoroughly for 15 minutes, and consult a physician.

Spills & disposal

Wear gloves and eye protection; cover spills with sodium bicarbonate or soda ash/calcium carbonate mixture; mix and carefully add water to form a slurry, avoiding heat, spattering, fumes; scoop up, add water and flush to drain. Dispose by stirring very slowly into a large volume of soda ash and calcium hydroxide, pour neutralised solution plus water down drain; dispose according to local regulations.

Precautions

Handling

Use gloves, eye protection and a lab coat.

Hygiene

Avoid inhalation and contact with skin and clothing.

Storage

Store away from bases, metal powders, combustible materials.

5.3 Summary of MSDS sheet: dissolved oxygen test: modified Winkler titration method

Sodium thiosulfate 4169

Hazardous ingredients

Sodium hydroxide [CAS# 1310-73-2].

Physical

Clear colourless liquid; no odour.

Fire and explosion

Not a fire hazard.

Reactivity

Stable; avoid heat and light.

Health hazard

Not toxic; primary routes of entry are skin and ingestion; not carcinogenic; can cause skin and (if taken in large doses by mouth) gastrointestinal irritation.

First aid

Eye contact

Flush with water for 15 minutes.

Ingestion

Drink plenty of water, consult a physician.

Skin contact

Rinse skin, wash with soap and water.

Spills and disposal

Neutralise with vinegar or other dilute acid and mop up. Dispose after neutralizing by flushing down drain; dispose according to regulations.

Precautions

Handling

Use gloves, eye protection and a lab coat.

Hygiene

Avoid inhalation and contact with skin.

Storage

Store away from heat and light.

Starch indicator 4170

Hazardous ingredients

Salicylic acid [CAS# 69-72-7].

Physical

Colourless liquid; soluble in water; no odour.

Fire and explosion

Not a fire hazard.

Reactivity

Stable; avoid heat and light.

Health hazard

Toxic (salicylic acid investigated as a possible mutagen); primary route of entry is ingestion; not carcinogenic; may be harmful if swallowed.

First aid

Eye contact

Flush with water.

Ingestion

Drink water or milk, consult a physician.

Skin contact

Flush with water.

Spills and disposal

Mop up, flush down drain. Dispose by flushing with water down drain; dispose according to local regulations.

Precautions

Handling

Use eye protection.

Hygiene

Avoid inhalation and contact with skin.

Storage

Store at room temperature out of direct light.

SECTION 6

Interpreting your results

The Australian and New Zealand Environment Conservation Council (ANZECC) has developed guidelines for classifying the quality of water in rivers, lakes, estuaries and marine waters. Waterwatch has also developed guidelines for linking water quality parameters to the health of ecosystems.

Students and community members can apply the ANZECC and Waterwatch guidelines to the results of the water quality tests they conduct at their site and come up with an assessment of the health of their catchment. In doing so they will learn about the concept of trigger values and how they can help identify potential environmental problems.

Recording and interpreting the results of your water quality tests is made easy with the recording sheets provided. Careful use of these recording sheets will guarantee that all the information is recorded at the site and is ready to upload to the Waterwatch online database at www.waterwatch.nsw.gov.au

Included in this section: *Page*

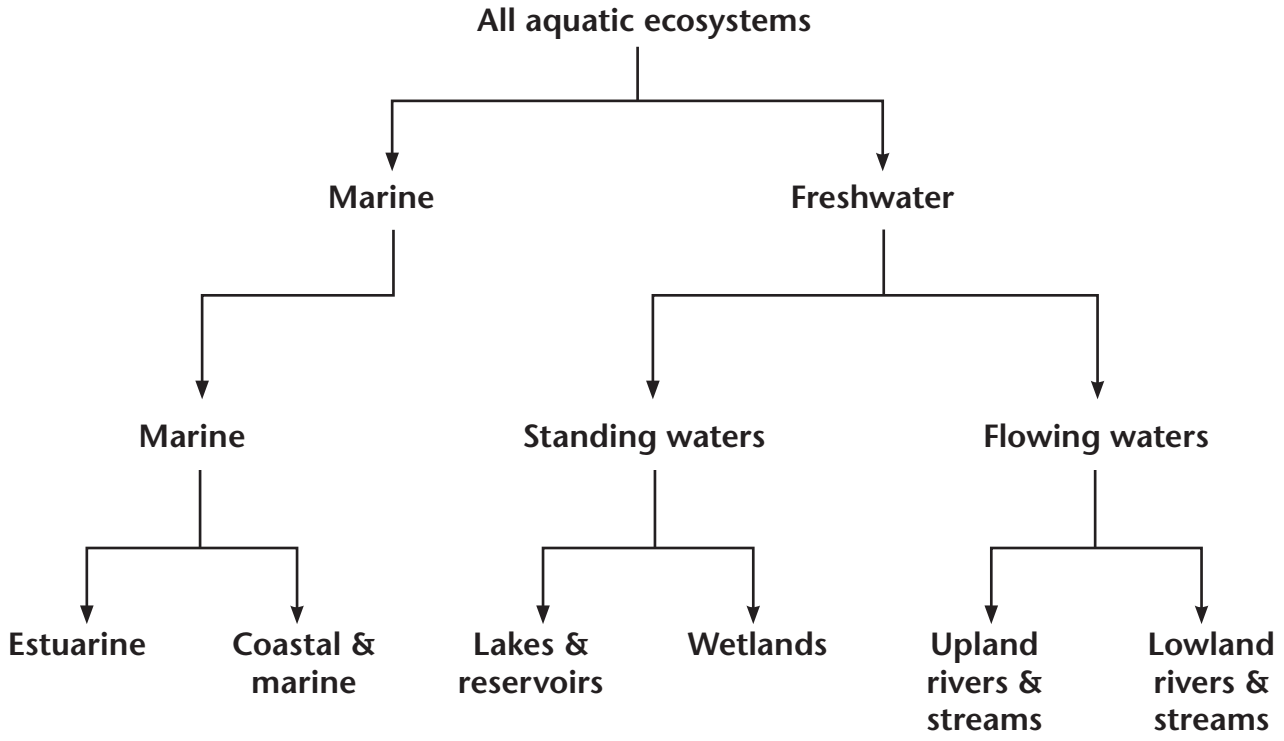
6.1 ANZECC water quality guidelines

In 2000, the Australian and New Zealand Environment Conservation Council (ANZECC) released its water quality guidelines for rivers, lakes, estuaries and marine waters. It is important to know the height above sea level of your location so that your waterway can be classified according to the ANZECC guidelines.

Measurement tip:
Upland stream: Above 150 metres above sea level
Lowland stream or coastal stream: Below 150 metres above sea level
Estuary/marine

Aquatic ecosystems

There are different types of aquatic ecosystems. These include:



Adapted from *ANZECC Guidelines 2000*

Water quality stressors

Changes in water quality may put pressure on an ecosystem. They may be due to either increases or decreases in the various water quality parameters. For example, an increase in salinity (EC or TDS) may cause stress on an ecosystem while any change in temperature may affect the same ecosystem. Such changes are called water quality stressors.

Water quality guidelines

A water quality guideline is a recommended value or range for a given parameter. Water quality guidelines help to identify when changes in a water quality parameter have the potential to cause an environmental problem. A significant departure from a guideline may trigger further investigation and thus is called a trigger value.

Waterwatch groups collect data at sites which may not be monitored by any other group or organisation. This information helps to develop guidelines for water quality and trigger values at their site.

The *ANZECC Guidelines 2000* (water quality) identified trigger values for water quality based on the location within a catchment.

Trigger value guidelines can:

- provide information that helps to identify potential environmental problems
- assist with management of key environmental issues
- assess the impact of management actions.

Note that **primary contact** includes activities that require complete submersion such as swimming, diving etc., while **secondary contact** includes activities where you come into contact with water but are not completely immersed, e.g. boating, fishing.

Trigger values for upland rivers

Temperature (°C)	Turbidity (NTU)	Electrical conductivity (µS/cm or mS/cm)	pH	Available phosphate (mg/L)	Dissolved oxygen (% saturat.)
Stressor: change to indicator (+ or –)	**Stressor**: increase to indicator (+)	**Stressor**: increase to indicator (+)	**Stressor**: change to indicator (+ or –)	**Stressor**: increase to indicator (+)	**Stressor**: change to indicator (+ or –)
Effect: loss of native plants and animals	**Effect**: loss of native plants and animals	**Effect**: loss of native plants and animals	**Effect**: loss of native plants and animals	**Effect**: nuisance plant growth	**Effect**: loss of native plants and animals (fish kills)
Trigger value: 15–35°C for prolonged exposure	**Trigger value**: 25 NTUs	**Trigger value**: 350 µS/cm or 0.35 mS/cm	**Trigger value**: 6.5–8.0	**Trigger value**: 0.05 mg/L (this figure represents 3.06 times the filterable reactive phosphate calculated within the ANZECC guidelines)	**Trigger value**: 90–110%

Adapted from *ANZECC Guidelines 2000* for NSW ecosystems

Trigger values for lowland rivers

Temperature (°C)	Turbidity (NTU)	pH	Electrical conductivity (µS/cm or mS/cm)	Available phosphate (mg/L)	Dissolved oxygen (% saturat.)
Stressor: change to indicator (+ or –)	**Stressor**: increase to indicator (+)	**Stressor**: change to indicator (+ or –)	**Stressor**: increase to indicator (+)	**Stressor**: increase to indicator (+)	**Stressor**: change to indicator (+ or –)
Effect: loss of native plants and animals	**Effect**: loss of native plants and animals	**Effect**: loss of native plants and animals	**Effect**: loss of native plants and animals	**Effect**: nuisance plant growth	**Effect**: loss of native plants and animals (fish kills)
Trigger value: 15–35°C for prolonged exposure	**Trigger value**: 50 NTUs	**Trigger value**: 6.5–8.5	**Trigger value**: 200–300 µS/cm or 0.2–0.3 mS/cm	**Trigger value**: 0.06 mg/L (this figure represents 3.06 times the filterable reactive phosphate calculated within the ANZECC guidelines)	**Trigger value**: 85–110%

Adapted from *ANZECC Guidelines 2000* for NSW ecosystems

Trigger values for lakes and reservoirs (standing water*)

Temperature (°C)	Turbidity (NTU)	Electrical conductivity (µS/cm or mS/cm)	pH	Available phosphate (mg/L)	Dissolved oxygen (% saturat.)
Stressor: change to indicator (+ or –)	**Stressor**: increase to indicator (+)	**Stressor**: increase to indicator (+)	**Stressor**: change to indicator (+ or –)	**Stressor**: increase to indicator (+)	**Stressor**: change to indicator (+ or –)
Effect: loss of native plants and animals	**Effect**: loss of native plants and animals	**Effect**: loss of native plants and animals	**Effect**: loss of native plants and animals	**Effect**: nuisance plant growth	**Effect**: loss of native plants and animals (fish kills)
Trigger value: 15–35°C for prolonged exposure	**Trigger value**: 20 NTUs	**Trigger value**: 200–300 µS/cm or 0.2–0.3 mS/cm • amended for disturbed ecosystems in SE NSW	**Trigger value**: 6.5–8.0	**Trigger value**: 0.015 mg/L (this figure represents 3.06 times the filterable reactive phosphate calculated within the ANZECC guidelines)	**Trigger value**: 90–110%

Adapted from *ANZECC Guidelines 2000* for NSW ecosystems

* Standing water bodies have trigger values that differ from streams. No trigger values have been developed for wetlands.

Trigger values for the estuarine and marine catchments

Temperature (°C)	Turbidity (NTU)	Electrical conductivity (µS/cm or mS/cm)	pH	Available phosphate (mg/L)	Dissolved oxygen (% saturat.)
Stressor: change to indicator (+ or –)	**Stressor**: increase to indicator (+)	**Stressor**: increase to indicator (+)	**Stressor**: change to indicator (+ or –)	**Stressor**: increase to indicator (+)	**Stressor**: change to indicator (+ or –)
Effect: loss of native plants and animals	**Effect**: loss of native plants and animals	**Effect**: loss of native plants and animals	**Effect**: loss of native plants and animals	**Effect**: nuisance plant growth	**Effect**: loss of native plants and animals (fish kills)
Trigger value: 15–35°C for prolonged exposure	**Trigger value**: Estuary: 10 NTU Marine: <10 NTU	**Trigger value**: These values do not exist due to tidal influence	**Trigger value**: Estuary: 7.0–8.5 Marine: 8.0–8.4	**Trigger value**: Estuary: 0.015 mg/L Marine: 0.03 mg/L (figures represent 3.06 times the filterable reactive phosphate calculated within the ANZECC guidelines)	**Trigger value**: Estuary: 80–110% Marine: 90–110%

Adapted from *ANZECC Guidelines 2000* for NSW ecosystems

6.2 Australian drinking water guidelines

The Australian drinking water guidelines are applicable to any water intended for drinking, irrespective of its source. The guidelines were developed by the National Health and Medical Research Council (NHMRC) and Agriculture and Resource Management Council of Australia and New Zealand (ARMCANZ). The Waterwatch tests that have drinking water and aesthetics guidelines are faecal coliforms, dissolved oxygen, pH and turbidity.

NHMRC (1996)

Water quality parameter	Health	Aesthetic
Turbidity (NTU)	N/A	5
pH	N/A	6.5–9
Dissolved oxygen (% saturation)	N/A	>95

Summary water quality parameters

Upland >150 m	Healthy	Fair	Poor
Temperature (°C)	18–22	N/A	>22 <15 human impact
Turbidity (NTU)	<10	10–25	>25
EC (µS/cm)	<350	350–800	>800
EC (mS/cm)	<0.35	0.35–0.8	>0.8
pH	6.5–8.0	N/A	<6.5–>8.0
PO_4 (mg/L)	<0.05	0.05–0.3	>0.3
DO (% saturation)	90–110	N/A	<90–>110

Lowland <150 m	**Healthy**	**Fair**	**Poor**
Temperature (°C)	18–22	N/A	>22 <15 human impact
Turbidity (NTU)	<10	10–50	>50
EC (µS/cm)	<300	300–800	>800
EC (mS/cm)	<0.3	0.3–0.8	>0.8
pH	6.5–8.5	N/A	<6.5–>8.5
PO_4 (mg/L)	<0.06	0.06–0.3	>0.3
DO (% saturation)	85–110	N/A	<85–>110
Lakes and dams	**Healthy**	**Fair**	**Poor**
Temperature (°C)	18–22	N/A	>22 <15 human impact
Turbidity (NTU)	<10	10–20	>20
EC (µS/cm)	<300	300–800	>800
EC (mS/cm)	<0.3	0.3–0.8	>0.8
pH	6.5–8.0	N/A	<6.5–>8.0
PO_4 (mg/L)	<0.015	0.015–0.3	>0.3
DO (% saturation)	90–110	N/A	<90–>110
Estuaries	**Healthy**	**Fair**	**Poor**
Temperature (°C)	N/A Affected by tides	N/A Affected by tides	N/A Affected by tides
Turbidity (NTU)	<10	10–20	>20 (may be influenced by tides)
EC (µS/cm or mS/cm)	N/A Affected by tides	N/A Affected by tides	N/A Affected by tides
pH	7–8.5	N/A	<7–>8.5
PO_4 (mg/L)	<0.02	0.02–0.3	>0.3
DO (% saturation)	80–110	N/A	<80 or >110

Adapted from *ANZECC Guidelines 2000*

6.3 Collecting and recording quality data

Quality assurance

Quality assurance means that the data collected is accurate and reliable because a consistent standard has been used when collecting and testing water samples. Waterwatch has quality assurance and quality controls (QA/QC) incorporated into the program. This ensures confidence in the data derived within the program.

The following checklist will help your group produce high quality data:

✓ Use only approved Waterwatch equipment.

✓ Training is required to master the testing skills and to learn about important occupational health and safety issues.

✓ Take care of the equipment – always store in a cool, dry place and clean regularly.

✓ Calibrate your TDS/EC meter before use.

✓ Be involved in quality assurance/quality control events held in your area.

Recording sheets

The remaining pages in this section are recording sheets for recording and interpreting the results of water quality testing activities correctly. Different sheets are provided for:

- minimum data confidence checks – to be printed on the back of all recording sheets
- a range of water quality assessments at multiple sites
- a range of water quality assessments at one site over time
- salinity (EC) measurements at one site over time
- a range of water quality assessments at one freshwater site with interpretation.

Uploading data to the Waterwatch online database

The NSW Waterwatch database is hosted and maintained by the Department of Environment, Climate Change & Water NSW (DECCW). NSW Waterwatch groups upload their data to the website, which is then verified (i.e. checked and approved) by their local coordinator. The database can be accessed by anyone with internet access – other groups, government departments, local councils, Catchment Management Authorities and interested individuals. Data collected by Waterwatch groups can be used to provide important baseline condition information for waterways and assist in monitoring impacts and/or improvements at local sites.

6.4 Minimum data confidence checks

This page is to be printed on the back of all recording sheets

waterwatch NEW SOUTH WALES
Communities Caring for Catchments

All data collected **must** meet the data confidence checks in **bold** below prior to data entry on the online Waterwatch database.

			Yes	No
1. Group is part of the NSW Waterwatch Program	A. The group has registered a site on the Waterwatch online database.	**A**		
	B. The group has developed a Waterwatch Plan.	**B**		
2. Training	C. The data is collected by group members who have been trained in Waterwatch methods and procedures.	**C**		
	D. The group follows the Waterwatch procedures provided for each test.	**D**		
3. Equipment	E. Only equipment used by NSW Waterwatch is used to collect data.	**E**		
	F. EC meters are calibrated.	**F**		
	G. Equipment is cleaned and maintained according to procedures.	**G**		
4. Chemicals	H. Chemical use-by dates have been checked to make sure they are within the date range.	**H**		
5. Recording	I. Standard units are used for all parameters.	**I**		
	J. Waterwatch recording sheets are used in the field.	**J**		
6. Data management	K. Care is taken when converting results from one measurement to another. For example, EC units to µS/cm, dilution factor conversions and faecal coliform counts.	**K**		

Improving data confidence: Groups should use the data confidence checks above to improve their monitoring. Quality assurance tests are conducted each year to allow groups to check the accuracy of their results for equipment and procedure errors. **It is essential that groups participate in these events.**

Note: Additional QA checks are provided through the verification of data by your Waterwatch coordinator.

6.5 Result sheet for a range of water quality assessments at multiple sites

waterwatch NEW SOUTH WALES
Communities Caring for Catchments

Group: ..... Date: .....

EC meter calibrated? ☐ Yes ☐ No (do not enter data)

Calibration solution: High range meter: ..... mS/cm Low range meter: ..... µS/cm

Site	Temperature (°C)	pH	EC (mS/cm)	EC (mS/cm x1000 = µS/cm)	Turbidity (NTU)	Available phosphate (mg/L)	Dissolved oxygen (mg/L)

Upload your data as soon as possible after testing to www.waterwatch.nsw.gov.au.

6.6 Result sheet for a range of water quality assessments at one site over time

waterwatch NEW SOUTH WALES
Communities Caring for Catchments

DATE:						
TIME:						
RAIN IN LAST WEEK (nil, little, a lot, etc.)						
OBSERVATIONS						
FLOW (dry, no flow, low, high)						
NUMBER OF VOLUNTEERS						
TIME TAKEN						
AIR TEMPERATURE (°C)						
WATER TEMPERATURE (°C)						
pH (units)						
ELECTRICAL CONDUCTIVITY (µS/cm)	CAL: ☐ YES ☐ NO	CAL: ☐ YES ☐ NO	CAL: ☐ YES ☐ NO	CAL: ☐ YES ☐ NO	CAL: ☐ YES ☐ NO	CAL: ☐ YES ☐ NO
TURBIDITY (NTU)						
AVAILABLE PHOSPHATE (mg/L)						
DISSOLVED OXYGEN (mg/L)						
DISSOLVED OXYGEN (% saturation)						
E. *coli* (CFU/100 mL)						
Comments:						

Upload your data as soon as possible after testing to www.waterwatch.nsw.gov.au.

6.7 Waterwatch freshwater result sheet

Group: .....

Date sampled: .....

Site name: .....

Time sampled: .....

Number of volunteers: ..... Time taken: .....

Rainfall last week *(please tick)*:
- ☐ within 24 hours
- ☐ 1–7 days

Rainfall description *(please tick)*:
- ☐ light
- ☐ medium
- ☐ heavy

EC meter calibration *(please tick)*:
- ☐ YES
- ☐ NO (if NO do **not** enter data)

Calibration solution *(please tick)*:
- ☐ High range meter: ..... mS/cm
- ☐ Low range meter: ..... µS/cm

Flow *(please tick)*:
- ☐ no flow
- ☐ slow
- ☐ fast
- ☐ flood

Observations (weather conditions, visible pollution, wildlife present, odour, algae, etc.)

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Test	Units	Results	Result range*	Guidelines (tick the box)
Temperature	°C		There are no trigger values for temperature to apply a healthy or poor rating. The acceptable temperature range will depend specifically on your site.	
pH	pH units		<6	☐ poor
			6–8	☐ healthy
			>8	☐ poor
Electrical conductivity (salinity)	µS/cm		<300	☐ healthy
			300–800	☐ fair
			>800	☐ poor
Turbidity	NTU		≤10	☐ healthy
			15–30	☐ fair
			>30	☐ poor
Available phosphate	mg/L		<0.05	☐ healthy
			0.05–<0.3	☐ fair
			≥0.3	☐ poor
Dissolved oxygen	% saturation		<80	☐ poor
			80–110	☐ very good
			>110	☐ poor
E. *coli*	CFU/100 mL		<150	Primary contact
			<1000	Secondary contact

* Adapted from *ANZECC Guidelines 2000*

Upload results to the Waterwatch online database: www.waterwatch.nsw.gov.au

Dissolved oxygen

Results: ..... mg/L

Less than 2 mg/L is very low – will kill many organisms that rely on oxygen in the water

Less than 5 to 6 mg/L is low – will cause increased stress to aquatic organisms

Calculate % saturation from graph: ..... %

Tick the box below that relates to your result:

Poor: ecosystems at risk
☐ Less than 90%

Healthy
☐ 90 to 110%

Poor: ecosystems at risk
☐ Greater than 110%

6.8 Summary water quality results: lakes and dams (EC meters)

Field results sheet: lakes & dams

Site name: .....

Date: .....

Catchment: .....

Location in catchment: .....

Observations at the site: .....

Record your results as soon as possible after testing: www.waterwatch.nsw.gov.au

Water quality triggers

The point where a change in water quality affects river health.

	Temp*	pH Increase or decrease may affect waterway health	EC Increase may affect waterway health	Turbidity Increase may affect waterway health	Available phosphate	DO saturation
Value	N/A	6.5–8.0	300 µS/cm 0.30 mS/cm	20 NTUs	0.015 mg/L	90–110%

* Guidelines apply only to human exposure.
Source: *ANZECC Guidelines 2000*

Temperature

Results: Water Temp. ..... °C Air Temp. ..... °C

No set trigger value. Animals and plants in and around aquatic ecosystems are adapted to living within a particular temperature range to maintain their survival and provide reproduction cues. Taking note of 'normal' temperature conditions at this site will allow you to determine whether an increase or decrease in temperature might be harmful to this ecosystem.

pH

Results: .....

An increase or decrease in pH may cause a loss of native plants and animals.

