Image by: John Lugg, DECCW

Waterwatch Estuary Field Manual

A manual for on-site use in the monitoring of water quality and estuary health

Waterwatch NEW SOUTH WALES
Communities Caring for Catchments

NSW GOVERNMENT Environment, Climate Change & Water

Australian Government

Acknowledgements

This *Waterwatch Estuary Field Manual* and the accompanying *Waterwatch Estuary Guide* have been developed by Waterwatch coordinators to provide additional monitoring techniques for Waterwatch groups in coastal areas of New South Wales. The methods and procedures have been adapted and enhanced to ensure the success of monitoring within estuaries.

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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

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How to use this manual

This *Waterwatch Estuary Field Manual* and the *Waterwatch Estuary Guide* have been designed to accompany the Waterwatch field manuals and guides for monitoring freshwater waterways. The two estuary documents contain specific information and activities required to monitor in estuaries. They have been designed to provide information and sampling techniques for all ages, as a guide to designing and implementing a Waterwatch estuary monitoring program within schools and the community.

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.

The manual is divided into numbered sections:

Section 1: Assessing your site

Section 2: Estuary monitoring in the field

Section 3: Interpreting your results

Section 4: Investigating habitats

Section 5: Habitat zones within estuaries

Section 6: Saltmarsh monitoring

Section 7 Seagrass monitoring

Section 8: Investigating salt water invertebrates

This field manual is to be used in conjunction with the *Waterwatch Estuary Guide*, as well as either the *Waterwatch Field Manual* or *Junior Waterwatch Field Manual*, and contains cross-references to those documents. For further assistance see the Waterwatch website or contact your nearest Waterwatch coordinator.

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.

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 and checklists to assist participants to do a detailed assessment of the estuary site they have chosen for investigation.

Section 1.3 contains a basic estuary site assessment. For a more detailed site assessment please refer to Section 1.8 of the Waterwatch Field Manual for freshwater monitoring.

Included in this section: *Page*

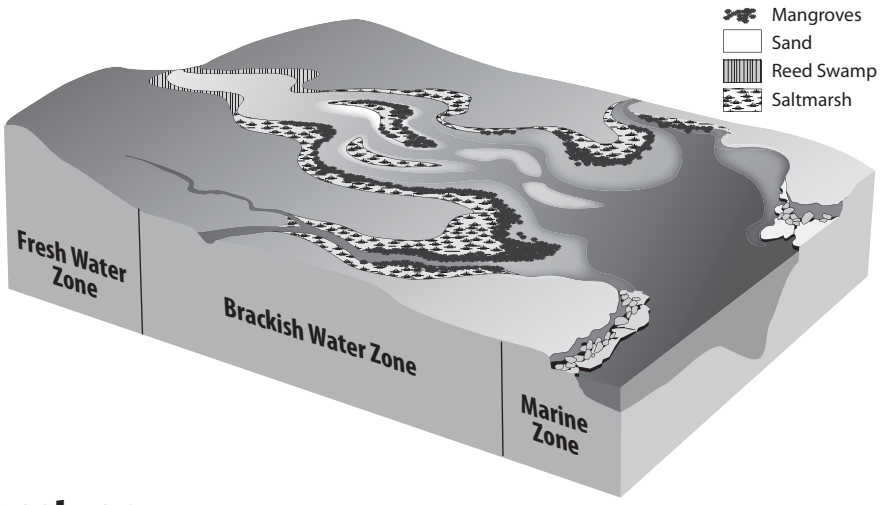
1.1 Identifying and mapping your estuary

Identify your estuary type

The following block diagrams show common estuary types in New South Wales. Use these diagrams to assist you to identify the type of estuary you are monitoring.

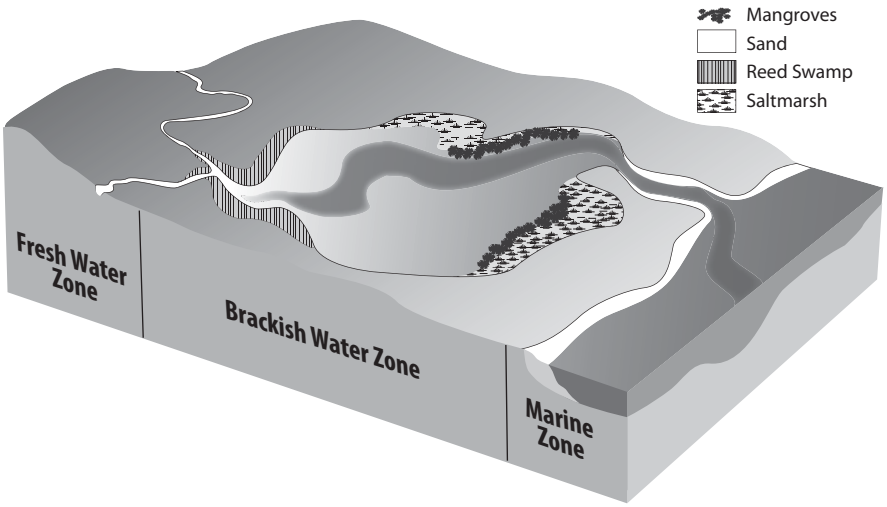
1. Coastal river estuary

Estuary mouth with salt flats and tidal sand banks
Example: Wagonga Inlet, Narooma



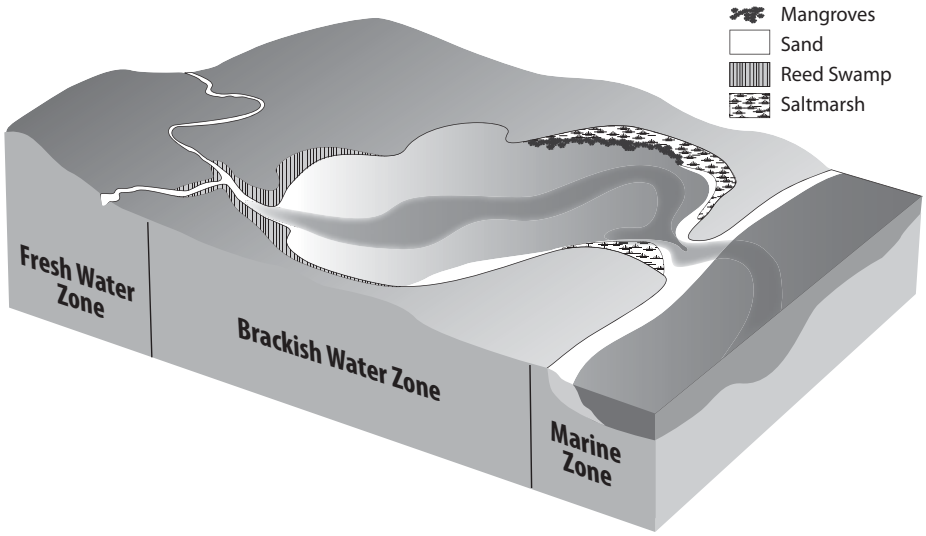
2. Barrier estuary

Coastal river with intertidal flats
Example: Pambula Lake, Tuggerah Lakes, Narrabeen Lagoon and Lake Macquarie