Tick the boxes below that relate to your result:

1	Acid	Neutral 7	Alkaline	14

My result is: ☐ Acid ☐ Neutral ☐ Alkaline

Healthy
☐ 6.5 to 8.0

Poor: ecosystems at risk
☐ <6.5 or >8.0

Turbidity

Results: ..... NTU

Increases in turbidity may cause a loss of native plants and animals.

Tick the box below that relates to your result:

Healthy
☐ Less than 10 NTU

Fair: may affect ecosystems
☐ 10 to 20 NTU

Poor: ecosystems at risk
☐ Greater than 20 NTU

Salinity

Results: ..... mS/cm X 1000 = ... µS/cm

Salinity is measured by its electrical conductivity (EC)

Tick the box below that relates to your result:

Healthy
☐ Less than 300 µS/cm
Less than 0.3 mS/cm

Fair: may affect ecosystems
☐ 300 to 800 µS/cm
0.30 to 0.8 mS/cm

Poor: ecosystems at risk
☐ Greater than 800 µS/cm
Greater than 0.8 mS/cm

Available phosphate

Results: ..... mg/L

Tick the box below that relates to your result:

Healthy
☐ Less than 0.015 mg/L

Fair: may affect ecosystems
☐ 0.015 to 0.3 mg/L

Poor: ecosystems at risk
☐ Greater than 0.3 mg/L

Dissolved oxygen

Results: ..... mg/L

Less than 2 mg/L is very low – will kill many organisms that rely on oxygen in the water

Less than 5 to 6 mg/L is low – will cause increased stress to aquatic organisms

Calculate % saturation from graph: ..... %

Tick the box below that relates to your result:

Poor: ecosystems at risk
☐ Less than 85%

Healthy
☐ 85 to 110%

Poor: ecosystems at risk
☐ Greater than 110%

6.9 Summary water quality results: lowland rivers (EC meters)

Field results sheet: lowland rivers

Site name: .....

Date: .....

Catchment: .....

Location in catchment: .....

Less than 150 metres above sea level.

Observations at the site: .....

Record your results as soon as possible after testing: www.waterwatch.nsw.gov.au

Water quality triggers

The point where a change in water quality affects river health.

	Temp*	pH Increase or decrease may affect waterway health	EC Increase may affect waterway health	Turbidity Increase may affect waterway health	Available phosphate	DO saturation
Value	N/A	6.5–8.5	300 µS/cm 0.30 mS/cm	50 NTUs	0.06 mg/L	85–110%

* Guidelines apply only to human exposure.
Source: *ANZECC Guidelines 2000*

Temperature

Results: Water Temp. ..... °C Air Temp. ..... °C

No set trigger value. Animals and plants in and around aquatic ecosystems are adapted to living within a particular temperature range to maintain their survival and provide reproduction cues. Taking note of 'normal' temperature conditions at this site will allow you to determine whether an increase or decrease in temperature might be harmful to this ecosystem.

pH

Results: .....

An increase or decrease in pH may cause a loss of native plants and animals.

Tick the boxes below that relate to your result:

1	Acid	7 Neutral	Alkaline	14

My result is: ☐ Acid ☐ Neutral ☐ Alkaline

Healthy
☐ 6.5 to 8.5

Poor: ecosystems at risk
☐ <6.5 or >8.5

Turbidity

Results: ..... NTU

Increases in turbidity may cause a loss of native plants and animals.

Tick the box below that relates to your result:

Healthy
☐ Less than 10 NTU

Fair: may affect ecosystems
☐ 10 to 50 NTU

Poor: ecosystems at risk
☐ Greater than 50 NTU

Salinity

Results: ..... mS/cm X 1000 = ... µS/cm

Salinity is measured by its electrical conductivity (EC).

Tick the box below that relates to your result.
Convert mS/cm to µS/cm by x 1000 in brackish water:

Healthy
☐ Less than 300 µS/cm
Less than 0.3 mS/cm

Fair: may affect ecosystems
☐ 300 to 800 µS/cm
0.30 to 0.8 mS/cm

Poor: ecosystems at risk
☐ Greater than 800 µS/cm
Greater than 0.8 mS/cm

Available phosphate

Results: ..... mg/L

Tick the box below that relates to your result:

Healthy
☐ Less than 0.06 mg/L

Fair: may affect ecosystems
☐ 0.06 to 0.3 mg/L

Poor: ecosystems at risk
☐ Greater than 0.3 mg/L

Dissolved oxygen

Results: ..... mg/L

Less than 2 mg/L is very low – will kill many organisms that rely on oxygen in the water

Less than 5 to 6 mg/L is low – will cause increased stress to aquatic organisms

Calculate % saturation from graph: ..... %

Tick the box below that relates to your result:

Poor: ecosystems at risk
☐ Less than 90%

Healthy
☐ 90 to 110%

Poor: ecosystems at risk
☐ Greater than 110%

Water quality triggers

The point where a change in water quality affects river health.

	Temp*	pH Increase or decrease may affect waterway health	EC Increase may affect waterway health	Turbidity Increase may affect waterway health	Available phosphate	DO saturation
Value	N/A	6.5–8.5	350 µS/cm 0.35 mS/cm	10 NTUs	0.05 mg/L	90–110%

* Guidelines apply only to human exposure.
Source: *ANZECC Guidelines 2000*

6.10 Summary water quality results: upland rivers (EC meters)

Field results sheet: upland rivers

Site name: .....

Date: .....

Catchment: .....

Location in catchment: .....

More than 150 metres above sea level.

Observations at the site: .....

Record your results as soon as possible after testing: www.waterwatch.nsw.gov.au

Temperature

Results: Water Temp. ..... °C Air Temp. ..... °C

No set trigger value. Animals and plants in and around aquatic ecosystems are adapted to living within a particular temperature range to maintain their survival and provide reproduction cues. Taking note of 'normal' temperature conditions at this site will allow you to determine whether an increase or decrease in temperature might be harmful to this ecosystem.

pH

Results: .....

An increase or decrease in pH may cause a loss of native plants and animals.

Tick the boxes below that relate to your result:

1	Acid	Neutral 7	Alkaline	14

My result is: ☐ Acid ☐ Neutral ☐ Alkaline

Healthy

☐ 6.5 to 8.0

Poor: ecosystems at risk

☐ <6.5 or >8.0

Turbidity

Results: ..... NTU

Increases in turbidity may cause a loss of native plants and animals.

Tick the box below that relates to your result:

Healthy

☐ Less than 10 NTU

Fair: may affect ecosystems

☐ 10 to 25 NTU

Poor: ecosystems at risk

☐ Greater than 25 NTU

Salinity

Results: ..... mS/cm X 1000 = ... µS/cm

Salinity is measured by its electrical conductivity (EC)

Tick the box below that relates to your result:

Healthy

☐ Less than 350 µS/cm
Less than 0.35 mS/cm

Fair: may affect ecosystems

☐ 350 to 800 µS/cm
0.35 to 0.8 mS/cm

Poor: ecosystems at risk

☐ Greater than 800 µS/cm
Greater than 0.8 mS/cm

Available phosphate

Results: ..... mg/L

Tick the box below that relates to your result:

Healthy

☐ Less than 0.05 mg/L

Fair: may affect ecosystems

☐ 0.05 to 0.3 mg/L

Poor: ecosystems at risk

☐ Greater than 0.3 mg/L

SECTION 7

Habitat: the aquatic zone

Land based activities affect water quality and river health. Biological monitoring complements water quality monitoring and helps measure change, identify risks and plan management actions.

This section provides background information and assessment sheets to assist Waterwatch participants to conduct biological assessments of the aquatic zone at their site. Waterbird and water plant identification charts and observation sheets are also provided. These resources will increase participants' understanding of the habitats associated with waterways and provide them with essential skills for doing their own biological assessments.

Note: The *Senior Waterwatch Teachers' Guide* and the *Community/Land Manager Waterwatch Guide* contain further background information relating to the biological assessments described in this section.

7.1 Common and widespread wetland plants

In identifying wetland plants, local species lists should be used where possible.

Plant type	Common name	Botanical name
Free-floating aquatic plants	Azolla	*Azolla* species
	Duckweeds	*Lemna, Spirodela* and *Riccia* species
Submerged aquatic plants	Water milfoil	*Myriophyllum* species
	Pondweeds	*Potamogeton* species
	Hornwort	*Ceratophyllum demersum*
	Water ribbons	*Triglochin procera*
	Ribbonweed	*Vallisneria gigantica*
	Stoneworts	*Chara* and *Nitella* species
Floating-leafed aquatic plants	Water primrose	*Ludwigia peploides*
	Wavy marshwort	*Nymphoides crenata*
	Floating pondweed	*Potamogeton tricarinartus*
	Swamp lily	*Otellia ovatifolia*
	Water lily	*Nymphaea* species
	Water snowflake	*Nymphoides* species
Emergent aquatic plants	Cumbungi or bulrush	*Typha* species
	Common reed	*Phragmites australis*
	Rushes	*Juncus* species
	• giant rush	*J. ingens*
	• common Rush	*J. usitatus*
	Sedges	*Cyperus* species
	• giant sedge	*C. exaltus*
	• tussock sedge	*Carex appressa*
	• common sedge	*Carex tereticaulis*
	Spike-rushes	*Eleocharis* species
	• common spike-rush	*E. acuta*
	• tall spike-rush	*E. sphacelata*
	Water plantain	*Alisma plantago aquatica*
	Water ribbons	*Triglochin procera*
	Water couch	*Paspalum. distichum*

Plant type	Common name	Botanical name
Semi-aquatic plants – shallow water	Billy buttons	*Craspedia* species
	Buttercup	*Ranunculus* species
	Nardoo	*Marsilea* species
	Swamp wallaby grasses	*Amphibromus* species
Semi-aquatic plants – damp or drying soil	Joyweed	*Alternantra denticulata*
	Dense stonecrop	*Crassula colorata*
	Swamp stonecrop	*Crassula helmsii*
	Spiny-head mat-rush or basket grass	*Lomandra longifolia*
	Sneezeweed	*Centipeda* species
	Knotweed or smartweed	*Persicaria* species
	Starwort	*Stellaria* species
Fringing woodland	Gums	*Eucalyptus* species
	• River red gum	*E. camaldulensis*
	• Black box	*E. largiflorens*
	• Coolibah	*E. coolabah*
	• Yapunyah	*E. yapunyah*
	• Apple box	*E. bridgesiana*
	• Swamp mahogany	*E. robusta*
	River she oak	*Casuarina cunninghamiana*
	River bottlebrush	*Callistemon sieberi*
	Teatree	*Leptospermum continentale*
	Paperbarks	*Melaleuca* species
	Wattles	*Acacia* species
	• Silver wattle	*A. dealbata*
	• River cooba	*A. stenophylla*
	Lignum	*Muehlenbeckia cunninghamii*
	Nitre goosefoot	*Chenopodium nitrariaceum*

Source: Adapted from *Are there seeds in your wetland?* (Brock, 2000)

7.2 Wetland plant key

Detached

Attached

Free-floating

Submerged

Duckweed

Hornwort

Rooted

Upright emergent

Submerged plants

Tall

Low

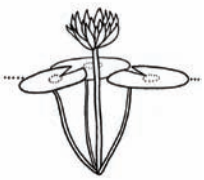
Cumbungi

Sedge

Leaves

Floating

Submerged



Water lily

Ribbonweed

Semi-aquatic

Fringing vegetation

Damp or drying soil

Shallow water

Knotweed

Nardoo

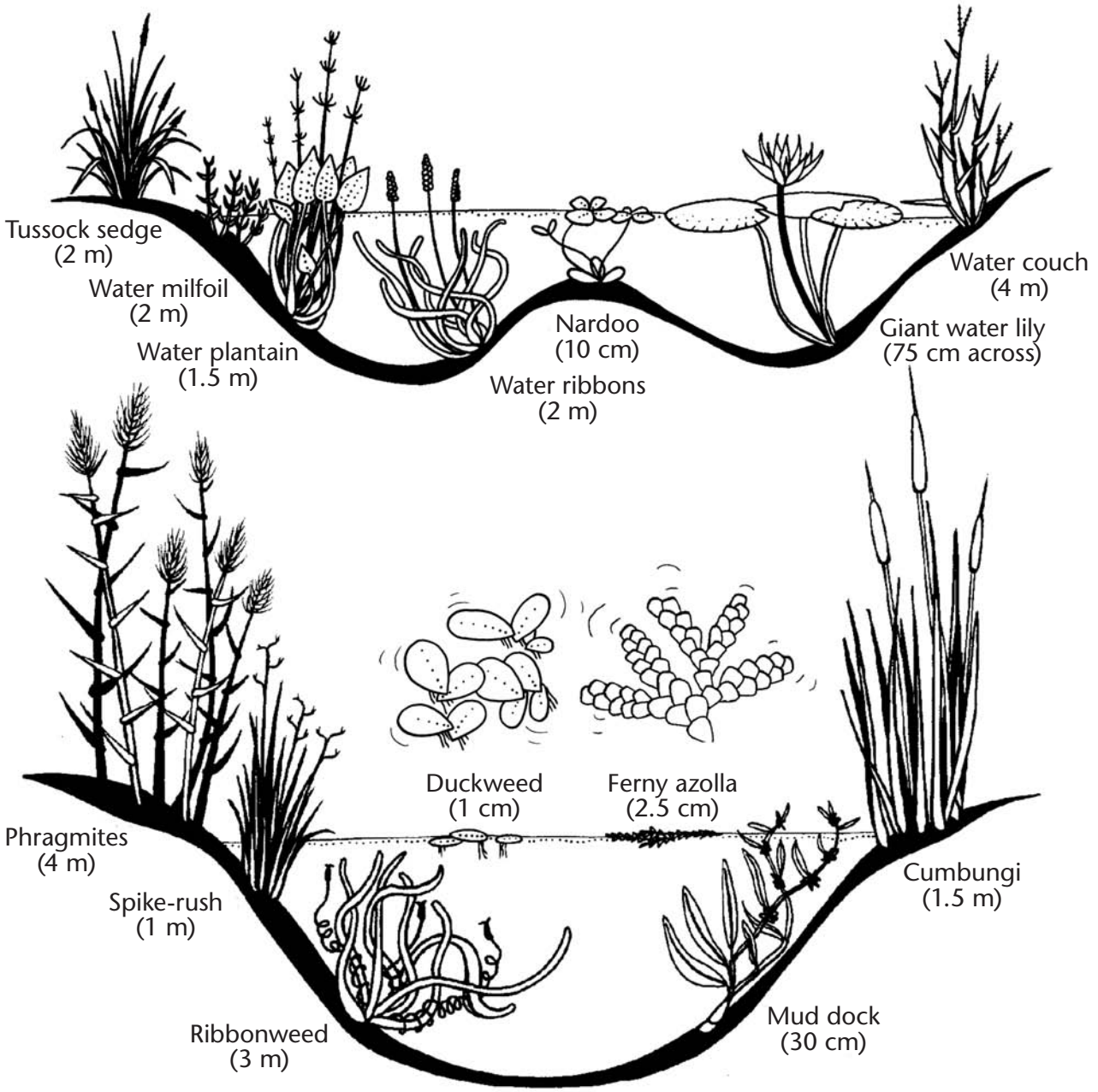
Trees and shrubs

Source: Adapted from *Are there seeds in your wetland?* (Brock, 2000)

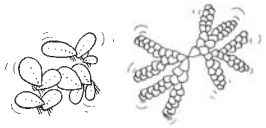
7.3 Wetland plant identification

Common water plants

Use the identification guide below to match the plant features and growth to water plants at your site.



7.4 Habitats in your wetland

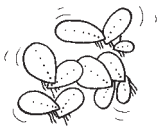
Habitat	Water presence	Types of plants
Terrestrial		
Above the highest water level	Dry – water rarely reaches this area	Eucalypts, wattles, she oaks, pasture grasses and dryland plants
Water's edge		
Between the water's edge and the highest water level	Water sometimes reaches this area	Reeds, grasses, herbs and pasture weeds
At the water's edge	Water levels fluctuate	Sedges, rushes, knotweed, milfoil, grasses and other flowering plants 
Shallow water	This area sometimes dries out	Algae, rushes and reeds
Submerged zone		
Deep water	Rarely dries out	Ribbonweed and pondweed 
Water surface	This habitat disappears when the water dries out	Floating duckweed and azolla 

Source: Adapted from *Are there plants in your wetland?* (Brock & Casanova, 2000)

7.5 Aquatic plants at the waterway

Tip: Macrophytes = water plants easily seen with the naked eye

Conduct a survey of the macrophytes at the site using the ID sheet below.

Habitat and form	Tick if present	Name or description of plants at your site
Free-floating 	☐	
Floating but attached 	☐	
Submerged (not feathery) 	☐	
Submerged and emergent (feathery) 	☐	
Emergent (narrow leaf) 	☐	
Emergent (broad leaf) 	☐	

7.6 Assessment of common freshwater plant, animal, bird and fish species of Aboriginal significance

Site name: .....

Date: .....

The following plants, animals and birds live in or near waterways and wetlands and are important to Aboriginal people. Tick the boxes to indicate the species observed or known to exist at this site.

Indicator trees	**Botanical names**	**Yes**	**No**
River she oak	*Casuarina cunninghamiana*	☐	☐
River red gum	*Eucalyptus camaldulensis*		
Indicator shrubs	**Botanical names**	**Yes**	**No**
River bottlebrush	*Callistemon sieberi*	☐	☐
Teatree	*Leptospermum continentale*	☐	☐
Water plants	**Botanical names**	**Yes**	**No**
Bulrush/cumbungi	*Typha orientalis* Presl	☐	☐
Common reed	*Phragmite communis*	☐	☐
Mat rushes	*Lomandra* species	☐	☐
Nardoo	*Marsilea drummondii*	☐	☐
Sedges/tall spike-rush	*Eleocharis sphacelata*	☐	☐
Water pepper	*Polygonum hydropiper*	☐	☐

Common animals	**Scientific names**	**Yes**	**No**
Platypus	*Ornithorhychus anatinus*	☐	☐
Water rat	*Hydromys chrysogaster*	☐	☐
Waterbirds	**Scientific names**	**Yes**	**No**
Black duck	*Anas superciliosa*	☐	☐
Black swan	*Cygnus atratus*	☐	☐
Brolga	*Grus rubicunda*	☐	☐
Pelican	*Pelicanus conspicillatus*	☐	☐
Great egret	*Ardea alba*	☐	☐
Fish/invertebrates	**Scientific names**	**Yes**	**No**
Macquarie perch	*Macquaria australasica*	☐	☐
Murray cod	*Maccullochella peelii peelii*	☐	☐
Silver/golden perch	*Bidyanus bidyanus/Macquaria ambigua*	☐	☐
Short finned eel	*Anguilla australis*	☐	☐
Fresh water mussel	*Unionidae* and other species	☐	☐
Murray River crayfish	*Euastacus armatus*	☐	☐
Shrimp	*Paratya* or *Macrobrachium* species	☐	☐
Trout cod	*Maccullochella macquariensis*	☐	☐
River black fish	*Gadopis marmoratus*	☐	☐
Catfish	*Tandanus tandanus*	☐	☐

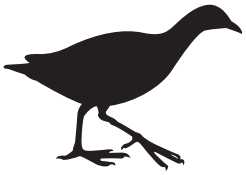
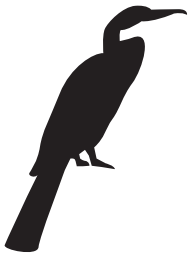
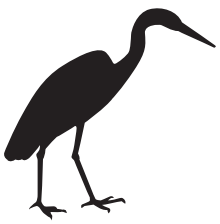
7.7 Water bird identification chart

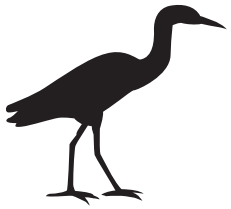
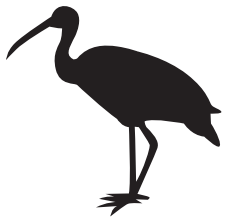
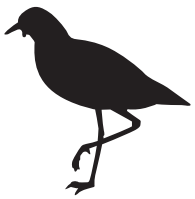
Nomadic: moves to meet need for food and water

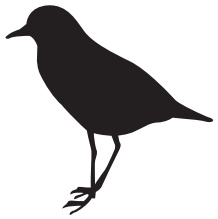
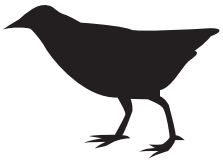
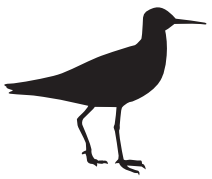
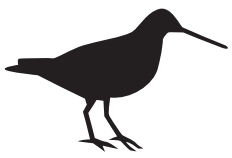
Resident: stays in the same location

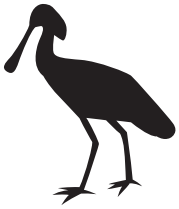
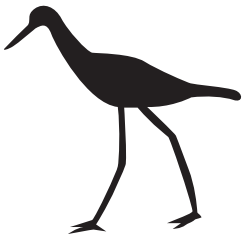
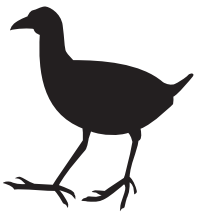
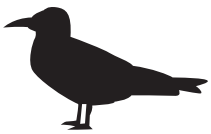
Migratory: moves with the seasons

Bird	Distribution	Diet	Preferred habitat	Nesting
Bittern 	Most of NSW except far NW Nomadic	Feeds mainly at night on frogs, fish, yabbies, spiders, insects and snails	Favours permanent freshwater wetlands with tall, dense vegetation, particularly bullrushes (*Typha* spp.) and spike-rushes (*Eleocharis* spp.)	Breeding occurs in summer from Oct–Jan, on a platform of reeds
Black duck 	Throughout NSW Resident	Aquatic invertebrates and water weeds. Filter feeds. Puts head in water and stirs up bottom	Flowing and still water (can live in polluted water)	July–Nov. Nests in vegetation – can use poor quality vegetation
Coot 	Throughout NSW Nomadic	Plant matter	Lakes and still water	Aquatic vegetation carried back to the nest after being collected by diving. Nests may be floating
Cormorant 	Throughout NSW Migratory	Aquatic invertebrates, frogs and fish	Large water bodies several metres deep	Breed in colonies only after inland flooding

Bird	Distribution	Diet	Preferred habitat	Nesting
Crake 	Most of NSW except far north. Resident	Aquatic insects, larvae, hatching flies, tadpoles, invertebrates. Always feeds under cover	Shallows and margins of freshwater or saline wetlands	Nests in reeds Aug–Feb
Crane: brolga 	SE Australia Nomadic	Fleshy parts of aquatic plants, aquatic insects such as dragonflies and beetles, spiders, frogs and small fish	Shallow swamps and flooded grasslands	July–Dec in herb and sedge swamps with nests of floating vegetation away from the shore
Darter 	Throughout Australia Resident	Insects, fish or tortoises	Freshwater and saline lakes, swamps and rivers, prefers sheltered areas	Breeds in spring and the male will defend a site and decorate it with leafy twigs. Nests tends to be solitary in summer
Egret 	Throughout NSW Nomadic	Fish, frogs and invertebrates	Still water or still parts of flowing water	Sept–Nov in small colonies
Grebe 	Throughout NSW Resident	Small fish and water insects	Wetlands	Sept–Jan in the south and Jan–Apr in the north

Bird	Distribution	Diet	Preferred habitat	Nesting
Heron 	Throughout NSW Resident and nomadic depending on type	Fish, frogs, tadpoles, aquatic invertebrates and vegetation	Still water (e.g. wetlands and dams) and still parts of flowing water. Hunts in the shallows	Different species breed at different times throughout the year. May breed outside of season in response to rainfall
Ibis 	Throughout NSW Nomadic	Terrestrial and aquatic invertebrates, crayfish and mussels	Swamps, lagoons, flood plains, grasslands, parks and gardens	One or two broods may be reared in a year
Lapwing 	Throughout NSW Resident	Insects, crabs, worms, yabbies and other small crustaceans, invertebrates, seeds and herbage	Swamps, flooded grounds with short grass, paddocks with dams, airfields, near beaches and wetlands	Masked lapwings may breed at any time when conditions are suitable
Moorhen 	Throughout NSW Nomadic	Seeds and aquatic vegetation	Found in freshwater wherever there is aquatic vegetation	Nests of aquatic vegetation and may be floating
Pelican 	Throughout NSW Nomadic	Fish, aquatic animals including crustaceans, tadpoles and turtles	Throughout the continent wherever there is water. Widely distributed in cool temperate to tropical lakes, rivers and estuaries	Colonial breeder – may breed at any time throughout the year

Bird	Distribution	Diet	Preferred habitat	Nesting
Plover 	Widespread across NSW and not usually found outside Australia	Invertebrates such as worms, snails and water beetles, and plant material such as seeds	Small bodies of water and farm dams	Nests scraped out of gravel within about 15 metres of the water's edge
Rail 	Throughout NSW Resident	Crustaceans, molluscs, insects, seeds, fruit, frogs, carrion and refuse. Mostly feeds early in the morning and the evening	Dense reeds and vegetation bordering many types of wetlands or crops. Makes widespread use of artificial wetlands like sewage ponds and drainage channels	Nests in long grass, tussocks, rushes or crops
Sandpiper 	Throughout NSW Migratory	Invertebrates such as worms, snails and water beetles, and plant material such as seeds	They are waders and feed along the edge of lakes	Main breeding season May–Aug in Northern Hemisphere
Snipe 	Throughout NSW Migratory	Invertebrates such as worms, snails and water beetles, and plant material such as seeds	Inhabits shallow freshwater wetlands, vegetated ephemeral and permanent lakes and swamps, and inundated grasslands	Nests on the ground amongst tall vegetation such as grass tussocks and reeds

Bird	Distribution	Diet	Preferred habitat	Nesting
Spoonbill 	Throughout NSW Nomadic	Fish and other water animals, such as shellfish, crabs and frogs	Wetlands and in the shallow parts of lakes and rivers	Main breeding season Oct–Apr. When they are breeding, long white feathers grow from the back of their heads
Stilt 	Throughout NSW Resident	Molluscs, insects, diatoms, brine shrimp from mud	Edge of still water (up to feather line)	Aug–Nov in small colonies. Don't need much vegetation to nest
Swamphen 	SE NSW Nomadic	Reeds, stems, grass and little animals	Still waters	July–Nov in reeds. Also uses reeds for shelter
Swan 	Prefers southern parts NSW Resident	Vegetation – submerged plants, algae. Grazes on pasture on banks	Salt, brackish or fresh waterways and permanent wetlands, requiring 40 metres or more of clear water to take off	April–Jan. Prefers wet season when sufficient vegetation can be uprooted to form a platform nest
Tern 	Migratory bird to Australia	Insects taken on the wing (dragonflies), water insects (beetles) and grasshoppers. Small fish and crustaceans also eaten	Shallow swamps, coastal dwellers	Breeding in summer on islands or a twig structure anchored to the bottom

7.8 Water bird field observation sheet

Site name: .....