3. Coastal lagoon

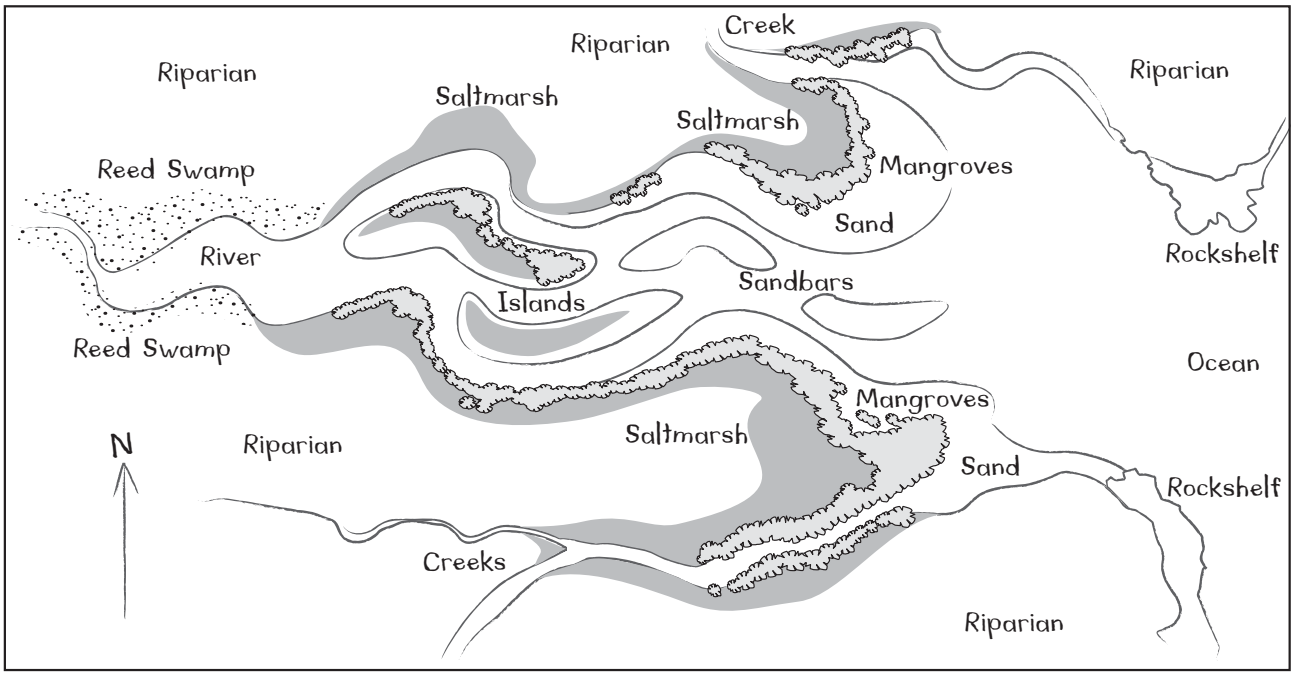
Example: Dee Why and Manly lagoons, Wollumboola Lake and Wallaga Lake



Map your estuary

A bird's eye view map shows a site as if seen from above. This kind of map shows areas of differing habitat, as well as structural and natural features. A series of bird's eye view maps can be used to show changes over time. An example of a bird's eye view sketch of a river dominated estuary is given below.

Arial views of the NSW coast can be found on Google Earth or Google Maps.



1.2 Draw a bird's eye view map of your estuary work sheet

Name: _____

Date:_____ /_____ /_____

Type of estuary:_____

Estuary name: _____

Draw and label a bird's eye view map of your estuary or find an aerial image and paste it here.

1.3 Estuary site checklist

(To be done the first time you visit your site.)

Complete the site summary checklist to describe estuarine features:

Group:	**Date:**	**Time:**

Tide: ☐ high ☐ low ☐ flood (coming in) ☐ ebb (going out) ☐ not tidal

Location:

The estuary monitoring site could be described as:
☐ coastal river/creek ☐ bay ☐ harbour ☐ lagoon/lake ☐ other_____

Identify the main landform features of the site:
☐ features formed by waves (sand bars) ☐ features formed by rivers (floodplain)
☐ features formed by both waves and the rivers

The site is open to the sea:
☐ permanently under natural conditions ☐ permanently due to man made structures
☐ sometimes open ☐ cut off most of the time

Salt water mixes with fresh water at the site:
☐ through a narrow channel ☐ through a wide mouth
☐ there is little or no mixing of fresh and salt water

The site has:
☐ large daily changes in water level due to tides
☐ small daily changes in water level due to tides
☐ no daily change in water level due to tides

At the site:
☐ salt and freshwater mix evenly ☐ salt and freshwater do not mix evenly
☐ there is very little mixing of fresh and salt water

Tick each habitat type at the site:
☐ saltmarsh ☐ mangroves ☐ seagrass
☐ freshwater stream ☐ other _____

Are native plants growing at the site? Tick each type at the site:
☐ native trees on the banks ☐ freshwater aquatic plants ☐ algae
☐ saltwater aquatic plants ☐ other_____

Are any of the following at or near the site?
☐ industrial area ☐ shops ☐ houses
☐ farming land ☐ native bushland/conservation area ☐ parks
☐ tourist facilities ☐ jetties/wharves ☐ fishing

Are there any stormwater outlets or drains at or near your site?
☐ yes ☐ no If yes, how many? _____

1.4 Site observation checklist

(To be done each time you visit your site.)

Name of monitoring group:	
Name of site:	
Date:	Time: AM/PM
Tide: ☐ high ☐ low ☐ flood (coming in) ☐ ebb (going out) ☐ not tidal	
Location in the estuary:	
Number sampling:	Time taken:

Answer the following questions by ticking the box which best describes your site today:

What is the weather like today?

☐ sunny ☐ cloudy ☐ light rain (showers) ☐ heavy rain

Was there any rain last week?

☐ none ☐ light rain (showers) ☐ heavy rain ☐ floods

The depth of the water at the site is:

☐ deep (more than 2 metres)

☐ medium (more than 1/2 metre but less than 2 metres)

☐ shallow (less than 1/2 metre)

Tick the boxes that best describe these features of your site.

1. Bottom		
Healthy	**Fair**	**Poor**
Clear – can see the bottom, signs of active animals	Milky or thick muddy layer of sediment	Cannot see the bottom (turbid/film on surface)
2. Top		
Healthy	**Fair**	**Poor**
Clear/no human litter	Cloudy/bubbles; some human litter/oil	Green algae covers top, lots of human litter or oily film
3. Foreshore cover		
Healthy	**Fair**	**Poor**
Stable bank, rock wall	Little erosion	Unstable bank, erosion and slumping
4. Smell		
Healthy	**Fair**	**Poor**
Odour is from natural sources (e.g. salt)	Minor odour (mostly natural or normal)	Odour caused by significant human impact such as sewage or grey water/ chlorine or chemicals
5. Appearance		
Healthy	**Fair**	**Poor**
Clear or with little colour OR stained brown by natural factors	Discoloured such as cloudy/ green/ brown	Green with algal growth

6. Pollution	
Healthy	**Poor**
No pollution	Some pollution (name it)_____

Rate the health of your site by adding up the number of healthy, fair and poor site features you have observed: _____

Observations: (water colour, land use, changes since last sampling)

1.5 Assessing the condition of the foreshore

Assess the condition of the foreshore: place a tick from 1–5 to give the foreshore a rating out of 50.

GROUP		DATE		TIME			NAME
LOCATION							
ESTIMATE THE LENGTH OF FORESHORE ASSESSED: METRES				FROM			TO
	WORSE	Increasing score and decreasing effect on lake foreshore and water quality					BETTER
	Greater impact	**1**	**2**	**3**	**4**	**5**	**Less impact**
1	Bare shoreline – subject to erosion						Natural sloping shoreline – allows dead seagrass to wash out, improving water quality
2	Hardened seawall shoreline - no 'natural filter', degrades water quality and causes erosion						'Soft' vegetated shoreline absorbs wave energy with less erosion – natural filter for runoff
3	Large buildings close to foreshore standing out from natural environment						Buildings set back from foreshore and in character with the natural environment
4	Mainly introduced vegetation – more gardening and more runoff occurs						Mainly native vegetation – less water and chemical use
5	No shade trees along foreshore						Naturally growing trimmed shade trees
6	Ornamental shrubs and manicured lawns – require chemicals and extra work fertiliser and chemical runoff from lawn damages water quality						Low maintenance native plants for stabilisation and filtration – provides shoreline buffer, mulched native gardens and shrubs
7	Paved driveways – pollution in runoff flows to waterway						Stepping stones for footpath – less chance of runoff and erosion, improves filtration
8	Direct stormwater pipe outlets to waterway – erodes sediment and stops seagrass growth						'Soft' stormwater outlets – artificial wetlands, energy barriers, pollution traps, provide a buffer to reduce erosion
9	Solid deck on jetty – affects sea life habitat under jetty due to poor light penetration						No jetty or small shared jetty with open mesh deck – low impact on seagrass habitat
10	Boats – poor fuel management and fuel spills affect aquatic life, inefficient 2 stroke or poorly maintained motors						Boats – good fuel management with well maintained modern motor – electric or 4 stroke, operated with low wake, no fuel spills
	TOTAL SCORES (add columns)	x1=	x2=	x3=	x4=	x 5=	

We have given the foreshore at _____ a TOTAL SCORE of _____ out of 50. Make a list of things that can be improved.