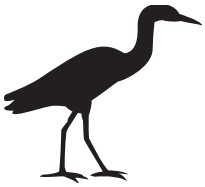
Date: .....

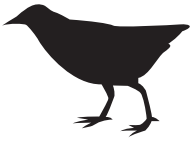
Below are some common waterbirds that you may see at your site. Record the number for each waterbird you observe.

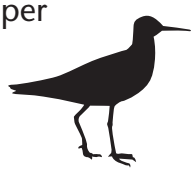
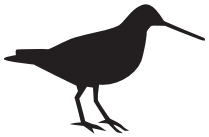
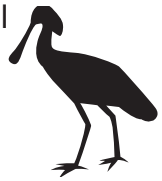
Number
R = rare (0–10)
U = uncommon (11–100)
C = common (101–1000)
A = abundant (1000s)

Activity
R = roosting
F = feeding
N = nesting
F = flying

Bird	Number	Activity
Australian bittern 		
Black duck 		
Coot 		
Cormorant 		

Bird	Number	Activity
Darter 		
Duck 		
Grebe 		
Heron 		

Bird	Number	Activity
Ibis 		
Lapwing 		
Moorhen 		
Pelican 		
Plover 		
Rail 		

Bird	Number	Activity
Sandpiper 		
Snipe 		
Spoonbill 		
Stilt 		
Swan 		
Tern 		

Source: Kingsford (1991), *Australian waterbirds: a field guide* (Kangaroo Press, NSW, 1991).

Make a list of other birds seen at the site that may not be waterbirds, and record the number. For example, cockatoos, magpies and kookaburras.

Bird	Number	Activity

Bird	Number	Activity

7.9 Field observation sheet: Where are the birds?

Site name: .....

Date: .....

Refer to the information provided in Section 7.7 to complete this exercise. A field guide to birds would also be helpful.

For the following groups of birds:

1. Count the number of each type.
2. Record where they were seen: open water, edge or among plants.

Bird group	**Number**	**Location** O = open water E = edge V = in vegetation	**Beak/feet activity** F = feeding S = swimming D = diving W = wading WOP = walking on plants
Ducks			
Cormorants and darters			
Stilts and herons			
Crakes and rails			
Spoonbills			
Other			
Other			

SECTION 8

Habitat: the riparian zone

The riparian zone extends along the banks of a river, creek or wetland. It is an important link between the aquatic environment and the adjoining land. It provides food and shelter for aquatic, semi-aquatic and land animals such as lizards, snakes, bats, frogs and birds.

This section provides important essential skills for doing biological assessments in the riparian zone. It also contains field assessments in the riparian zone including rapid assessments of riparian condition and habitat, and an assessment of the level of weed infestation at a site.

Included in this section: *Page*

Note: The *Senior Waterwatch Teachers' Guide* and the *Community/Land Manager Waterwatch Guide* contain further background information relating to the biological assessments described in this section.

8.1 Essential skill: quadrats

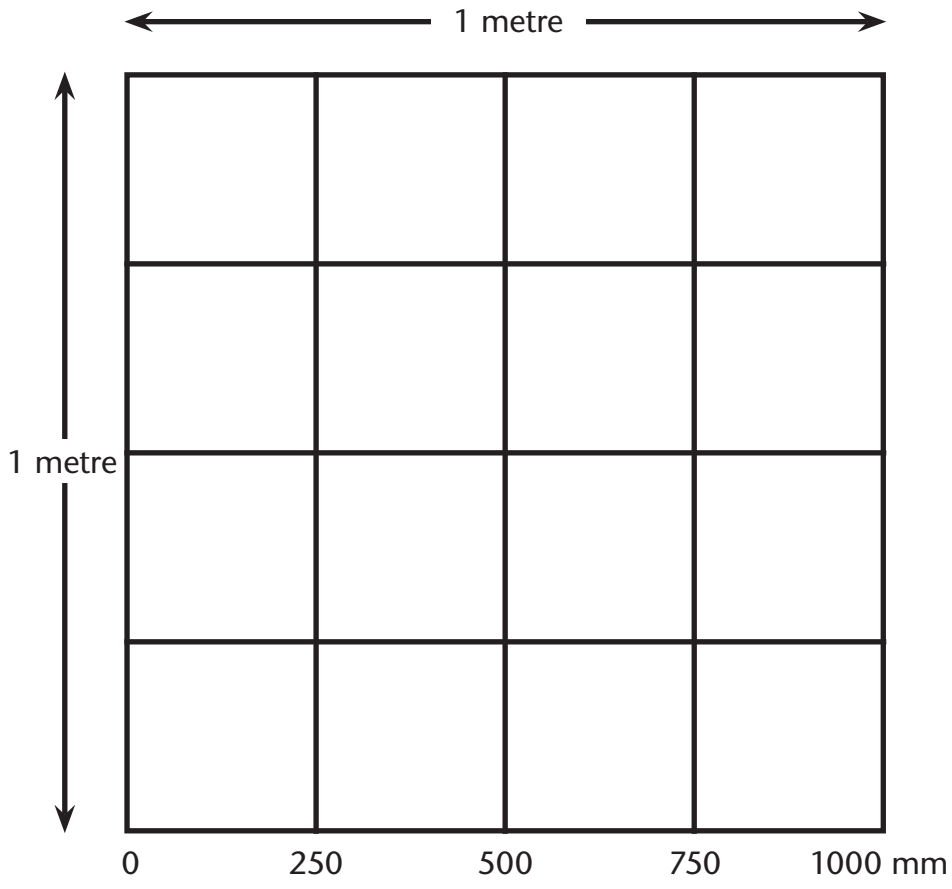
What is a quadrat?

To find out more detail about your site, you can focus on a small area. This area can be identified by using a quadrat.

A quadrat will vary in size depending on the purpose of monitoring. For vegetation transects a quadrat of 1.0 metre square can be used. Quadrats can be made using pegs and string or PVC pipe joined together with connectors.

Drawing a quadrat map

Record the **date** and the **name of the site** and locate **species** within the quadrat. Use a key to develop a species list for the quadrat.



8.2 Essential skill: transects

What is a transect?

Observing changes at your site can be made using a transect. This is a line between two points. The edge of a lake, stream or boardwalk or a tape measure can provide the line.

Types of transect

There are two types of transect:

1. A **line transect** is where individual plants touching a measuring tape stretched across an area are recorded at regular intervals. This method can be used to determine the variety of species from the riverbank to the riparian zone or to measure disturbances within the riparian zone.
2. A **belt transect** is one where one side of the quadrat frame is placed next to the tape at regular intervals. This provides a closer look at species composition within a quadrat. This type of transect may also involve an assessment between parallel transect lines along the full length of the transect. Belt transects can be used to conduct surveys in the aquatic and riparian zone.

How to establish a transect

1. Mark the start of the transect (0 m) with a peg, pole or other object that can be fixed into the ground.

2. Record the intended direction of the transect as a compass bearing.

3. To lay out the transect, set the compass to the bearing and note a feature or landmark on the same bearing. A photo may be helpful for future reference.

4. Attach the end of the tape measure to the transect marker and walk in a straight line following the bearing or walk directly towards the feature you have noted.

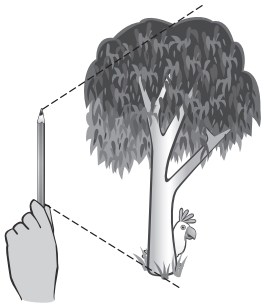
5. Stop when the length of the transect or the boundary of the study area has been reached.

8.3 Essential skill: estimating the height of a tree without measuring tape

Equipment: pencil, ruler or tape measure

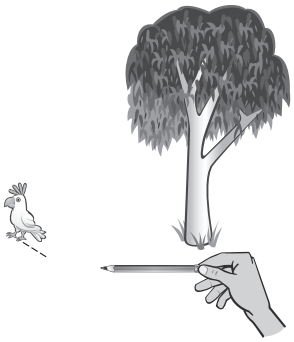
1. Ask a person to stand facing you in front of the tree.

2. Stand quite a distance away from the tree and hold a pencil out in front of you.



3. Line up the tip of the pencil with the top of the tree and the bottom of the pencil with your friend.

4. Turn the pencil so that it is lying down. Keep your thumb aligned with the base of the tree.



5. Ask the person to walk to the side until your friend is in line with the tip of the pencil.

6. Measure the distance using a tape measure or pace the distance from your friend to the base of the tree.

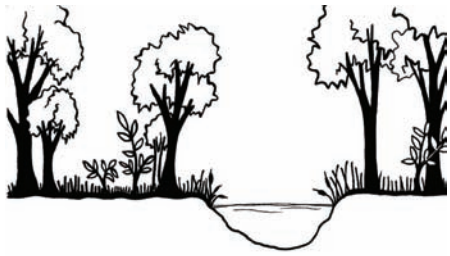
7. This is the height of the tree.

Note: A more accurate measurement can be obtained by using a clinometer.

8.4 Rapid observations at the site: riparian condition

Site name: .....

Date: .....

Appearance of my site Tick the box that most looks like your site	**Stream condition** Tick the boxes for the site that looks most like your site
1 	**Good** ☐ Native vegetation along the entire stretch ☐ Distinct tree, shrub and grass layers of native vegetation ☐ Area protected from human use, e.g. fences keep out stock, no pollution evident
2 	**Average/moderate** ☐ Vegetation patchy ☐ Distinct vegetation layers not visible ☐ Many weeds ☐ Some bank erosion ☐ Some human use of the area evident, e.g. drains or stock access
3 	**Poor** ☐ Little native vegetation ☐ Exotic tree species present ☐ Some trees have dead limbs or look unhealthy ☐ Weeds ☐ Human use obvious, e.g. stock access, drains, vegetation removal
4 	**Very poor** ☐ Erosion across a wide area ☐ Little riparian vegetation or absent ☐ Bank erosion evident ☐ Many changes caused by human use, e.g. pollution, stock access, drains

8.5 Weed assessment at your site

Site name: .....

Date: .....

This assessment should be conducted using a weed guide for your local area.

Complete the table below:

Step 1: Assess the weed cover in each vegetation layer.

Step 2: Identify the main weeds in each vegetation layer – tree, understorey and groundcover.

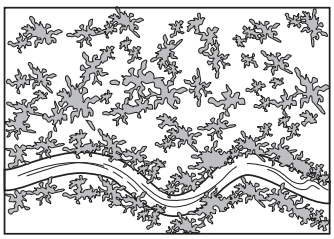
Step 3: Assess the level and type of weed infestation and complete the table:

Vegetation layer	Level of infestation				Main species
	Heavy (>60%)	Mod (30–60%)	Low (10–30%)	Very Low (<10%)	
Canopy					
Understorey					
Groundcover					

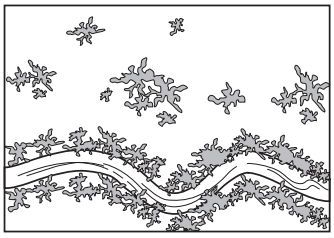
Note: A weed is a plant that is not native and not wanted in the area.

Assess the level and type of infestation

Heavy to moderate infestations

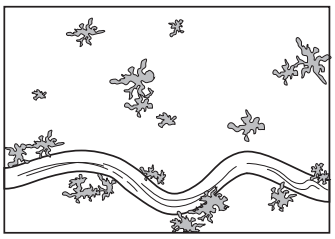


Continuous



Patchy distribution

Low level infestations



Well spread

Weeds at the site

Location	**Level of infestation** Heavy Moderate Low	**Distribution of weeds** Continuous (C) Patchy (P) Scattered (S) Widespread (W)	**Possible causes of infestation:** 1. Natural factors 2. Soil disturbance/nutrients 3. Stock damage/tracking 4. Water availability – drainage line/drain or channel 5. Dumped rubbish/garden plants 6. Urban influences
Aquatic weeds			
Bank (from water to top)			
Along the top of the bank to 2 metres			
Riparian width – 2 metres to a maximum of 30 metres			

Comments:.....

.....

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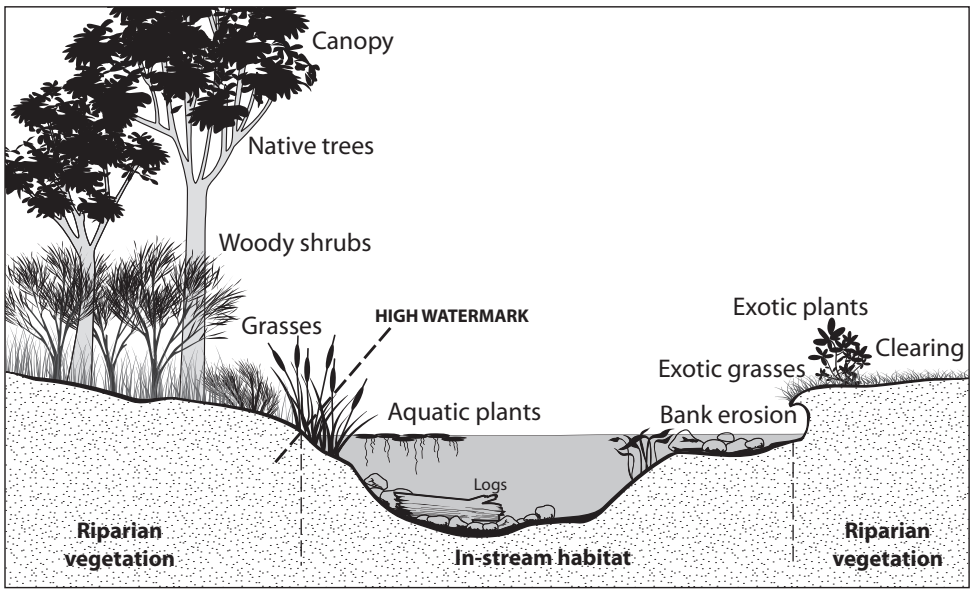
8.6 Habitat awareness assessment

Riparian vegetation



Habitats at the waterway

Bank vegetation: The trees, shrubs and grasses actually growing on the bank. The canopy is the overhanging tree cover. This vegetation provides food and shelter for aquatic organisms.

Vegetation from top of bank to edge of the riparian zone: This is an area up to about 30 m wide and can include vegetation such as trees, shrubs and grasses.

In-stream cover: In-stream cover includes overhanging banks and aquatic vegetation, snags, fallen trees, logs and rocks. Streams with a rich diversity of in-stream cover allow fish and macroinvertebrates to shelter from the current, feed and reproduce. Aquatic plants provide food and oxygen, and protruding snags provide roosting and preening sites for birds.

Bank erosion and stability: Streams naturally erode on the outside of bends (meanders) and deposit sediment on the inside of bends. However, changes in nearby land uses can cause a stream to become unstable, resulting in continuous erosion along its channel. You may find steep-walled gullies, bank collapse, slumping and hanging roots from riparian vegetation. If the stream has been channelled or stabilised with concrete banks, it will obviously be stable with little erosion, but it will have little or no vegetation cover and few of the habitats needed by macroinvertebrates to live.

8.7 How to carry out a habitat awareness survey

(Refer to the habitat descriptions in Section 8.6, the habitat rating table below and the habitat survey: field assessment in Section 8.8.)

1. Survey both sides of the stream for approximately 100 m and extending out from the water by 40 m for a medium/large size waterway or 10–20 m for a small stream.
2. For each feature in the stream habitat rating table, circle the category (excellent, good, fair, poor or very poor) that is most like your habitat.
3. Record the score for each feature in the totals row.
4. For an overall assessment of the site, add up the scores to obtain a total score for your stream habitat.

> ***i*** **Note: Bank and verge vegetation and in-stream cover are more important in determining the health of the habitat than either bank erosion and stability or riffles, pools and bends.**

HABITAT RATING RECORDING SHEET

Date: ..... Time: ..... Name: .....

Site name: .....

Position in catchment: ☐ upper ☐ middle ☐ lower ☐ estuary

Type of water body: *(tick box)*

☐ wetland ☐ lake/dam ☐ drain ☐ estuary

☐ creek/stream ☐ river ☐ irrigation channel ☐ spring

STREAM HABITAT RATING

Circle the appropriate score for each part of the habitat below:

Habitat rating	Bank vegetation	Top of bank to edge of riparian vegetation	In-stream cover	Erosion and stability	Pools, riffles and bends
Excellent	10	10	10	5	5
Good	8	8	8	4	4
Fair	6	6	6	3	3
Poor	4	4	4	2	2
Very poor	2	2	2	1	1
TOTALS					

Total score (all columns) ..... Stream habitat rating .....

Compare the total score with the range of total scores below to find a description of the general condition of your stream habitat.

Total score	Rating	Condition
36–40	Excellent	Site in natural or virtually natural condition; excellent habitat condition.
29–35	Good	Some alteration from natural state; good habitat condition.
20–28	Fair	Significant alterations from the natural state but still offering moderate habitat; stable.
12–19	Poor	Significant alterations from the natural state with reduced habitat values. May have moderate to severe erosion or sedimentation problems.
8–11	Very poor	Very degraded, often with severe sedimentation and erosion problems.

8.8 Habitat survey: field assessment

Excellent	Good	Fair	Poor	Very poor
Bank vegetation				
10	**8**	**6**	**4**	**2**
Mainly undisturbed native vegetation. No signs of alteration	Mainly native vegetation. Little disturbance or no signs of recent disturbance	Medium cover, mixed native and introduced or one side cleared and one side undisturbed	Introduced ground cover, little native under- or over-storey. Mainly introduced vegetation	Introduced ground cover with lots of bare ground, occasional tree. Also includes sites with concrete-lined channels
Vegetation from the top of the bank to the edge of the riparian zone (or 30 metres)				
10	**8**	**6**	**4**	**2**
Mainly undisturbed native vegetation on both sides of the stream. Riparian width more than 30 metres	Well-vegetated wide verge corridor. Mainly undisturbed native vegetation on both sides of the stream. Some introduced or reduced cover of native vegetation	Wide corridor of mixed natives and exotics OR one side cleared and the other a wide corridor of native vegetation	Very narrow corridor of native vegetation or introduced vegetation	Bare cover or introduced cover such as pasture-land

Excellent	Good	Fair	Poor	Very poor
In-stream cover				
10	**8**	**6**	**4**	**2**
Abundant cover. Frequent snags, logs or boulders with extensive areas of in-stream vegetation and overhanging bank vegetation	A good coverage of snags, logs or boulders, with considerable area of in-stream and overhanging vegetation	Some snags or boulders present and/or occasional areas of in-stream or overhanging vegetation	Only slight cover. The stream is largely cleared, with occasional snags and very little in-stream or overhanging vegetation	No cover, no snags, boulders submerged, no overhanging vegetation. No undercut banks. Site may have lock or concrete lining
Bank erosion and stability				
5	**4**	**3**	**2**	**1**
Stable: no erosion or sedimentation evident. No undercutting of banks, usually gentle bank slopes, lower banks covered with root mat, grasses, reeds or shrubs	Only spot erosion, little undercutting of bank, good vegetation cover, usually gentle bank slopes, no significant change to bank structure	Localised erosion. Relatively good vegetation cove. No continuous damage to bank structure or vegetation	Significant active erosion evident, especially during high flows. Unstable excessive areas of bare banks, little vegetation cover	Extensive or almost continuous erosion. Over 50% banks have some form of erosion; very unstable with little vegetation cover
Riffles, pools and bends (flowing water only)				
5	**4**	**3**	**2**	**1**
Wide variety of habitats. Riffles and pools present	Good variety of habitats – riffle, pool or bend. Some variation in depth	Some variety of habitats – occasional riffle or bend. Some variation in depth	Only slight variety of habitats. All riffles and pools with slight variety of depth	Uniform habitat. Straight stream, all shallow riffles or pools, uniform depth, channelled stream; irrigation channel

Adapted from *National Technical Manual*, Module 3.

SECTION 9

Habitat: riparian condition assessment

This assessment is designed to monitor changes in riparian vegetation due to investment in on-ground works. The assessment can be uploaded to the Waterwatch online database and will provide information about riparian areas that can be related to water quality testing and macroinvertebrate sampling at Waterwatch sites.

Included in this section: *Page*

Note: The *Senior Waterwatch Teachers' Guide* and the *Community/Land Manager Waterwatch Guide* contain further background information relating to the riparian condition assessment described in this section.

9.1 Riparian condition assessment: background information

(adapted from Sydney Catchment Authority GEM tool)

Riparian vegetation provides a surrogate measure for water quality when 'water quality' refers more broadly to 'river health'. This assessment provides a tool to assess changes in the riparian condition that impact on water quality, biodiversity and river health outcomes. A high level of risk means that the condition of the riparian zone may be detrimental to river health. A low level of human disturbance or a well managed site may have a very low level of risk to these outcomes.

Management of riparian areas aims to reduce the level of risk by improving the condition of the riparian zone directly or indirectly. It can be used to target management or to monitor changes due to previous management actions.

Very high	High	Moderate	Low	Very low

← Risk level →

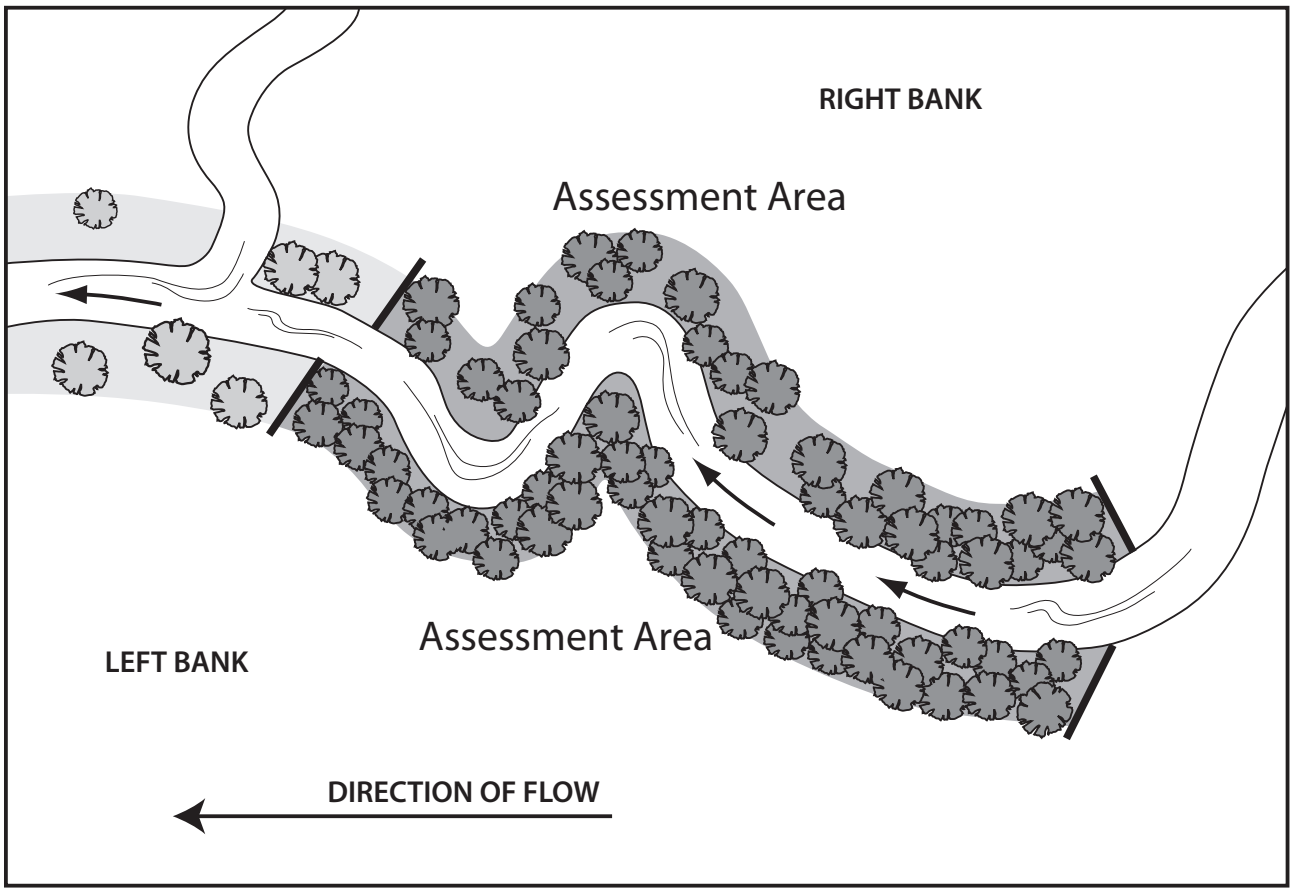
9.2 Setting up a riparian condition assessment

1. Site selection

Select a homogeneous length of stream where the attributes to be assessed are similar along the length.

When there is a clear change in one or more of the assessment criteria, a new assessment should be completed. The site length is determined by the length of the homogeneous unit and may be 100 metres or more than 1 kilometre,

The assessment can be conducted on one bank or both banks as each bank is scored separately. However, there may be more than one assessment required on each bank along the same length. Make sure that a new assessment is conducted with each change in the assessment attributes.



2. Parts of the assessment

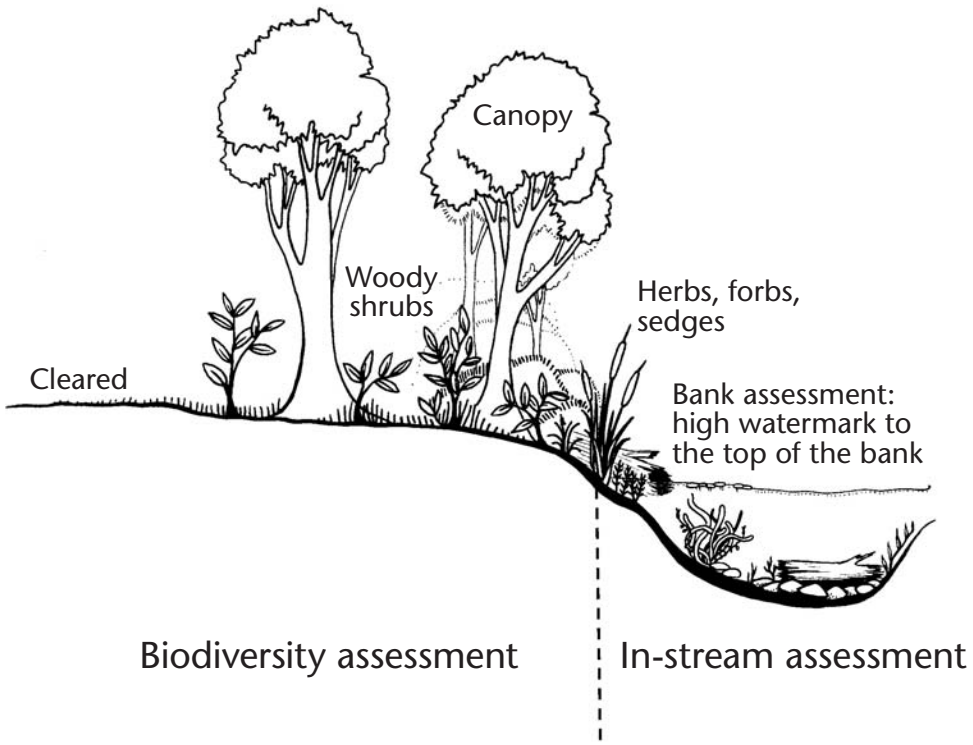
Identify key components of the assessment

There are five aspects of the field assessment:

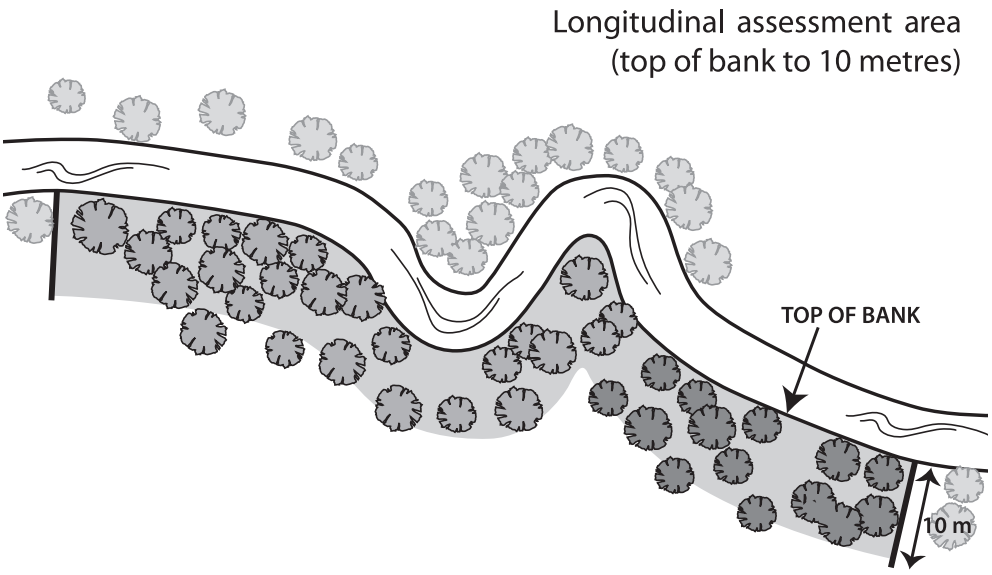
1. **Biodiversity/habitat assessment** is conducted from the top of the bank to the edge of the riparian zone or to a maximum width of 50 metres.

2. **Water quality assessment** of the buffer zone from the top of the bank to a width of 10 metres from the stream.

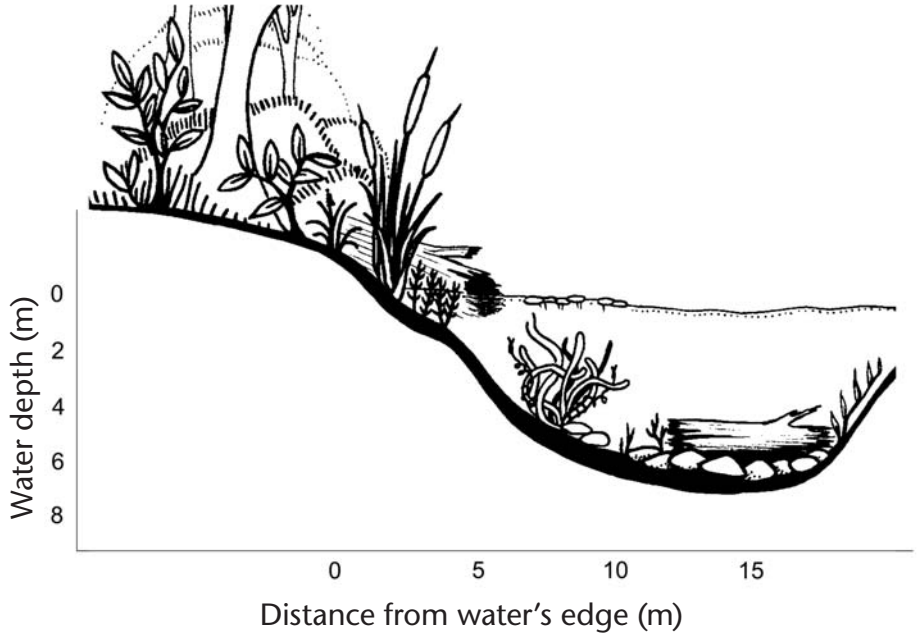
Assessment sites



3. Longitudinal assessment of riparian continuity and overhanging vegetation. An assessment along the top of the bank to a width of approximately 2 metres.



4. Bank assessment from the high watermark of the stream to the top of the bank OR **gully assessment** from the base of the gully to the top of the bank.



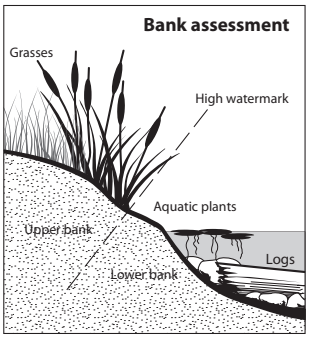
5. In-stream assessment of logs/snags and organic matter and sediment deposition within the channel.

3. Site features

Identify the left and right banks

> **Tip: To determine the left and right bank, face downstream.**

Identify the top of the bank

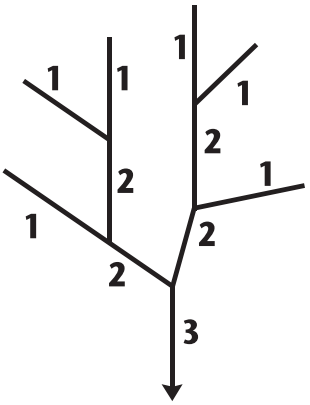


The top of the bank is the point of transition from the bank and the adjacent flat land. If banks have a stepped appearance, the top of the bank is the highest part of the highest step.

Identify the high watermark

The bank commences at the high watermark and extends to the top of the bank.

Identify the stream order



Stream order is determined using the Strahler system, which starts with a 1st order stream at the top of the stream network. It is usually a small non-perennial stream. Larger perennial streams usually have a stream order ≥3.

The stream order affects the width of the riparian zone required to provide an adequate stream buffer and the contribution of bank vegetation to the functioning of aquatic ecosystems, such as the amount of shade, temperature, light and organic matter provided within the stream.

9.3 Conducting a riparian condition assessment

Equipment:

riparian assessment guide

field assessment recording sheets

regional field guides such as:

- **native trees/shrubs in your region**
- **exotic species/weeds**
- **water plants**

clipboard and pen

hand-held global positioning system (GPS) or map to identify the position of the site

measuring tape/string

quadrat (optional)

camera

Biodiversity and habitat assessment

Step 1: Measure the width of riparian vegetation

Measure the width of the vegetation on the bank by running a tape measure from the high water mark to the first gap of >50 metres in the canopy.

The measurement will be in metres up to a maximum width of 50 metres.

Classify the riparian width into one of the three categories below:

Width of riparian zone <10 m
Width of riparian zone 10–30 m
Width of riparian zone >30 m

Step 2: Assess linear continuity

1. Observe the linear continuity of undisturbed native vegetation along the streambank.

2. Rate the degree of continuity along the banks by the number of gaps in tree cover or bare areas according to the following descriptions:

Riparian vegetation is highly fragmented* (or absent)
Continuous cover or minor gaps >50 metres
Continuous cover or minor gaps <50 metres

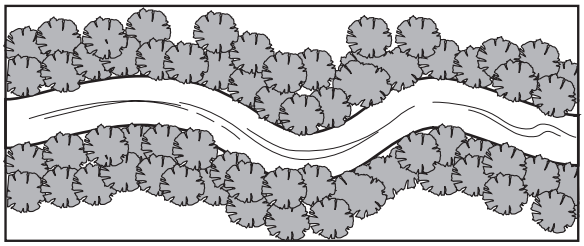
*When vegetation is not continuous, it can be described as fragmented. This includes riparian areas with a few isolated trees or clumps of native vegetation that may be widely separated.

If the vegetation is patchy, or has numerous grassy or bare breaks >50 metres, it lacks continuity and will be a high risk to biodiversity and water quality outcomes.

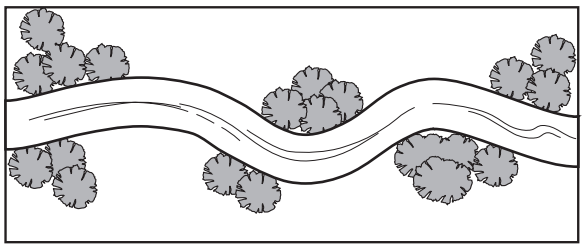
If a bank has a well-vegetated riparian zone that has a continuous cover along its length, it will provide a low risk to biodiversity and water quality outcomes.

Continuity of riparian vegetation

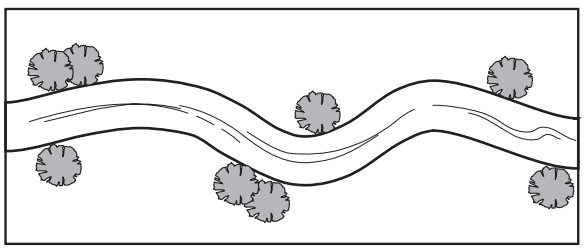
Continuous cover



Cover with gaps greater than 50 metres



Fragmented cover



Step 3: Assess native riparian vegetation cover

This assessment is conducted from the top of the bank to the width of the riparian zone to a maximum of 50 metres.

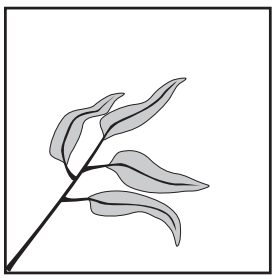
Use a native plant guide for your region to identify native trees, shrubs and grasses within your area.

Estimate the % cover of native vegetation or foliage density. This is the imaginary shadow cast by native vegetation.

Assess the cover of native species using the following criteria:

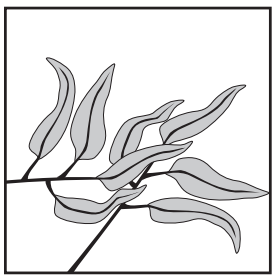
1. Canopy (% cover) of native trees >5 m tall by looking vertically upwards.

Zero to low presence of native trees (<30% canopy)
Moderate to high density of native trees (30–60% canopy cover)
High density of native trees (>60% canopy cover)



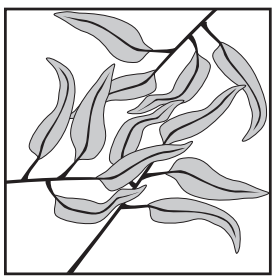
<5%

60–70%



5–30%

70–90%



30–60%

>90%

2. Understorey (% cover) of plants 1–5 m tall (mostly shrubs and small trees but also tall grasses) by looking vertically upwards.

Zero to low presence (<5% ground area)
Low to moderate presence (5–30% ground area)
Moderate to high density of native understorey (>30% ground area)

3. Ground cover (% cover) of plants <1 m tall (grasses, herbs, small trees and shrubs) looking vertically downwards.

Zero to low presence (<30% ground cover)
Moderate to low presence (30–60% ground cover)
High density (>60% ground cover)

Tip: As only native vegetation cover is required, estimate the total cover and deduct the amount of cover provided by non-native species.

For ground cover, estimate the % cover including leaf litter. A quadrat may assist with your assessment.

Step 4: Assess native species regeneration

Identify the dominant native vegetation at the site and any regeneration of these species and other native vegetation (seedlings, new shoots and grasses).

Methods of assessing native regeneration

1. Visually assess riparian regeneration by walking through the riparian zone.

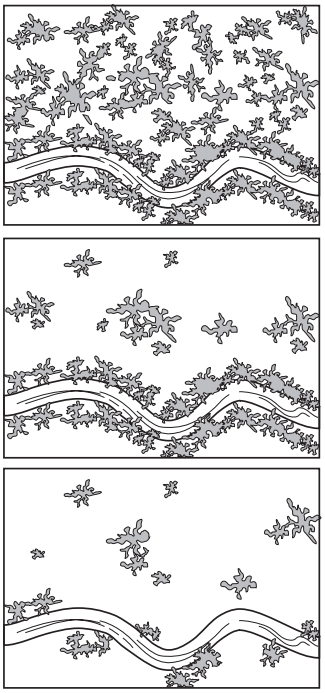
2. Assess the extent of native species regeneration based on the following criteria:

Nil or very limited native seedlings (<1%); uniform stem size
Scattered native seedlings (1–5%); some variety of canopy stem size
Abundant native seedlings (>5%); variety of canopy stem sizes

Step 5: Exotic species/weeds

Use a weed identification guide to assist with this assessment.

Assess the level of infestation across the whole riparian zone. This may include a combination of exotic trees, shrubs and grasses such as willows, blackberries and exotic grass species.



Rate the level of infestation according to the following criteria:

Riparian corridor heavily infested with exotic species/weeds
Riparian corridor moderately to heavily infested with exotic species/weeds
Riparian corridor with a low level of exotic species/weeds

9.4 In-stream habitat assessment

Conduct a visual assessment of overhanging native vegetation on the banks and in-stream features. This is best conducted from the opposite bank.

Step 1: Assess overhanging bank vegetation

Conduct a longitudinal assessment of overhanging bank vegetation based on the descriptions below:

Little or no overhanging canopy of native trees and shrubs
Moderate to good overhanging canopy of native trees and shrubs
Abundant overhanging canopy of native trees and shrubs

Note: Assess the cover relative to an undisturbed site in your area.

Overhanging vegetation of >30% of the bank would usually provide good cover.

Step 2: Assess recruitment of logs/snags and organic matter

Conduct a visual assessment of logs, snags and organic matter within the channel.

1. Make observations of the character of woody debris within the stream channel.

2. Distinguish between new and 'seasoned' logs.

3. Make an assessment of the recruitment of logs/snags and organic matter based on the criteria below:

Little or no new logs/organic matter; signs of habitat removal or replacement
Reasonable mix of newly fallen logs, snags and other plant matter
Wide variety of new and 'seasoned' logs/snags

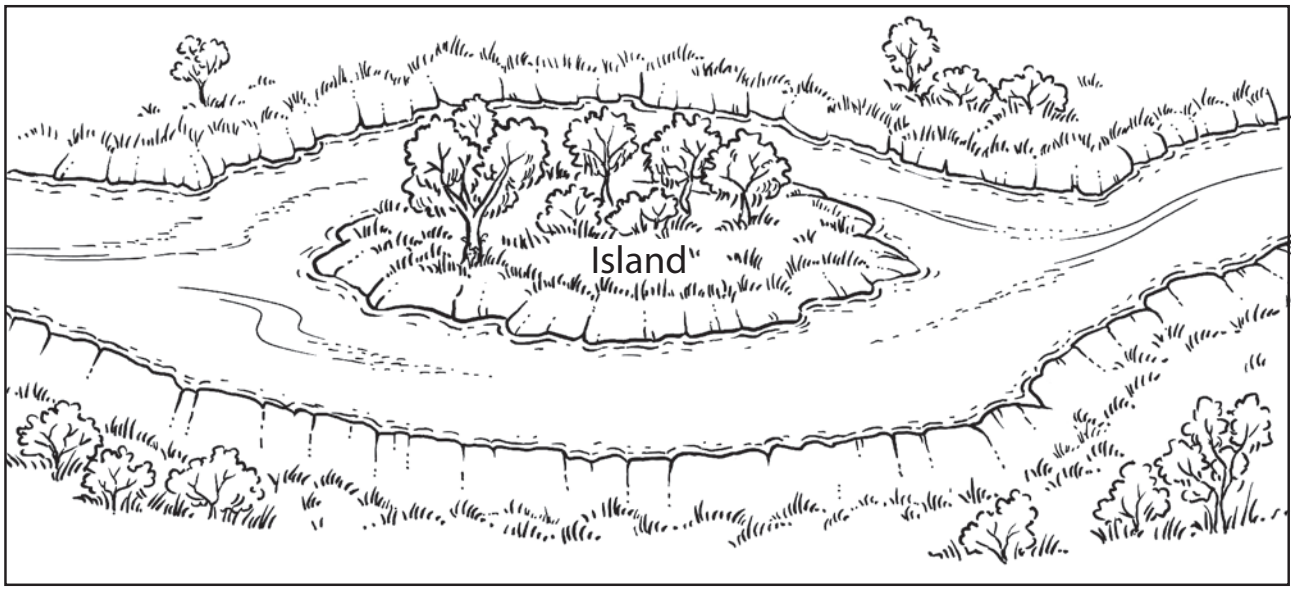
Step 3: Assess sediment deposition within the channel

The build up of mobile sediments in stream can reduce habitat diversity and impact on aquatic life.