1.6 Assessing foreshore activities

Look at the activities going on around your estuary foreshore and decide which are environmentally friendly and which are harmful. Make some comments about them.

GROUP	DATE	TIME	NAME
LOCATION			
ESTIMATE THE LENGTH OF FORESHORE ASSESSED: METRES	FROM	TO	

	Actions	Environment friendly action	Harmful or illegal action	Comments
1	Damaging or removing live seagrass or collecting more than 20 kg per day of dead seagrass			
2	Dumping litter or rubbish			
3	Composting your grass clippings and garden waste and keeping them away from foreshore reserves. NOT littering			
4	Leaving dead seagrass along the foreshore beach to reduce erosion and to provide habitat			
5	Helping to protect and rehabilitate foreshore bushland by getting involved in Landcare			
6	Being a responsible dog owner. Picking up your dog poo and disposing of it carefully			
7	Constructing a seawall or filling or dredging the shoreline without a permit			
8	Constructing new jetties, moorings, slips or ramps without approval			
9	Launching boats only from properly constructed and authorised boat ramps. Storing boats within your property			
10	Removing or damaging mangroves or removing saltmarsh without approval			

Assessing foreshore activities (continued)

Actions		Environment friendly action	Harmful or illegal action	Comments
11	Dumping grass clippings in waterways or where they can be washed or blown into waterways			
12	Leaving at least a metre buffer along the edge of the foreshore when mowing			
13	Restricting access or causing damage to foreshore reserves by fences, walls, hedges, landscaping, boats or equipment			
14	Planting local native species in the waterfront buffer zone. Avoiding plants that can 'escape' into bushland			
15	Damaging native vegetation on waterfront reserves, including by mowing			
16	Erecting structures within 6 metres of high watermark			
17	Storing all privately-owned equipment and materials within your property boundary			
18	Restoring a natural slope to your water frontage and ensuring access around the foreshore			
19	Leaving dead and fallen trees to provide wildlife habitat			
20	Allowing your dog to foul near waterways			

SECTION 2

Estuary monitoring in the field

Estuaries are highly valued aquatic ecosystems; however, rising populations and human use of coastal areas is placing increasing pressure upon them, causing many changes. Some of the visible changes in estuaries include habitat destruction while other changes are long-term, gradual and/or not noticeable to the human eye.

By monitoring an estuary over the long term, the status of indicators can be determined (i.e. whether they are stable, improving or declining) and early changes can be detected. Without monitoring, it can be very difficult to prove or gauge the amount of change.

This section includes information about safety considerations when working at the estuary and essential skills such as using quadrats and transects. It also outlines the water quality tests appropriate for the estuarine environment.

Included in this section: *Page*

This section should be read in conjunction with the corresponding section in the *Waterwatch Field Manual* or *Junior Waterwatch Field Manual*, which contains important additional safety information about the water quality tests and ensuring that data collected is of the highest quality.

2.1 Safety at the estuary

In addition to the considerations provided below, an OHS Risk Management Plan should be completed each time an on-site monitoring activity occurs. A template for this plan is provided in both the senior and junior Waterwatch teachers' guides, and in the *Community/Land Manager Waterwatch Guide.*

To ensure the safety of all participants in Waterwatch on-site activities, include the following considerations in your planning:

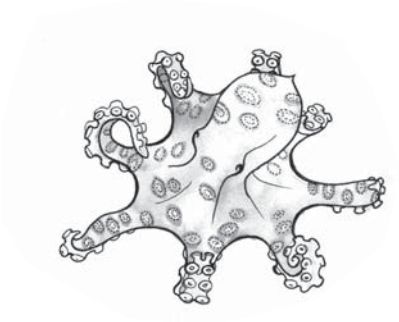
- Obtain a tide chart so that you know when sites will be accessible and for how long. These are available online at www.bom.gov.au and www.tidetimes.com.au
- Many activities can only be conducted at low tide. Always start work from the lowest point and work towards the high-tide mark when conducting transect surveys.
- Let someone else know when, for how long and where your group will be sampling.
- Always wear sturdy, enclosed shoes when wading in the water or walking along shoreline areas due to the risk of lacerations from broken glass and sharp objects.
- Keep an eye out for sharp items such as broken glass, syringes, oysters.
- When planning a visit to your local estuary to look for water bugs, let the local office of the Marine Parks Authority or NSW Fisheries know, and obtain all relevant permits.
- Never sample in or near very fast flowing water.
- Be sun smart: wear a hat and sunscreen.
- Wear appropriate clothing when sampling.
- Always work at the site with a buddy.
- Do not put yourself or others at risk of falling into the water.
- Beware of dangerous sea creatures that may be washed up on the foreshore such as the blue-ringed octopus, blue-bottles other jellies. Even when dead or dying, they can still sting if stepped on or handled.

Harmful or dangerous creatures

Australia is home to many fascinating marine creatures. Foreshores, rockpools and estuaries are prime habitat for many species, some of which are dangerous or harmful to humans. The best defence is to wear sturdy shoes, avoid close contact with all marine creatures and keep a sharp eye out when walking. A few of the more dangerous creatures are listed below, but there are others. When undertaking an estuary monitoring program, it is important to research the creatures in your area and be able to identify the harmful ones before commencing monitoring.

Blue ringed octopus

The adult blue ringed octopus is 10–20 cm wide and can be found in very small places including in shallow coral and rock pools, under rocks, in cracks and crevices on the reef, in tidal pools, inside shells, and in discarded bottles, cans and other detritus on the sea floor. It can also be found in sandy or muddy stretches of the sea bottom where seaweed and seagrass is plentiful. There are two species found in Australia: the greater blue ringed octopus, found in northern Australia, the Pacific Islands and New Guinea; and the lesser blue ringed octopus, found in southern Australia, from Western Australia to south-eastern Victoria and New South Wales.



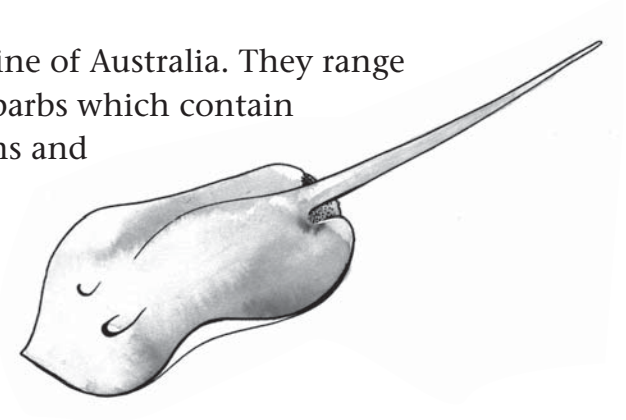
The blue ringed octopus is highly poisonous. Numbness, breathing problems and paralysis swiftly occur after being bitten. The pressure–immobilisation technique is recommended for use after a bite, with mouth-to-mouth resuscitation if needed. Victims must be transported immediately to the nearest hospital or medical centre for urgent treatment.

Razor shell/clam

The razor shell/clam, also called ‘razor fish’ is a widespread species found around most Australian shores, especially in muddy shallow estuaries. Common razor clams, *Pinna bicolor*, are large, triangular bivalves, up to 35 cm long.

The razor shell/clam buries itself vertically in the sediment, leaving a very thin and sharp blade of shell exposed for the unwary to stand on. Wear solid-soled shoes (wetsuit boots, gumboots or old sneakers) to prevent injury and seek medical attention if cut by one.

Stingray

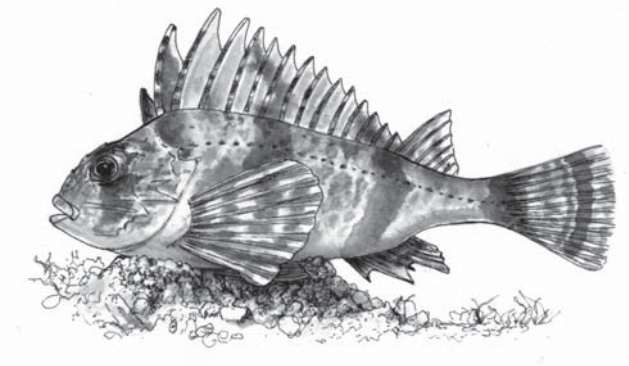


Stingrays are common along the entire coastline of Australia. They range in size from 5 cm to 7 m and most have two barbs which contain venom. They are usually quite wary of humans and will move away in preference to defending themselves. If you shuffle your feet whilst wading through the water, stingrays will generally move away quickly. If stung, it is important to control the bleeding but leave the barb in place and seek medical attention immediately.