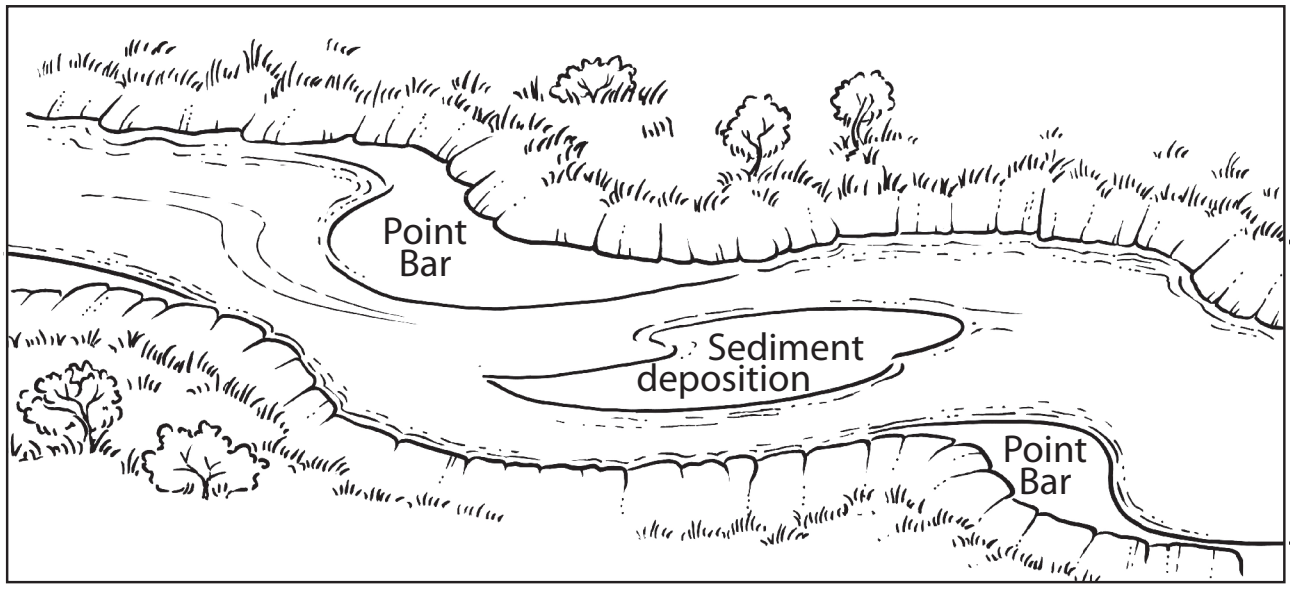
Conduct a visual assessment of sediment build up within the channel.

1. Observe the build-up of sediments within the channel.

2. Identify in-stream features formed by deposition such as islands and point bars or the deposition of sediment in pools.



Features formed by deposition: islands



Features formed by deposition: point bars and sediment deposition

3. Estimate the % of the streambed covered with mobile sediments using the following criteria:

A large % of the bed is covered with mobile fine sediment that smothers stones/cobbles
Significant build-up of gravel, sand or fine sediment on bends and at obstructions
Some deposits of sand, gravel and silt in pools, on bars and bends
Little sediment deposited with no reduction in channel depth

Note: To assist with the criteria above, the following percentages can be used as a guide:

Bed covered with mobile sediment	
Large	>80%
Significant	50–80%
Some	20–50%
Little	<20%

Water quality: filter for overland flow (runoff)

Step 1: Assess stock access to streams

These assessments are conducted from the top of the bank to the channel.

Make observations about stock access to the stream.

Consider:

1. evidence of stock near the site on adjacent land

2. the presence of fences to restrict stock

3. the extent of damage caused by stock, including tree ring-barking, vegetation trampling, soil compaction and track formation

4. tracks and animal droppings at the site.

Based on these observations, rate stock access to streams as:

Heavy – significant evidence of stock access and damage, including severe stock tracking, loss of vegetation on banks and no fencing to restrict stock

Moderate to light – minor tracking on banks and some loss of vegetation. Some fencing and alternative watering points for stock may be evident

Stock excluded – streambanks fenced with no evidence of stock tracking

No stock – no evidence of stock at the site.

Identify the type of stock as:

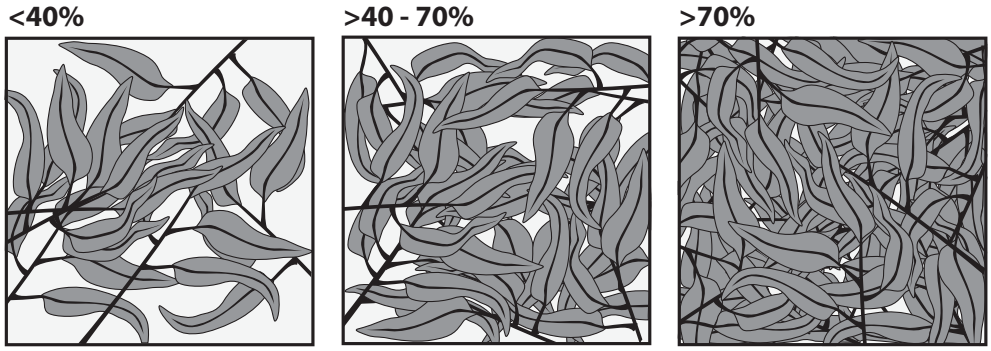
- cattle including dairy cattle
- other stock such as sheep and horses
- no stock.

Step 2: Assess percentage of groundcover

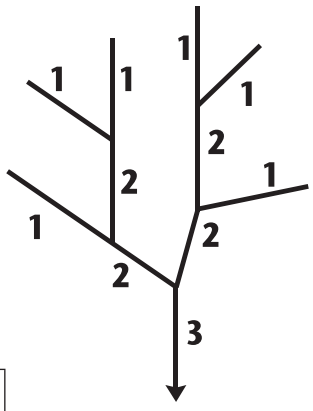
This assessment is conducted from the top of the bank to a width of 10 metres.

Conduct an assessment to determine the % groundcover. Look vertically downwards and use the visual guide below to assist:

<40% native groundcover
40–70% native groundcover
>70% native groundcover



Step 3: Determine the stream order at your site



1st and 2nd order – usually small non-perennial streams

3rd order and higher – usually large perennial streams

Step 4: Classify local land use according to the following criteria

High impact land use – cropping, high density grazing
Low impact land use
No human activity – native bushland

Water quality: filter for subsurface flow

This assessment is conducted from the top of the bank to a width of 10 metres.

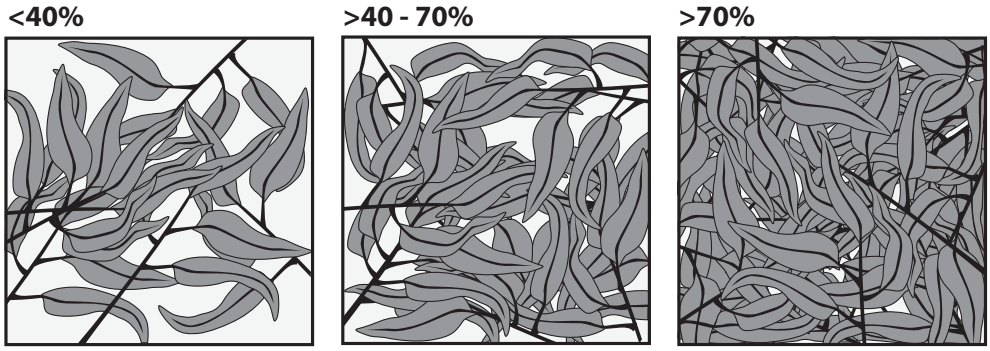
Step 1: Assess riparian root density

The roots of plants take up excess moisture and reduce the amount of subsurface flow.

Canopy cover % can be used as a surrogate measure for root density.

Look vertically upwards and estimate the % of the sky blocked out by the canopy. Use the visual guide to assist:

<40% native canopy cover
40-70% native canopy cover
>70% native canopy cover



Step 2: Determine the stream order at your site

1st or 2nd order stream – usually small non perennial

3rd order and higher – usually large perennial streams

Step 3: Classify local land use according to the following criteria

High impact land use – such as unsewered residential, irrigated cropping or horticulture
Low impact land use
No human use – native bushland

Note that high impact land use may vary according to the location of the site. Examples are unsewered residential, intensive agricultural practices, tourism, etc.

9.5 Bank stability assessment

Conduct a streambank OR gully assessment. If both exist at your site, a separate assessment sheet should be completed for each.

Streambank assessment

Identify the parts of the bank: watermark, lower bank, upper bank and top of the bank (extending up to 2 m on the flat land).

Step 1: Estimate the height of the bank

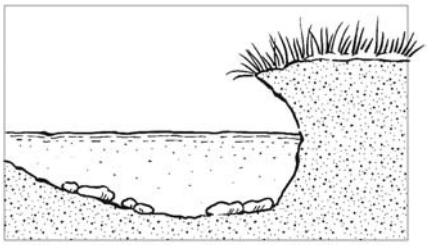
Steep banks have a higher risk of erosion. Classify the bank as:

- less than 2 metres
- greater than 2 metres.

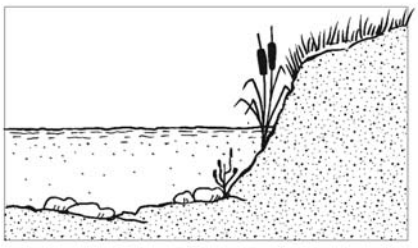
Step 2: Assess the level of bank erosion

Observe and rate the severity of erosion occurring on the bank. Banks that are actively eroding will have little vegetation and there may be signs of bank slumping, undercut banks and erosion of soil from banks and around tree roots.

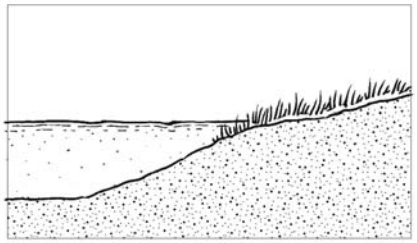
Banks with highly erodable or dispersive soils are more likely to erode when vegetation is absent.



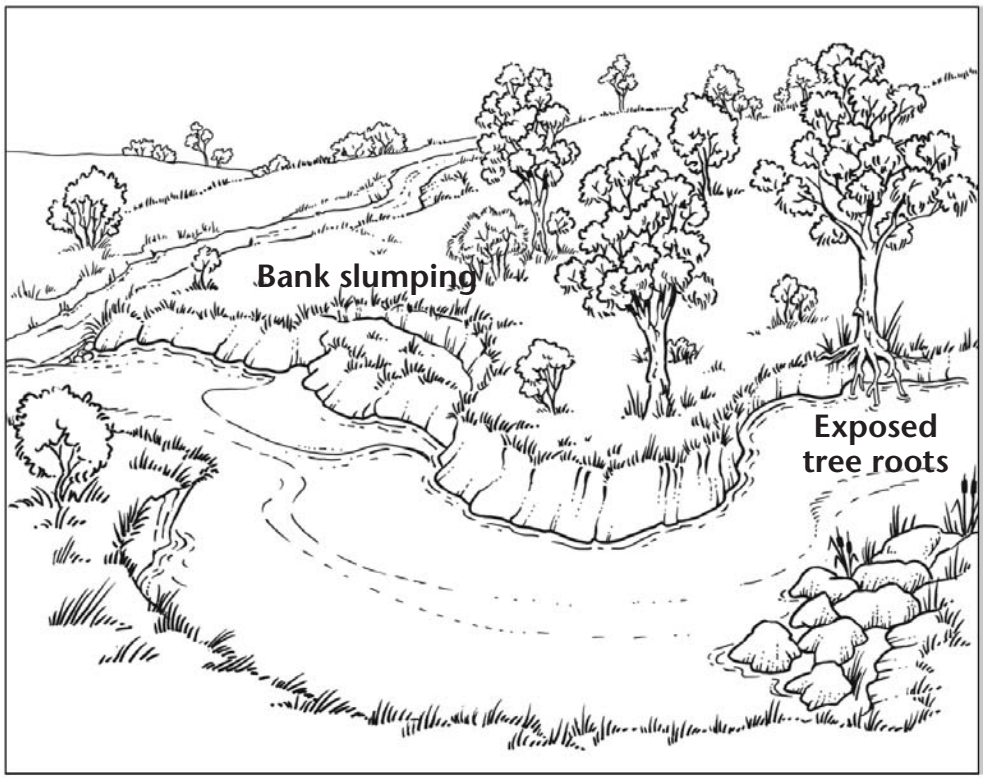
Undercut



Steeply sloping



Gradually sloping



Step 3: Assess vegetation on banks

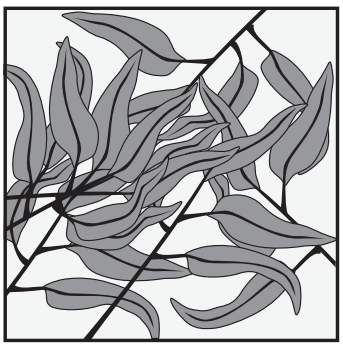
Stable banks may be more rounded and covered with vegetation.

Assess the vegetation on the banks by using the following measures:

1. **The root density of bank vegetation** – this can be measured as % cover: 100% cover provides a high root density and reduces the risk of streambank erosion. Record your result based on the following description:

<60% of area with deep rooting plants/trees
>60% with deep rooting plants or trees

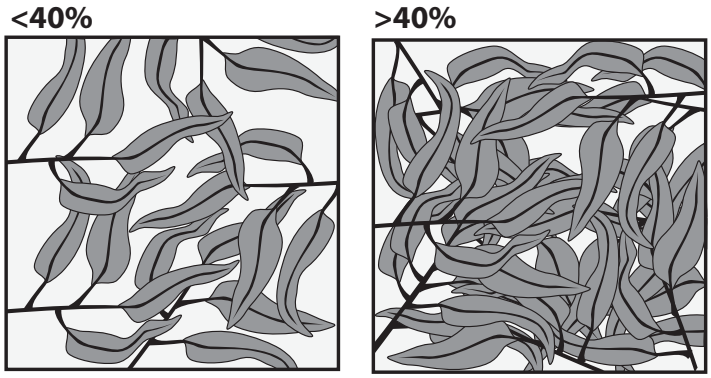
<60%



>60%

2. Ground cover on banks – estimate the % cover of grasses, herbs and forbs on the bank and select the description or that describes ground cover at your site. A quadrat may assist.

<40% of the ground covered by well-binding ground cover
>40% of the ground covered by well-binding ground cover



Step 4: Assess channel flow characteristics

Sites that are below a major dam may be subject to high volume water transfers.

These transfers can erode banks.

Record whether your site is affected by high volume transfers.

Step 5: Assess stock tracking on banks

Observe land use and stock pressure on adjacent land. Damage by stock includes tree ring-barking, vegetation trampling, soil compaction and track formation.

Tracking is used in this assessment as the measure of stock impact on banks.

High level – obvious animal track with compacted soil and little or no vegetation cover

Light tracking – faint trails or no animal impact seen.

Record the level of stock tracking.

9.6 Gully stabilisation assessment

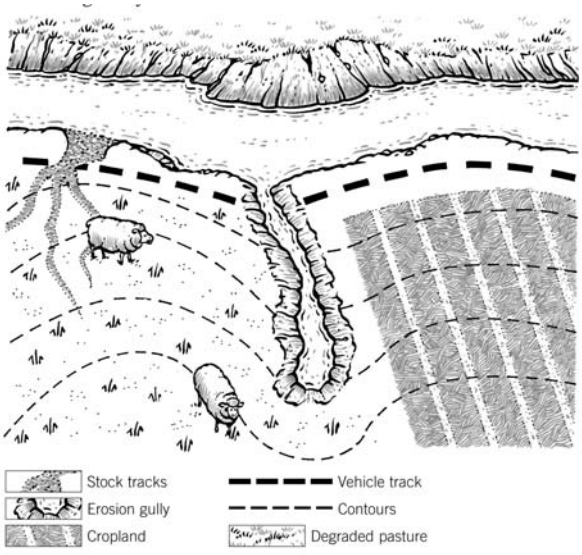
This assessment should be undertaken from the base of the gully to the top of the bank.

Active gullies are unstable and may be increasing in size. Stable or natural gullies may have vegetation, rocks or other structures supporting their walls and head (top of gully).

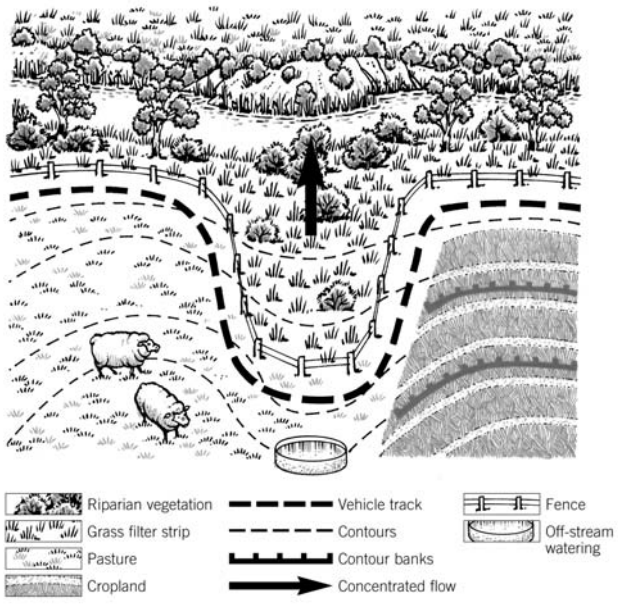
Conduct a visual assessment based on the following criteria:

Severe active head cutting and lateral extension – deep gullies, rills and tunnels
Low level active erosion and head cutting – minor rills only are present
Stable gully – no rill, tunnel or gully activity

Note: Small alluvial fan deposits may be found where an active gully discharges into the channel.



Degraded stream



Managed stream with riparian filter strip

Source: Land and Water Australia
Illustrations: Paul Lennon

9.7 Summary field guide: conducting a riparian assessment

Note: Refer to Sections 9.1 to 9.5 above for a detailed outline of the purpose and measures included in this assessment.

1. Select a **homogenous length of stream**. This means that there is little variation in riparian structure and stream characteristics such as stream width and continuity.

2. The left and right bank will be determined by **looking downstream**.

3. Complete the site features table as this has important information about your site and the assessment.

4. Complete the riparian condition assessment sheet for each measure, matching the descriptions with observations/ investigations at the site.

5. Upload the information about your site to the Waterwatch online database www.waterwatch.nsw.gov.au

6. Match the score to the level of risk for that measure.

Note: There is no overall assessment for the site.

Completing the field recording sheet

There are field recording sheets for each outcome: biodiversity, water quality and bank stability. Select all OR the most relevant outcomes and complete the assessment using the following steps:

Step 1: Circle the scores that best describe your assessment at the site.

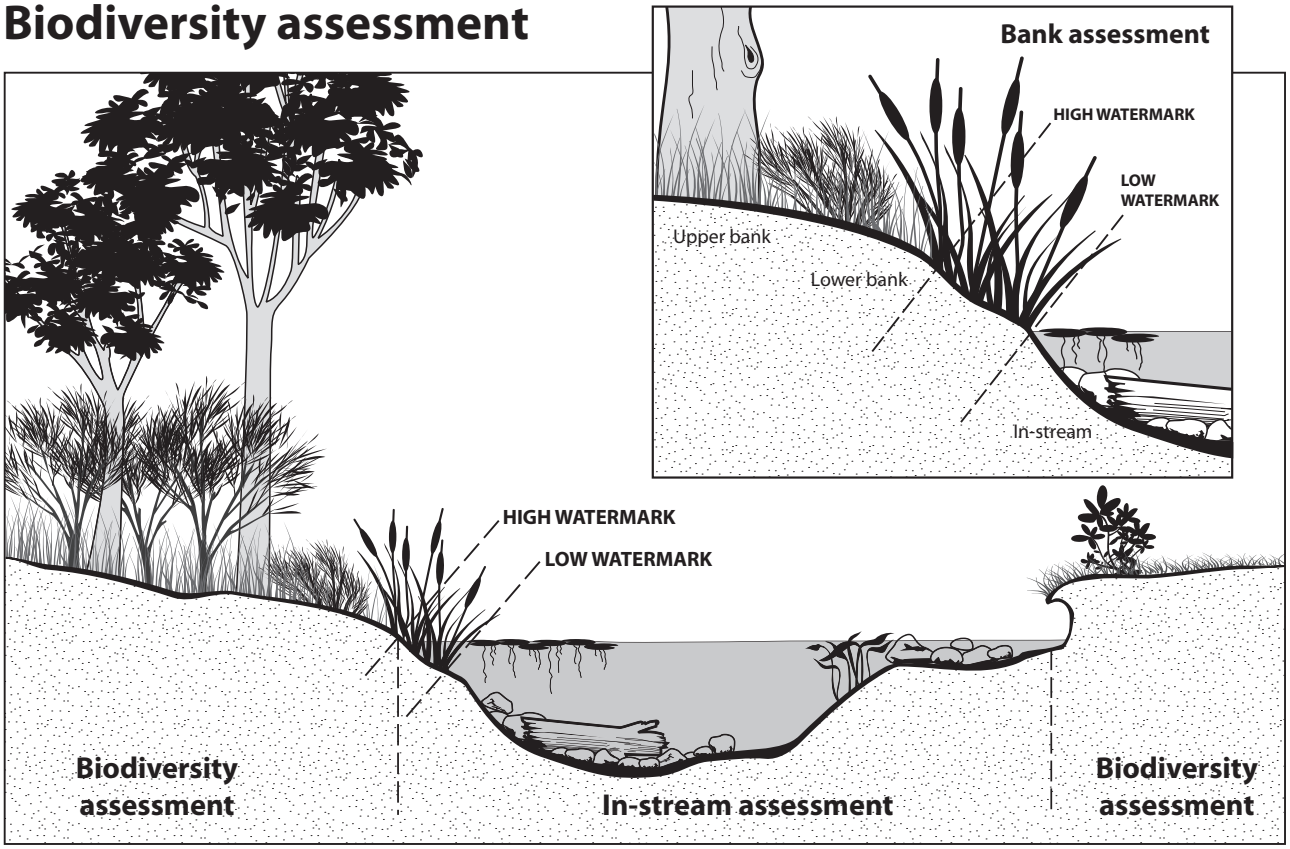
Step 2: Add the scores you have circled for each outcome.

Step 3: Transfer the total scores into the table, showing the level of risk on the following page of your assessment.

Step 4: Determine the level of risk for each criteria assessed.

Assessment locations

Biodiversity assessment



9.8 Riparian condition assessment

Date: ..... Start time: .....

Site name: .....

Length of bank assessed (metres): Left bank .....

Right bank .....

Left bank starting point ..... End point .....

Right bank starting point ..... End point .....

Use GPS coordinates or a topographic map and a clear description of start and end points.

Number of volunteers: ..... Time taken: .....

Complete the riparian condition assessment sheets.

Identifying the level of risk

After completing the site assessments, determine the level of risk for **each** criterion by following the steps below:

1. Add your total score for each assessment to the 'Your total score' column in the table. The left and right banks appear underneath each other in the table.

2. Circle the risk category for that score for each bank.

Bank	Level of risk	Your total score	Very high	High	Moder-ate	Low	Very low
Biodiversity							
Left	Width and connectivity		7	5–6	4	2–3	1
Right	Width and connectivity		7	5–6	4	2–3	1
Left	Riparian structure		9–11	7–8	4–6	2–3	1
Right	Riparian structure		9–11	7–8	4–6	2–3	1
Left	In-stream habitat		7	5–6	4	2–3	1
Right	In-stream habitat		7	5–6	4	2–3	1

Bank	Level of risk	Your total score	Very high	High	Moder-ate	Low	Very low
Water quality							
Left	Stock access		6	5	4	3	1
Right	Stock access		6	5	4	3	1
Left	Overland flow		7	5–6	4	2–3	1
Right	Overland flow		7	5–6	4	2–3	1
Left	Subsurface flow		7	5–6	4	2–3	1
Right	Subsurface flow		7	5–6	4	2–3	1
Bank stability							
Left	Stream bank erosion		10	8	6–7	2–5	1
Right	Stream bank erosion		10	8	6–7	2–5	1
Left	Gully erosion		7	5–6	4	2–3	1
Right	Gully erosion		7	5–6	4	2–3	1

Note: Some columns may have a maximum score.

3. Compare your results to the description of the level of risk in the table below:

Level of risk	Site features
Very high	The site is in a very poor condition and will be unable to achieve the desired outcome in the current state. Identifying the key areas of risk and implementing a management strategy is highly recommended.
High	The site is in a poor condition and will be unable to achieve the desired outcome in the current state. Identifying the key areas of risk and implementing a management strategy is highly recommended.
Moderate	This site is in fair condition. Some measures at this site may require management to achieve the desired outcome.
Low	This site is in good condition. Maintain existing management and target areas of identified risk to maintain and enhance this low risk environment.
Very low	This site is in very good condition and may be a well-managed site or an area of native bushland. Maintain existing management or protect the site to continue achieving the desired outcome.

9.9 Riparian condition assessment: water quality outcomes

Complete the water quality assessment, which includes stock access, overland flow and subsurface flow objectives.
Circle the score that best describes your site features.

Stock access		**Banks**	
Criteria	**Measures**	**Left**	**Right**
Stock access	Evidence of heavy stock access	4	4
	Stock excluded (fenced)	2	2
	No stock	1	1
Type of stock	Cattle or dairy cows	2	2
	Cattle other than dairy cows	1	1
	No stock	0	0
Overland flow			
Riparian ground cover	<40% ground cover	5	5
	40–70% ground cover	2	2
	>70% ground cover	1	1
Stream order	1st and 2nd	1	1
	3rd +	0	0
Adjacent land use	High impact land use, e.g. cropping, high density grazing, urban land use	1	1
	Low impact land use	0	0
	No human use (native bushland)	0	0
Subsurface flow			
Riparian root density (% cover)	<40% cover native plants and trees	5	5
	40–70% cover native plants and trees	2	2
	>70% cover native plants and trees	1	1
Stream order	1st and 2nd order stream	1	1
	3rd order +	0	0
Adjacent land use	High impact land use, e.g. unsewered residential, irrigated cropping or horticulture	1	1
	Low impact land use	0	0
	No human use (native bushland)	0	0
TOTAL SCORE			

Note: The higher the total score, the higher the level of risk. High impact land use may vary according to the location of the site. These activities may increase the risk of groundwater movement and contamination. Examples are unsewered residential, intensive agricultural practices, tourism, etc.

9.10 Riparian condition assessment: biodiversity outcomes

Complete the biodiversity assessment for riparian vegetation and in-stream habitat. **Circle the score that best describes your site features.**

Riparian assessment

Riparian width and continuity		**Banks**	
Criteria	**Measures**	**Left**	**Right**
Riparian width and continuity	Width of riparian zone <10 metres	5	5
	Width of riparian zone 10–30 metres	2	2
	Width of riparian zone >30 metres	1	1
Riparian continuity	Riparian vegetation is highly fragmented or absent	2	2
	May contain one or more gaps >50 metres	1	1
	Continuous cover or minor gaps <50 metres	0	0
TOTAL SCORE			

Riparian vegetation structure		**Banks**	
Criteria	**Measures**	**Left**	**Right**
Riparian native canopy (% cover)	Zero to low presence (<30% canopy cover)	7	7
	Moderate to high density of native trees (30–60% canopy cover)	2	2
	High density of native trees (>60% canopy cover)	1	1
Riparian native understorey (% cover)	Zero to low presence of native understorey (<5% ground area)	1	1
	Low presence of native understorey (5–30% ground area)	1	1
	Moderate to high density of native understorey (>30% ground area)	0	0
Riparian native ground cover (% cover)	Zero to low presence of native ground cover (<30% ground area)	1	1
	Low to moderate density of native ground cover (30–60% ground area)	1	1
	Moderate to high presence of native ground cover (>60% ground area)	0	0

Riparian vegetation structure *continued*		Banks	
Criteria	**Measures**	**Left**	**Right**
Native vegetation regeneration (canopy stem size; number of native seedlings)	Nil or very limited native seedlings (<1%); uniform stem size	1	1
	Scattered native seedlings (1–5%); some variety of stem size	1	1
	Abundant native seedlings (>5%); variety of canopy stem sizes	0	0
Exotic species/ weeds	Riparian corridor moderately to heavily infested with exotic species/weeds	1	1
	Riparian corridor with low level of exotic species/weeds	0	0
TOTAL SCORE			

In-stream assessment

In-stream habitat		Banks	
Criteria	**Measures**	**Left**	**Right**
Overhanging native vegetation on banks	Little or no overhanging native vegetation on banks	5	5
	Moderate to good native vegetation overhanging banks	2	2
	Abundant native vegetation overhanging banks	1	1
Recruitment of logs/snags and organic matter	Little or no new logs/snags and organic matter; signs of habitat removal	1	1
	Reasonable mix of newly fallen and 'seasoned' logs/snags	1	1
	A wide variety of newly fallen and 'seasoned' logs/snags	0	0
Sediment deposition	A large percentage of the bed covered with mobile fine sediments	1	1
	Significant build-up of gravel, sand or fine sediment; build up on bends and at obstructions	1	1
	Some deposits of gravel, sand or fine sediment; some deposition in pools	1	1
	Little or no sediment deposition; no reduction in channel depth	0	0
TOTAL SCORE			

9.11 Riparian condition assessment: bank stability

Complete either the bank stability **or** gully stabilisation.
If both are present, use a separate assessment sheet.
Circle the score that best describes your site features.

Streambank stabilisation		**Banks**	
Criteria	**Measures**	**Left**	**Right**
Evidence of active erosion	Significant active erosion on banks >2 metres high	10	10
	Significant active erosion on banks <2 metres high	8	8
	Minor to moderate active erosion; bank slumping/ exposed tree roots	2	2
	Nil to minor active erosion	1	1
Bed lowering adjacent to/ downstream of site	Moderate active streambed erosion/channel deepening	3	3
	Minor active streambed erosion/channel deepening	1	1
	Little or no streambed erosion/channel deepening	0	0
Riparian root density (use % cover as the measure)	<60% of banks with deep rooting plants/trees	1	1
	>60% of banks with deep rooting plants/trees	0	0
Vegetation on banks (exclude litter)	<40% of the ground covered by well-binding ground cover	1	1
	>40% of the ground covered by well-binding ground cover	0	0
Water flow	Frequent large volume, high velocity flows due to water transfers	1	1
	No water transfers	0	0
Stock pressure	Moderate to high grazing pressure and/or moderate to heavy tracking on banks	1	1
	Stock exclusion or light tracking	0	0
TOTAL SCORE			

OR

Gully stabilisation		Banks	
Criteria	**Measures**	**Left**	**Right**
Evidence of active erosion	Significant active erosion/head cutting; deep channels/tunnels	5	5
	Low level active erosion; minor rills	2	2
	Little or no active erosion; banks stable	1	1
Bank vegetation cover (grass)	Poor (<40% cover)	1	1
	Good (>40% cover)	0	0
Stock pressure	Moderate to high grazing pressure or heavy stock tracking	1	1
	Low grazing pressure or light stock tracking	1	1
	Stock excluded/no stock	0	0
TOTAL SCORE			

9.12 Riparian condition identification sheet: water quality outcomes

Stock access

	
Very high to high risk: evidence of heavy stock access	Very low risk: stock excluded or fenced

Overland flow and subsurface flow

	
Very high to high risk: <40% ground cover	Very low risk: >70% ground cover
	
Very high risk: <40% native canopy cover (riparian root density)	Very low risk: >70% native canopy cover (riparian root density)

9.13 Riparian condition identification sheet: riparian vegetation cover

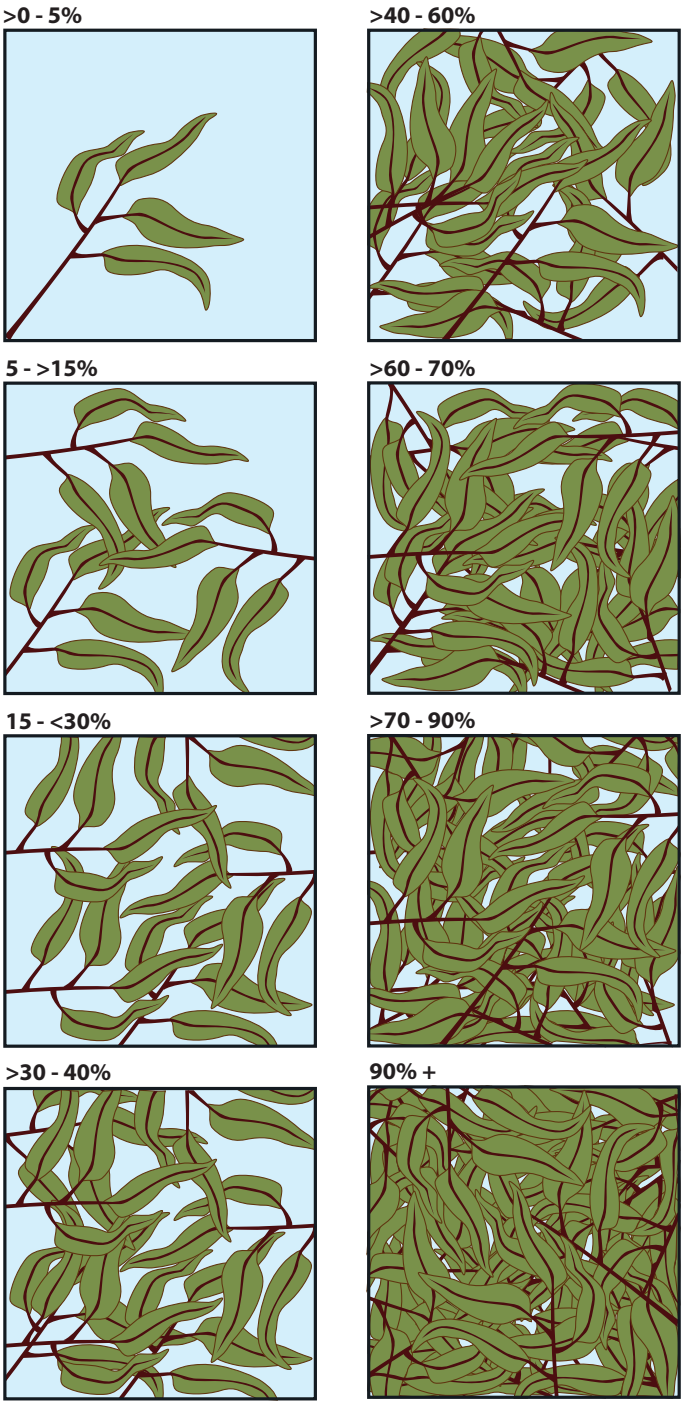
Riparian root density

Look upwards and estimate the % cover of native vegetation at the site.

Riparian ground cover

Look downwards and estimate the percentage cover including leaf litter (unless otherwise stated).

Riparian Vegetation Percentage Cover



9.14 Riparian condition identification sheet: biodiversity assessment

Riparian vegetation continuity

	
Very high risk: native riparian vegetation absent	Very high risk: native riparian vegetation highly fragmented
	
Moderate risk: cover may contain one or more gaps >50 metres	Very low risk: continuous cover or minor gaps

Vegetation structure: exotic species

	
Very high to moderate risk: riparian corridor moderately to heavily infested with exotic species/weeds	Very low risk: riparian corridor with low level of exotic species/weeds

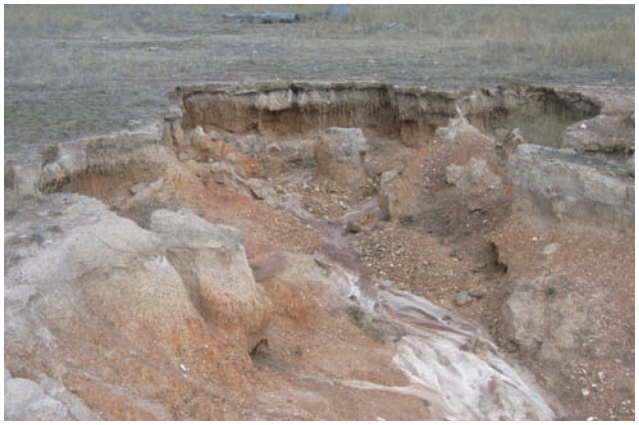
9.15 Riparian condition identification sheet: in-stream habitat assessment

	
Very high to high risk: little or no overhanging native vegetation; little recruitment of new logs, snags and organic matter	Very high to high risk: overhanging vegetation is mostly exotic species such as willows; new organic matter is derived mostly from these species
	
Moderate risk: some overhanging native vegetation; some recruitment of new logs, snags and organic matter	Low to very low risk: abundant overhanging native vegetation; a wide variety of logs, snags and organic matter
	
Very high to high risk: significant sediment build up within the channel	Moderate risk: deposits of sediment as islands and on bends and at obstructions
	
Low risk: some deposits of sand or fine sediment within the stream channel	Very low risk: little or no sediment deposits

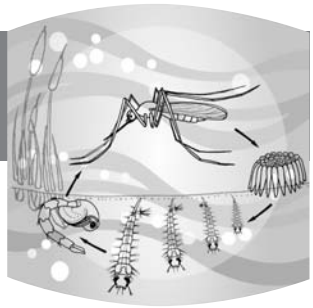
9.16 Riparian condition identification sheet: streambank stability assessment

	
Very high to high risk: active erosion on banks >2 metres	Very high to high risk: active erosion on banks <2 metres
	
Minor to moderate active erosion: bank slumping	Minor to moderate active erosion: exposed tree roots
	
Unstable banks: <40% banks covered by well-binding ground cover	Stable banks: >40% well-binding ground cover
	
High to moderate grazing pressure and stock tracking	Stock exclusion or light tracking

9.17 Riparian condition identification sheet: gully stabilisation assessment

	
Very high to high risk: significant active erosion and headcutting	Very high to high risk: active erosion tunnels
	
Very high to high risk: poor vegetation cover; <40% (grass) on banks	Very low to low risk: vegetation cover >40% (grass) on banks
	
High to moderate grazing pressure and stock tracking	Stock exclusion or light tracking

SECTION 10



Water bug (macroinvertebrate) survey

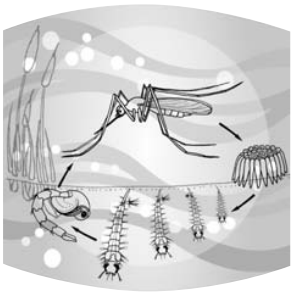
Water bugs, or macroinvertebrates, are small creatures with no backbone that can be seen with the naked eye. Different kinds of water bugs have different tolerances to pollution and can therefore provide an indication of the health of your waterway. A healthy waterway will have an abundance and wide diversity of macroinvertebrates.

Regular water bug surveys take place in Spring and Autumn. You can take part in these and use the results to ascertain the health of your creek or river and contribute valuable data on the changes in the health of our waterways.

Included in this section: *Page*

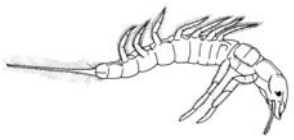
Note: The *Senior Waterwatch Teachers' Guide* and the *Community/Land Manager Waterwatch Guide* contain further background information relating to macroinvertebrates and their usefulness as an additional indicator of water quality.

10.1 Doing a water bug (macroinvertebrate) survey

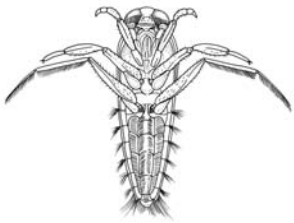


What are water bugs?

Water bugs, or aquatic macroinvertebrates, are small creatures that have no backbone and can be seen with the naked eye. They live all or part of their life in water, providing a food source for larger animals such as fish, frogs and birds. Macroinvertebrates include snails, beetles, dragonflies, yabbies and worms.

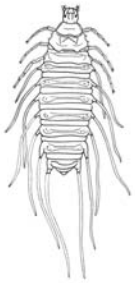


> **Note:** Macro = visible to the naked eye
> Invertebrate = animal without a backbone



Macroinvertebrate sampling can provide a rapid assessment of the condition of a site at a particular time. When compared to other locations, these studies can provide useful information about the health of the aquatic ecosystem.

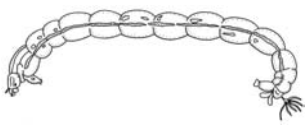
Designing a macroinvertebrate study

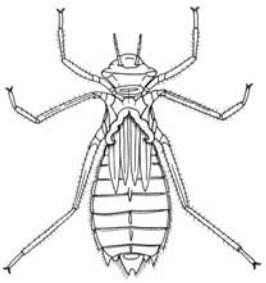


Step 1: Identify the sampling objectives

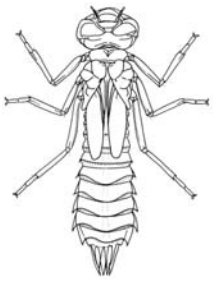
Identify the purpose of your study as this will determine sampling sites and methods. Some studies may be conducted to:

- gain a better understanding of the different types of macroinvertebrates
- compare the site with other sites in their natural condition
- estimate changes over time in the composition and abundance of water bugs
- compare changes in macroinvertebrates over time following management actions.





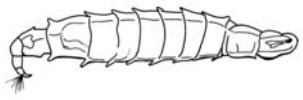
Step 2: Monitoring plan



Where in the catchment should I place my monitoring sites?

Select sites that meet the objectives of your study. This may involve the selection of more than one site if comparative studies are required.

Where should I sample in the stream?



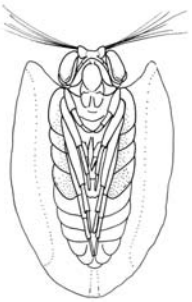
Within the stream, sample a range of habitats, including under stones, logs, fringing vegetation and pools and riffles.

Sample in roughly the same place each time you visit so that comparisons can be made between data collected at different times.

What equipment should I use?

Waterwatch prefers nets with a triangular frame and fine net dip bag. See tips for students at the end of this section.

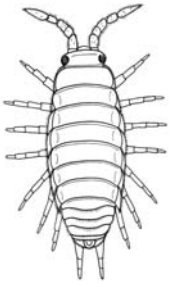
When should I sample?



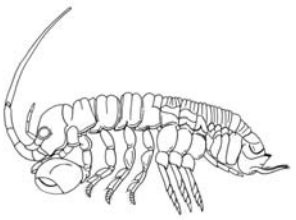
Sampling should occur twice a year, preferably in spring (October) and autumn (March).

Step 3: Type of sampling

There are two basic methods used to collect samples of water bugs: sweep sampling and kick sampling. Sweep sampling is generally done from the water's edge, while kick samples are taken from riffles. Refer to more detailed procedures below.



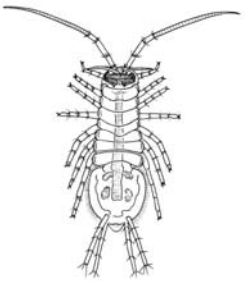
Collecting and identifying macroinvertebrates



Collecting a sample

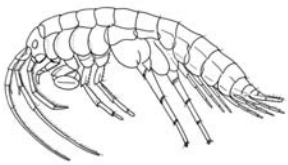
General procedure for both sweep and kick sampling

Time: 5–10 minutes

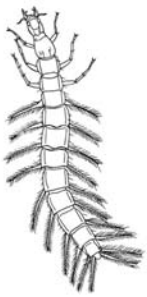


1. Pour clear stream water into a large white sorting tray to about 2 cm deep and put the tray close to the edge of the water.

Note: Where is it difficult to lie the tray flat at the water's edge, use a bucket and transfer the water into trays after sampling.

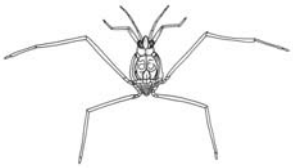


2. Use a short upward-sweeping motion to sweep the net through the stream.



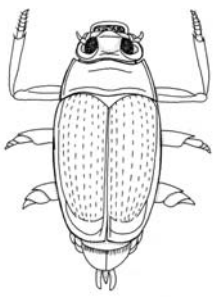
3. Stop regularly to transfer the macroinvertebrates gently into the tray. Turn the net inside out and wash its tip in the tray to transfer the bugs.

4. Rinse any mud or fine silt from your net. The sample should be free of sediment prior to sorting.



5. Spread the sample out in the tray and allow the water to settle so small macroinvertebrates can be seen.

> ***i*** **Testing tip: Make sure nets are disinfected with bleach and water between sampling events to prevent the transfer of sediment, seeds, bacteria, viruses or other unwanted materials between sites.**



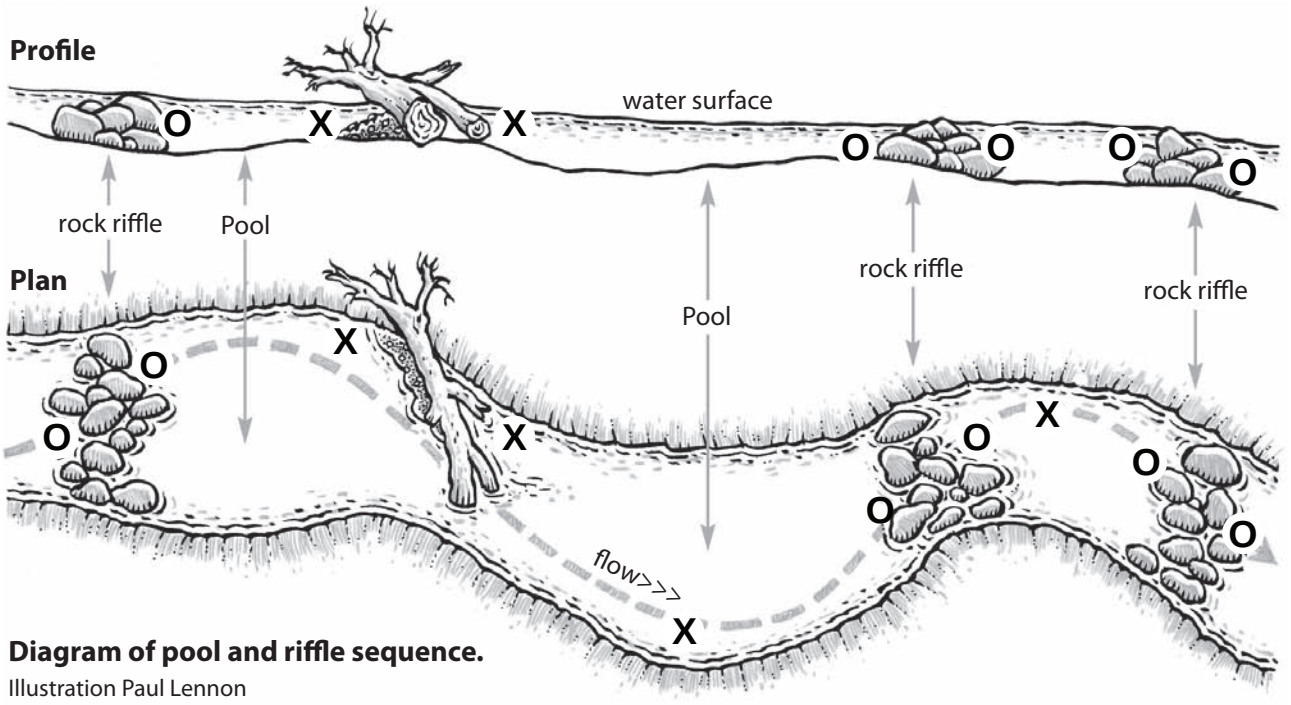


Diagram of pool and riffle sequence.
Illustration Paul Lennon

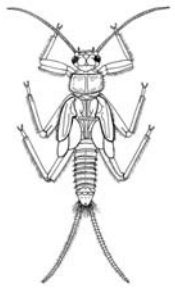
X sweep sampling **O** riffle sampling

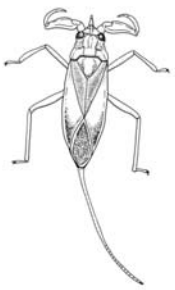
Procedure for sweep sampling

Sweep sampling can occur along the edge of the stream and should include a range of habitats such as under logs and tree roots, and in fringing vegetation. Sample the top, edge and bottom of the water along at least 10 metres of stream.

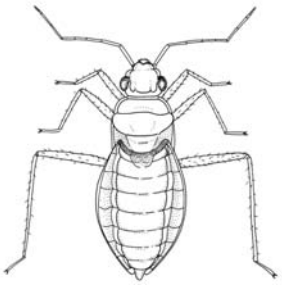
Procedure for kick sampling

In riffles, use a technique called kick sampling. Wearing rubber boots, stand in calf to knee deep water facing downstream. Hold the net in front of you with the opening facing upstream. Disturb the rocks underfoot by vigorously shuffling and kicking. The current will sweep dislodged macroinvertebrates into the net. Move slowly upstream while you do this to sample a 10 metre length of the streambed.





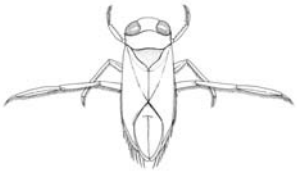
Sorting the sample



Time: 30–40 minutes

1. Observe the water bugs in the large white sorting tray.

> ***i*** **Testing tip: Aim to collect at least 50 macroinvertebrates per sampling area and as many types as possible.**
>
> **It is not possible to calculate the stream pollution index unless you have at least 50 macroinvertebrates.**

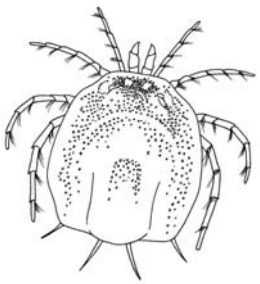


2. Each group should fill an ice block tray with a small amount of water.

3. Transfer bugs to the ice block trays using plastic spoons, pipettes and paint brushes.

4. Sort the macroinvertebrates into the cubes in the tray using a different cube for each type of bug.

Identifying the species and recording the results



1. A person trained in macroinvertebrate identification should be invited to assist. This may be a Waterwatch coordinator, professional person such as a CMA or local government staff member, or a teacher trained in water bug identification.

2. Use the *Water Bug Detective Guide* to help you identify the species.

3. Count the number of macroinvertebrates and the number of types.

4. Record the information on the recording sheet provided (refer to Section 10.3). This will give an indication of the health of your waterway based on the scores provided for each bug type and the number of macroinvertebrate types collected. The sensitivity score provides an indication of the tolerance of each macroinvertebrate to pollution and is sometimes called a SIGNAL score.

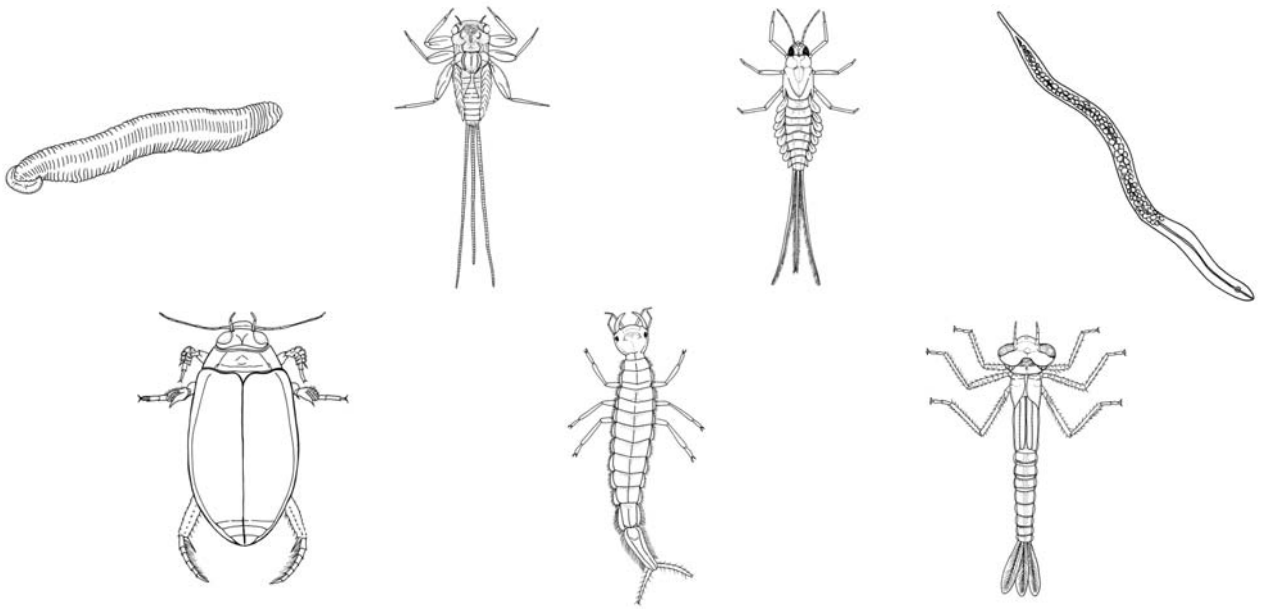
5. Gently return the macroinvertebrates to the water once you have finished, as close to the collection site as possible.

Classification of common macroinvertebrates

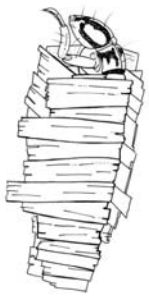
Phylum	Class	Order (suborder)	Family	Common name
Arthropoda	Insecta	Coleoptera	Elmidae	Riffle beetles
			Dytiscidae	Diving beetles
			Hydrophilidae	Scavenger water beetles
			Gyrinidae	Whirligig beetles
			Psephenidae	Water pennies
		Odonata (Zygoptera)		Damselflies
		(Anisoptera)		Dragonflies
		Hemiptera	Notonectidae	Backswimmers
			Corixidae	Water boatmen
			Nepidae	Water scorpions
			Hydrometridae	Water measurers
			Gerridae	Water striders
			Veliidae	Small water striders
		Diptera	Simuliidae	Black fly larvae
			Culicidae	Mosquitoes
			Chironomidae	Chironomids
			Ceratopogonidae	Biting midges
			Tipulidae	Craneflies
		Plecoptera		Stonefly larvae
		Ephemeroptera		Mayfly nymphs
		Trichoptera		Caddisfly larvae
		Megaloptera		Dobsonflies/ Alderflies
	Collembola			Springtails

Phylum	Class	Order (suborder)	Family	Common name
Arthropoda (continued)	Crustacea	Decapoda	Parastacidae	Freshwater crayfish
			Atyidae	Freshwater shrimp
			Palaemonidae	Freshwater prawn
			Sundathelphusidae	Freshwater crab
		Amphipoda		Sideswimmers
		Isopoda		Freshwater slater
	Arachnida	Acarina		Water mites
Annelida	Hirudinea			Leeches
	Oligochaeta			Segmented worms
Mollusca	Gastropoda			Snails
			Ancylidae	Limpets
	Bivalvia			Bivalve mussel
Platyhelminthes	Turbellaria			Flatworms
Nematoda				Roundworms
Cnidaria		Hydrozoa	Hydridae	Hydra

Source: Chessman 2003



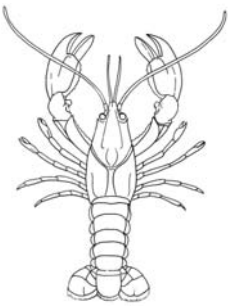
What do your results mean? – SIGNAL 2 and the stream pollution index (SPI)



The water quality of a river, creek or pond, sometimes called its 'ecological health' or 'river health', can be assessed on the basis of the presence or absence of animals living in the water. This indicates the ability of the water to sustain animal life.

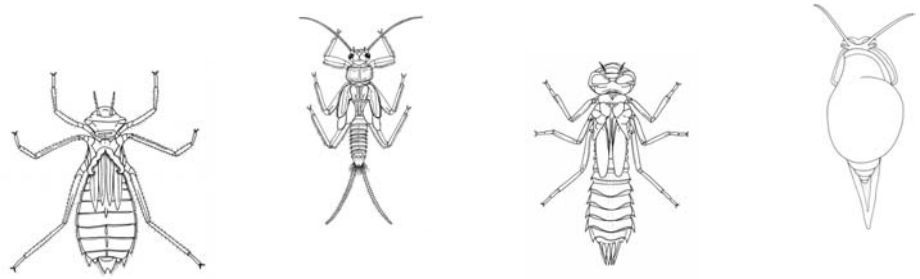
A system called SIGNAL2 has been developed to score the 'health' of the water, using aquatic macroinvertebrates (or water bugs). SIGNAL2 stands for Stream Invertebrate Grade Number Average Level. SIGNAL2 gives each type of macroinvertebrate a sensitivity rating from 1-10 to indicate their level of pollution tolerance.

'Pollution' can mean high levels of salinity, turbidity, nutrients (nitrogen or phosphorus) or a decrease in oxygen. This sensitivity rating, together with the number of types of bugs found, is used to create a Stream Pollution Index (SPI) for the river, creek or pond. Sites with high SPI scores are likely to have high levels of dissolved oxygen with low levels of pollution.

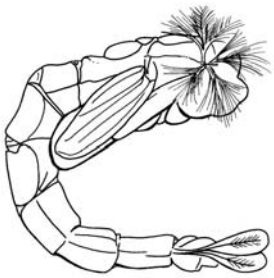


Still waters (wetlands, ponds and dams), inland and slow-flowing coastal rivers will always produce a lower SPI score because thier physical habitat and chemical levels are naturally different. Few macroinvertebrate types that are rated as very sensitive occur naturally in still waters or slow flowing lowland waters. By using the SPI score and considering the number of macroinvertebrate types found at your site, SIGNAL2 can provide an indication of the types of pollution and other physical and chemical factors that are affecting the macroinvertebrate community. In order for an SPI score to be calculated for your site, your sample MUST have at least 50 individual 'bugs'.

> ***i*** **Note: For further information about calculating the SPI, see the worked example in Section 10.4.**



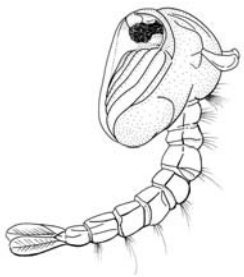
Tips for student macroinvertebrate sampling



Nets: These can be made from a kitchen strainer attached to a broom handle, stick or piece of dowel. This net is ideal for bugs as it will last many trips to the river and is inexpensive to make.

Scooping: Divide the class into groups of 4–5 students. Each group is to work in a specified location and remain there.

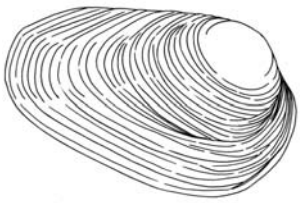
Sorting: After 10 minutes of scooping, students put the nets down and carry their trays away from the water's edge for sorting. This will ensure that students concentrate on the sorting of the bugs.



Identifying the species: Invite a trained person to assist with identification. Direct students to colour, shape, position of the legs and the number of tails. A two-way microscope or magnifying glass may assist with identification.

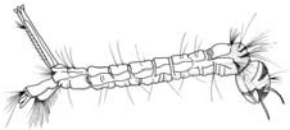
Calculating the stream pollution index (SPI): Add all group results together for a combined result. By entering the results of your bug survey on the Waterwatch website, the stream pollution index for your site and the number of macroinvertebrate types will be calculated. A description of your site will be provided **based on the macroinvertebrates you have collected**.

> ***i*** **Note: For more information check the website: www.waterwatch.nsw.gov.au**



Note: Reducing the risk of spreading chytrid fungus when sampling macroinvertebrates

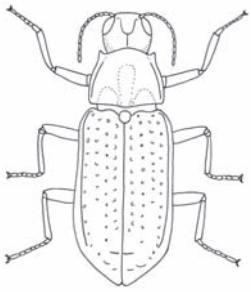
The chytrid fungus attacks keratin which is embedded within the sensitive skin of frogs and is often fatal. Chytrid can be transferred from one site to another by water and moist soil. It is possible to spread the fungus by contaminated nets and moist soil on car tyres and on the soles of shoes. To reduce the risk of spreading the fungus, spray nets, shoes and car tyres with a mixture of 50% bleach and 50% water to disinfect them prior to departure from a site. Never transfer water or bugs between sites.



10.2 Water bug survey: teacher field checklist

Date: ..... Class: .....

Item	Checked
TEACHER ORGANISATION	
Permission notes	☐
Class list	☐
Special needs student list	☐
Risk assessment sheet for completion	☐
Buses (if applicable)	☐
First aid kit	☐
Sunscreen	☐
Student medications	☐
Mobile phone	☐
STUDENTS CLOTHING	
Hats	☐
Closed toe shoes	☐
Drinking water	☐
FIELD EQUIPMENT	
Bucket	☐
Large trays	☐
Ice cube trays	☐
Spoons, pipettes, brushes	☐
Magnifying glass (optional)	☐
Macro nets	☐
RECORDING AND ID SHEETS	
Pencil case	☐
Marker pens	☐
Folder of result sheets + info	☐
Clipboards	☐
Camera	☐
Bug identification laminates	☐
Gambusia information sheet	☐



10.3 Water bug survey: SIGNAL 2 field recording sheet

Sampler group name:.....

Number in group:.....

Survey period: ☐ Spring ☐ Autumn ☐ Other .....

Date: ..... Time: ..... Time taken (hours): .....

Location of water body:

☐ western NSW river or stream <300 metres asl ☐ other rivers and streams ☐ wetland

Note: The rating of your stream pollution index (SPI) will be affected by the location of the sampling.

Habitats sampled:

Habitat *(tick the boxes where you sampled)*	**Still water**	**Moving water**
Silt and sand	☐	☐
Stones	☐	☐
Water plants	☐	☐
Leaves and twigs	☐	☐
Logs, branches, tree roots	☐	☐

Note: The more habitats sampled the greater the expected number of bug types.

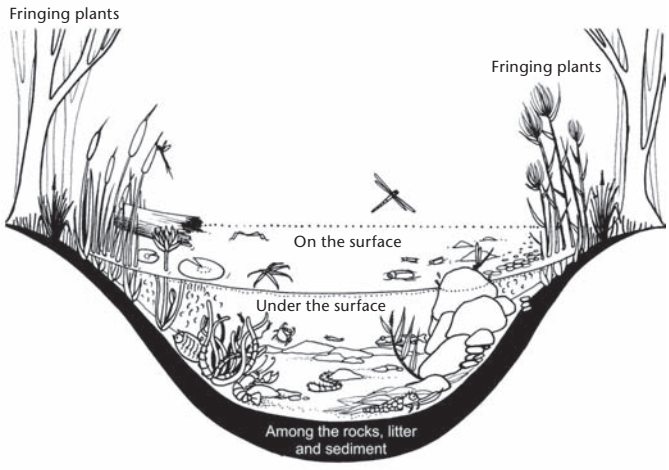
Sampling methods: *(tick the boxes)*

☐ sweep ☐ kick

Identification of bug species confirmed by a trained person: .....

.....

(e.g. Waterwatch coordinator, professional staff of council or agency, experienced teacher or community member)



Water bug survey: SIGNAL2 result sheet

Survey site name: .....

Step 1: Tick the bug type if present (see the Detective Guide in Section 10.5).
Step 2. Enter the number of each bug found in Column B.
Step 3: Refer to the weight table for the correct Weight factor for the number found.
Step 4: Enter the correct Weight factor for each bug in Column C.
Step 5: Multiply the Sensitivity rating (Column A) by the Weight factor (Column C) and enter the answer in Column D.
Step 6: Add up Column C (Weight factors).
Step 7: Add up Column D (Sensitivity rating x Weight factor).
Step 8: Add up the number of bug types.

WEIGHT TABLE	
No. of each bug found (Column B)	Weight Factor (Column C)
1-2	1
3-5	2
6-10	3
11-20	4
>20	5

Water bug recording table

MACROINVERTEBRATE TYPES			A	B	C	D
Sensitivity rating	**Taxa richness (bug types)**	**Tick if present**	**Sensitivity rating**	**Number of bugs**	**Weight factor**	**Column A X Column C**
Very sensitive	Stonefly nymph	☐	10			
	Mayfly	☐	9			
Sensitive bugs	Alderfly larva	☐	8			
	Caddisfly larva	☐	8			
	Riffle beetle & larva	☐	7			
	Water mite	☐	6			
Tolerant bugs	Beetle larva	☐	5			
	Dragonfly nymph	☐	4			
	Water strider	☐	4			
	Whirligig beetle & larva	☐	4			
	Freshwater yabby/crayfish	☐	4			
	Damselfly nymph	☐	3			
	Fly larva & pupa	☐	3			
	Midge larva & pupa	☐	3			
	Freshwater mussel	☐	3			
	Nematode	☐	3			
	Freshwater sandhopper	☐	3			
	Freshwater shrimp	☐	3			
	Water scorpion/needle bug	☐	3			
Very tolerant bugs	Diving beetle	☐	2			
	Flatworm	☐	2			
	Hydra	☐	2			
	Water treader	☐	2			
	Freshwater slater	☐	2			
	Water boatman	☐	2			
	Freshwater worm	☐	2			
	Backswimmer	☐	1			
	Bloodworm	☐	1			
	Leech	☐	1			
	Mosquito larva & pupa	☐	1			
	Freshwater snail	☐	1			
	TOTALS	☐				

Did you catch *Gambusia* at your site? ☐ Yes ☐ No ☐ Did not look

10.4 Calculating the health of your site

Step 1: Count the number of bug types (taxa richness). **No. of bug types:** ☐

Step 2: Calculate the SPI = $\frac{\textbf{Total Column D} \ \square}{\textbf{Total Column C} \ \square} = \square$

Step 3: Classify the number of bug types and stream pollution index (SPI) as high or low based on your site description and the levels in the rating table below:

Taxa richness (number of bug types) = ☐ **High** ☐ **Low**

SPI = ☐ **High** ☐ **Low**

Bug type and SPI rating table

Site description	SPI		Taxa richness (bug types)	
	Low	High	Low	High
Wetlands	0–3.1	>3.1	0–14	>14
Western NSW rivers or streams <300 metres asl	0–3.1	>3.1	0–11	>11
Other rivers and creeks	0–3.5	>3.5	0–15	>15

Step 4: Identify the site conditions based on your bug count.

SIGNAL 2 Scoring table

SIGNAL 2 scoring	Taxa richness	Site conditions based on the macroinvertebrate sample
High	High	Good water quality and a diversity of habitats. It may be a well-managed site, natural bushland or a national park.
Low	High	Water quality may be slightly affected by human activity or natural factors. There may be higher levels of salinity and/or nutrient levels at the site.
High	Low	Water quality is affected by a pollution source upstream or there are few habitats due to harsh physical conditions.
Low	Low	Water quality is affected by human use such as urban, industrial or agricultural pollution or by the downstream effects of dams.
Unable to calculate	Unable to calculate	Unable to calculate an SPI score as there are fewer than 50 macroinvertebrates in the sample. This may indicate poor sampling technique, or that your site is under stress. There may be poor habitat diversity and/or water quality. Make sure you sample in all habitats and keep an eye on the site.

Step 5: If the table does not represent your site, what other factors may influence water quality at your site? .....

.....

Note: These may change over time and may include rainfall, river flow, land use, drains, condition of banks and riparian vegetation.

Upload your results to the Waterwatch website at www.waterwatch.nsw.gov.au. The online database will calculate the stream pollution index (SPI) and provide a description of your site based on the bugs collected.

Worked example

Enter your results in the recording table and complete Column C by referring to the weight factor table (see blank recording sheet). Complete Column D by multiplying the sensitivity rating by the weight factor. For example, 3–5 bugs has a weight factor of 2. Multiply the sensitivity rating (Column A) by the weight factor (D). For stonefly in the example below, this is 10 x 2 = 20.

Extract from a water bug recording table

Bug type			**A**	**B**	**C**	**D**
Sensitivity rating	**Taxa richness (bug types)**	**Tick if present**	**Sensitivity rating**	**Number of bugs**	**Weight factor**	**Column A X Column C**
Very sensitive	Stonefly nymph	✓	10	3	2	20
Sensitive	Water mite	✓	6	20	4	24
Tolerant	Whirligig beetle & larva	✓	4	11	4	16
	Freshwater yabby/crayfish	✓	4	2	1	4
	Damselfly nymph	✓	3	5	2	6
	Freshwater shrimp	✓	3	30	5	15
Very tolerant	Water boatman	✓	2	16	4	8
	Freshwater worm	✓	2	15	4	8
	Mosquito larva & pupa	✓	1	12	4	4
	Freshwater snail	✓	1	33	5	5
	TOTALS	10		147	35	110

Calculate the stream pollution index (SPI).*

Step 1: Calculate the SPI = $\frac{\text{total of column D}}{\text{total of column C}} = \frac{110}{35} = 3.2$

Step 2: Count the number of bug types: Bug types = 10

Step 4: Classify as high or low the number of bug types using the table provided

Step 5: Based on your SPI and the number of invertebrate types, the condition of your site may be classified as:

SPI rating	**Number of bug types**	**Site conditions based on the macroinvertebrate sample**
Low	Low	Your results may indicate that water quality is affected by human use such as urban, industrial or agricultural pollution or by the downstream effects of dams.

If the table does not represent your site, what other factors may influence water quality at your site?

Note: These may change over time and may include rainfall, river flow, land use, drains, condition of banks and riparian vegetation.

SECTION 11

Human impacts on waterways

The human use of catchments has modified the natural environment. This has changed both the quality of our water, and the way that water flows (quantity), with large effects on our native vegetation, animal life and soils.

Some of the most significant changes to the natural environment brought about by human activity are increased soil and river salinity, land degradation, water pollution, loss of biodiversity and climate change. Management of these issues while maintaining the productivity and sustainability of the natural environment is a key challenge for the future.

This section comprises work sheets to help students explore these issues at their river, creek or estuary.

Included in this section: *Page*

11.1 Pipe and drain inventory work sheet

Site name: .....

Date: .....

Record observations on each pipe and drain found on the banks or in the stream.

Location of pipe/drain:

Location	Number
In-stream	
In-bank	
Near stream	

Types of pipes and drains at the sites:

Type of pipe/drain	Number of this type
Industrial	
Sewage treatment plant	
Agricultural	
Culvert	
Parking area drain	
Other *(please specify)*	

Condition of streambank below the pipe or drain:

☐ no problem evident ☐ rubbish (cans, paper)

☐ eroded ☐ lots of algae ☐ other .....

Discharge flow:

If there is a flow from the drain, circle the following:

Rate of flow	None	Trickle	Heavy	Intermittent	Steady
Appearance	Clear	Foamy	Turbid	Oily	Coloured
Odour	None	Sewage	Fishy	Chemical	Chlorine
Other *(please specify)*					

Additional comments: .....

.....

.....

11.2 Litter survey work sheet

Site name: .....

Date: .....

Collect litter from your site and sort into the following categories.

Tick whether the litter in each category is abundant, common or rare.

Type of litter	**Examples**	**Abundance**		
		Abundant	**Common**	**Rare**
Land litter	Non-floating materials, uncapped glass bottles, newspaper Others:	☐	☐	☐
Natural litter	Leaves, sticks, twigs Others:	☐	☐	☐
Hard plastic	Buckets, syringes, fragments Others:	☐	☐	☐
Foam/soft plastic	Plastic bags, food packaging Others:	☐	☐	☐
Glass	Bottles, jars, light globes Others:	☐	☐	☐
Aluminium	Drink cans, aerosol cans Others:	☐	☐	☐
Rubber	Gloves, thongs, hoses Others:	☐	☐	☐
Wood	Timber, planks Others:	☐	☐	☐
Cellulose	Paper, cardboard, waxed cartons Others:	☐	☐	☐
Cloth/hessian	Sacking, garments, hats Others:	☐	☐	☐

From the survey, list the **main** items in each category: abundant, common and rare.

Abundant : >70% of litter	Common: 10%–70%	Rare: <10%

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WATER BUG DETECTIVE GUIDE

Macroinvertebrate sampling and waterway health

Sampling will reveal information about the abundance and diversity of macroinvertebrates and their tolerance to pollution: This will provide an indication of the health of the waterway.

ABUNDANCE = the total number of macroinvertebrates present

DIVERSITY = the number of different types of bugs present. Healthy streams usually have a greater diversity of bug types

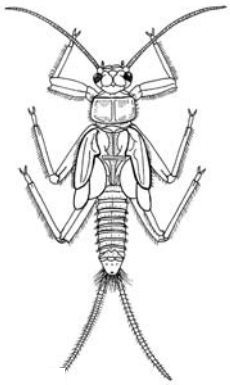
POLLUTION TOLERANCE = the ability of macroinvertebrates to withstand pollution. This is reflected by its SIGNAL 2 score based on their sensitivity to pollution.

STREAM POLLUTION INDEX = calculation based on the abundance and diversity of bugs and their SIGNAL 2 score.

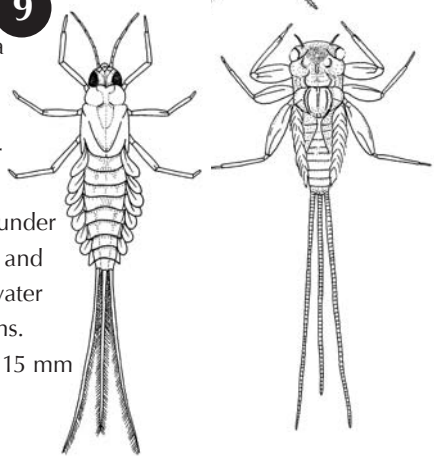
HEALTHY WATERWAYS = a high SIGNAL score and a large number of bug types

Very Sensitive Bugs - 10,9

Stonefly nymph **10**
Order: Plecoptera
Description: Two thin tails and gills extending from their abdomen.
Habitat: Found among stones or plants, in fast-moving waters.
Maximum size: 7-12 mm

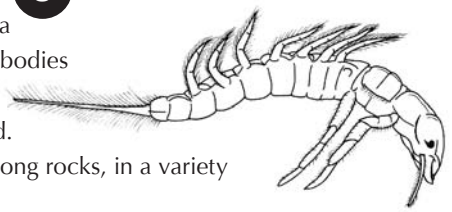


Mayfly nymph **9**
Order: Ephemeroptera
Description: Three long thin tails and gills along the sides of their bodies.
Habitat: Found on or under rocks or among plants and leaf litter in standing water and fast flowing streams.
Maximum size: Up to 15 mm

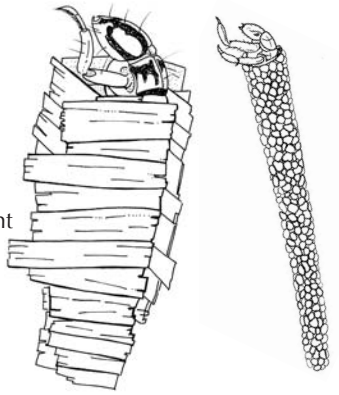


Sensitive Bugs - 8,7,6

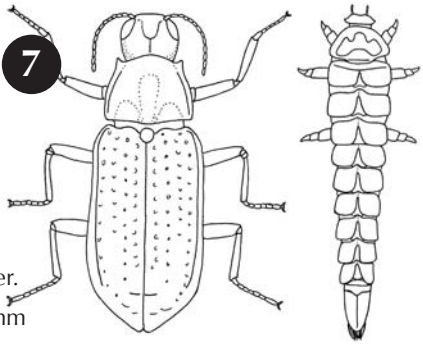
Alderfly larva **8**
Order: Megaloptera
Description: Their bodies are fleshy with a a hard-shelled head.
Habitat: Found among rocks, in a variety of flow conditions.
Maximum size: Up to 20 mm



Caddisfly larva **8**
Order: Trichoptera
Description: They are often enclosed within a case of twigs and plant material or silk.
Habitat: Found among sediment and rocks in streams, ponds and lakes.
Maximum size: Up to 20 mm

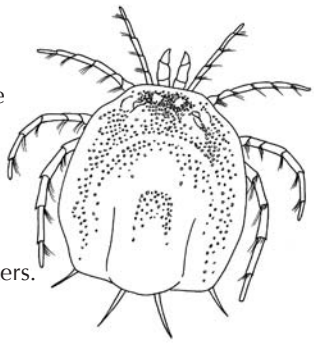


Riffle beetle and larva **7**
Order: Coleoptera
Description: Beetle-like, tiny and usually black. Larvae have circular stripes or rings.
Habitat: Fast moving water.
Maximum size: Up to 4 mm



Adult Larva

Water mite **6**
Order: Acarina
Description: Mites usually have simple rounded bodies with eight legs.
Habitat: Found among plants or stones on the stream bed in standing or slow-moving waters.
Maximum size: Up to 5 mm



Illustrations: Christine Rockley

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Tolerant Bugs - 5,4,3

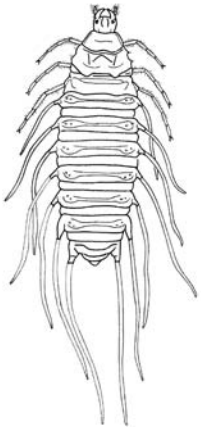
Beetle larvae 5

Order: Coleoptera

Description: Larvae are usually elongated with well-developed legs and a large head.

Habitat: A variety of habitats including still waters or quiet areas of flowing water.

Maximum size: Up to 35 mm



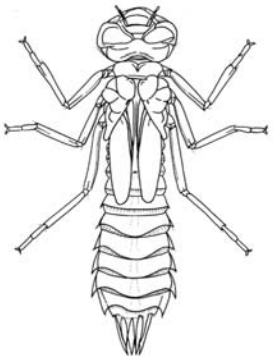
Dragonfly nymph 4

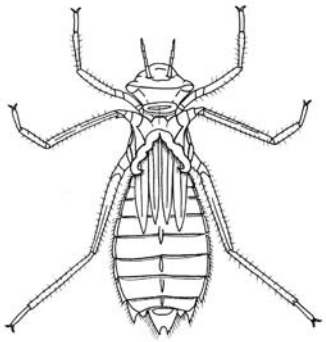
Order: Odonata

Description: Stout bodies, no external gills and extendable mouth parts.

Habitat: Found within the substrate of rivers and streams.

Maximum size: 12-50 mm





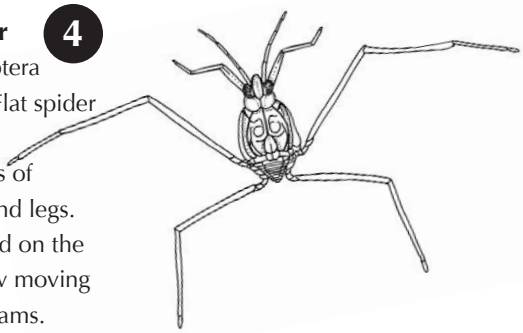
Water strider 4

Order: Hemiptera

Description: Flat spider appearance with long pairs of middle and hind legs.

Habitat: Found on the surface of slow moving rivers and streams.

Maximum size: 8-12 mm



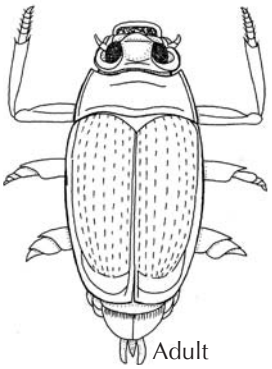
Whirligig beetle and larva 4

Order: Coleoptera

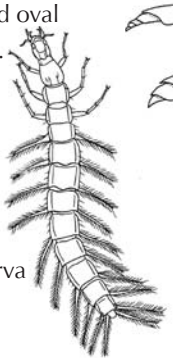
Description: A streamlined oval beetle that swim in circles.

Habitat: Found on the surface around the edges of ponds an streams.

Maximum size: 5-25 mm



Adult



Larva

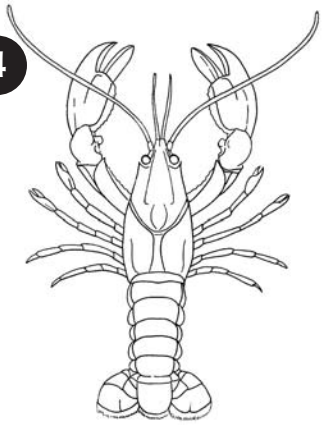
Freshwater yabby/crayfish 4

Order: Decapoda

Description: Fan tailed with well developed claws and prominent front end.

Habitat: Slow flowing and still waters and burrow into sediment.

Maximum size: Up to 400 mm



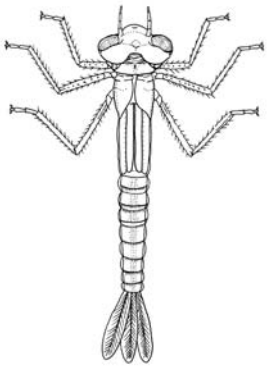
Damselfly nymph 3

Order: Odonata

Description: Nymphs have three gill structures extending from the tail.

Habitat: Found on plants, among rocks and leaf litter or burrowing into the sediments.

Maximum size: 16-33 mm



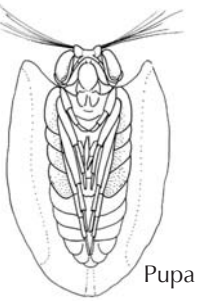
Fly larva and pupa 3

Order: Diptera

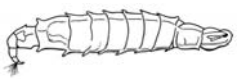
Description: Larva body is grub like and segmented with piercing and sucking mouthparts. They do not have true legs.

Habitat: Found in shallow regions of ponds and stream amongst mud and detritus.

Maximum size: Up to 30 mm



Pupa



Larva

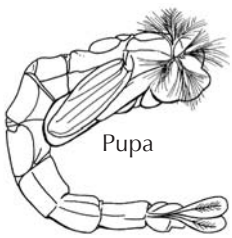
Midge larva and pupa 3

Order: Diptera

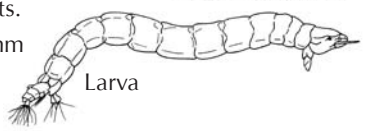
Description: Often small and C shaped.

Habitat: Attached to debris by their tiny legs and can be found anywhere that water collects.

Maximum size: Up to 50 mm



Pupa



Larva

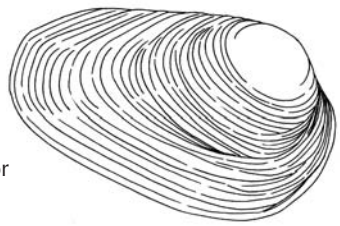
Freshwater mussel 3

Class: Bivalvia

Description: Freshwater mussels have paired hard shells (valves) with a fleshy body between them.

Habitat: Found in or on sandy or muddy stream beds.

Maximum size: Up to 150 mm



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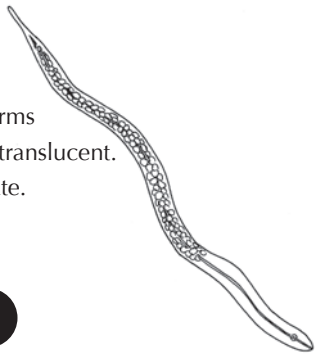
Tolerant Bugs - 5,4,3

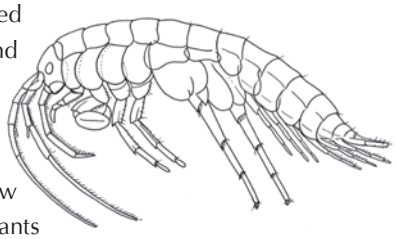
Nematode 3

Order: Nematoda
Description: Thin elongated worms without segments and can look translucent.
Habitat: Burrow into the substrate.
Maximum size: Up to 12 mm



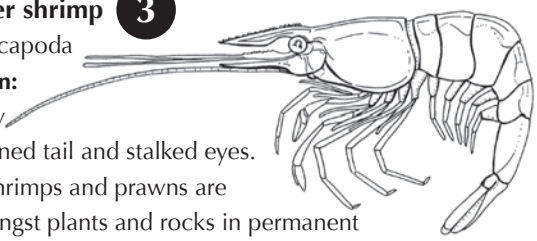
Freshwater sandhopper 3

Order: Amphipoda
Description: Slightly curled and flattened sideways and have hard segments each with a pair of legs for swimming or walking.
Habitat: The edges of slow moving water amongst plants and stones.
Maximum size: 6-20 mm



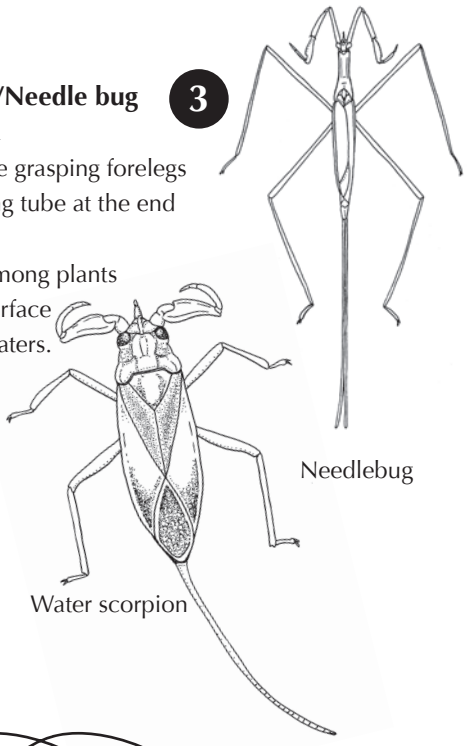
Freshwater shrimp 3

Order: Decapoda
Description: Covered by a shell, fanned tail and stalked eyes.
Habitat: Shrimps and prawns are found amongst plants and rocks in permanent slow-moving waters.
Maximum size: Up to 35 mm



Water scorpion/Needle bug 3

Order: Hemiptera
Description: Large grasping forelegs and short breathing tube at the end of their abdomen.
Habitat: Found among plants or on the water surface of slow-moving waters.
Maximum size: Up to 50 mm



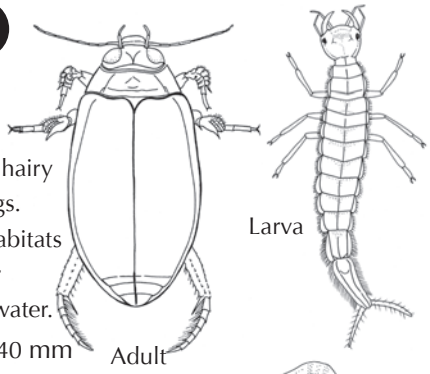
Needlebug

Water scorpion

Very Tolerant Bugs - 2,1

Diving beetle 2

Order: Coleoptera
Description: Sleek, shiny beetles with hard-shelled body and hairy paddle-shaped hind legs.
Habitat: A variety of habitats including still waters or quiet areas of flowing water.
Maximum size: Up to 40 mm



Larva

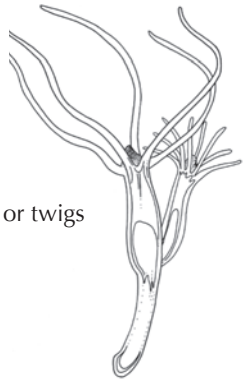
Adult

Flatworm 2

Class: Turbellaria
Description: Flat, thin, slow-moving worms with two simple eye spots.
Habitat: Found gliding over rocks and plants in a variety of flow conditions.
Maximum size: Up to 20 mm

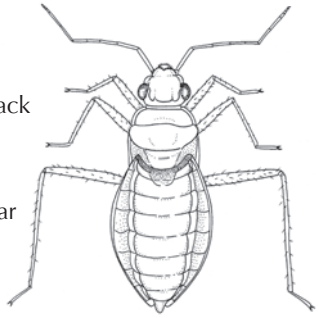
Hydra 2

Class: Hydrozoa
Description: Hydras have a simple sack-like body with a mouth encircled by tentacles.
Habitat: Found attached to rocks, plants or twigs in fast flowing water.
Maximum size: Up to 30 mm



Water treader 2

Order: Hemiptera
Description: Long middle and back legs and thick body.
Habitat: Found on the water's surface of slow flowing pools near banks and plants.
Maximum size: Up to 5 mm



Freshwater worm 2

Class: Oligochaeta
Description: Segmented worms with rounded ends with no suckers or legs and usually coloured red or flesh coloured.
Habitat: Found in soft sediment rich in organic matter.
Maximum size: Up to 30 mm

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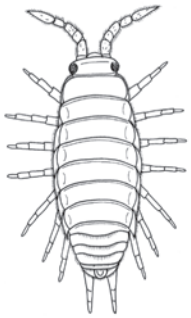
Freshwater slater 2

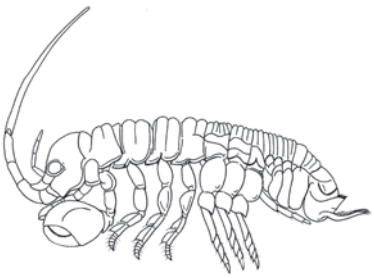
Order: Isopoda

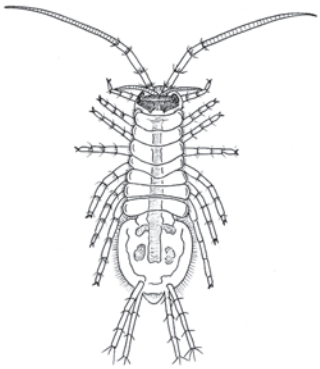
Description: Flattened from top to bottom with no body caraspace or shield.

Habitat: Found in still to slow-moving waters.

Maximum size: Up to 20 mm







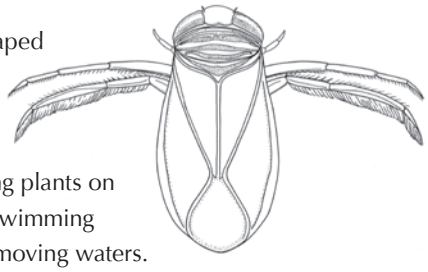
Waterboatman 2

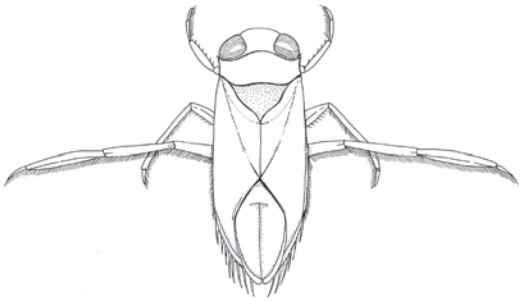
Order: Hemiptera

Description: Boat-shaped with piercing mouth parts and boat shape appearance.

Habitat: Found among plants on the water surface or swimming freely in still to slow-moving waters.

Maximum size: Up to 10 mm





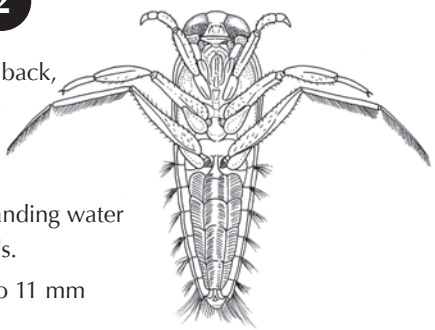
Backswimmer 2

Order: Hemiptera

Description: Curved back, large eyes, long hairy hind legs and swim on their backs.

Habitat: Found in standing water or slow flowing ponds.

Maximum size: Up to 11 mm



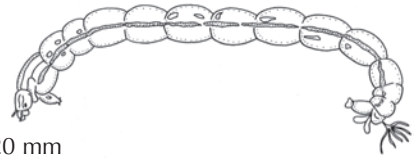
Bloodworm 1

Order: Diptera

Description: Worm-like and C shaped. Only the red ones are called Bloodworms.

Habitat: Found in soft sediment rich in organic matter.

Maximum size: Up to 20 mm



Leech 1

Class: Hirudinea

Description: Leeches are soft-bodied animals made up of 32 segments with a sucker on one or both ends.

Habitat: Found in standing or slow moving water.

Maximum size: 7-80 mm

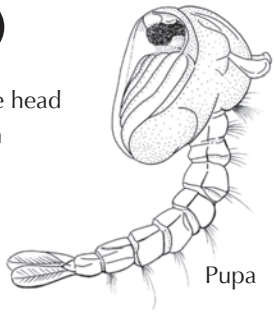
Mosquito larva and pupa 1

Order: Diptera

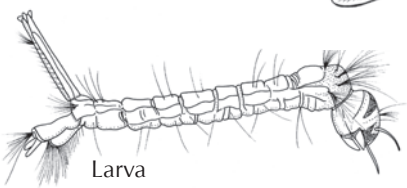
Description: Thorax wider than the head and breathes through a long siphon at the end of the abdomen

Habitat: Still water

Maximum size: Up to 25 mm



Pupa



Larva

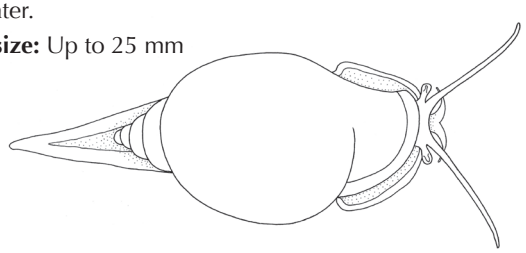
Freshwater snails 1

Class: Gastropoda

Description: Snails are soft-bodied animals enclosed in a hard, protective, coiled shell.

Habitat: Found on plants and rocks in slow flowing or standing water.

Maximum size: Up to 25 mm



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