Sea urchin

Sea urchins are spiny circular creatures. They are found in rock pools around the coastlines of Australia. They cause puncture wounds with their sharp, venous spines when touched, stepped on, or bumped. Seek medical assistance for treatment to remove the spines.

Fortescue and scorpion fish



Fortescues and scorpion fish are small, attractive and slow-swimming fish found in estuaries, often within seagrass beds. *Centropogon australis* grows to about 15 cm, while *Gymnapistes marmoratus* is smaller. They have venomous spines on their backs that cause severe pain if stepped on. Seek medical attention.

2.2 Equipment for testing and sampling

Water quality monitoring equipment

Equipment	Measurement units	Test shows
Turbidity tube	Nephelometric Turbidity Units (NTUs)	Turbidity
Secchi disk	Depth (metres)	
EC meter	EC units – mS/cm; µS/cm	Electrical conductivity (EC)
Thermometer	ºCelsius (ºC)	Temperature
pH papers	7–14 pH scale – 1 pH unit OR 4.5–10.0 – 0.5 pH units	pH
Colorimeter: Smart, Smart 2, DC1200	Milligrams per litre (mg/L	Available phosphate
Colorimeter: Smart, Smart 2 OR Dissolved oxygen titration	Milligrams per litre (mg/L)	Dissolved oxygen
1 litre sample bottle	CFU per 100 mL	Enterococci

Note: Additional parameters can be tested using a Smart and Smart 2 colorimeter.

Invertebrate and seagrass sampling equipment

Method	Equipment
Netting invertebrates	Dipnets, sorting trays, ice block containers, pipettes (with narrow ends cut off), plastic spoons, small paint brushes, bug viewer, identification sheets
Seagrass sampling	Hoop (50 cm diameter) or PVC pipe quadrat (50 cm x 50 cm), tongs, thick gloves, pen, clipboard, secchi disk, seagrass recording sheet, 100 m tape measure, Seagrass Identification Guide
Seagrass wrack sampling	Tongs, disposable gloves, sorting tray, ice cube containers, 2–way microscope and identification sheets
Core sampling	Core sampler or yabby pump, spade, sorting tray, sieve, pipettes, plastic spoons, mesh sieve, 2–way bug viewer, macroinvertebrate identification guide, recording sheets
Transects/quadrats	Hoop (50 cm diameter) or PVC pipe quadrat (50 cm x 50 cm), 30–50 m fibreglass tape measure, tent or wooden pegs, string line, viewing bucket or bathyscope, macroinvertebrate identification guide, recording sheets

2.3 Monitoring water quality in estuaries

Refer to the *Waterwatch Field Manual* or *Junior Waterwatch Field Manual* for detailed information about standardised monitoring and sampling techniques.

For monitoring in estuaries, water quality parameters that can be measured using standard Waterwatch methods and equipment are as follows:

All Waterwatch groups (junior, senior and community)

- Temperature
- pH
- Electrical conductivity
- Turbidity

Waterwatch secondary schools and community additional parameters:

- Dissolved oxygen
- Available phosphate
- Enterococci/E. coli
- Nitrate – This test requires the use of chemicals containing cadmium, which is carcinogenic. Volunteers need to carefully consider whether this test is essential or whether it would be better tested by a laboratory. Disposal of nitrate test wastes in hazardous waste containers is essential.

Estuary-specific tests

Monitoring water clarity

Estuaries often have high levels of water clarity compared to freshwater rivers and streams and a secchi disk may provide a better indication of water clarity for estuarine waters than a turbidity tube would. A secchi disk is a circular disk marked with black and white quarters. Water clarity is measured by depth.

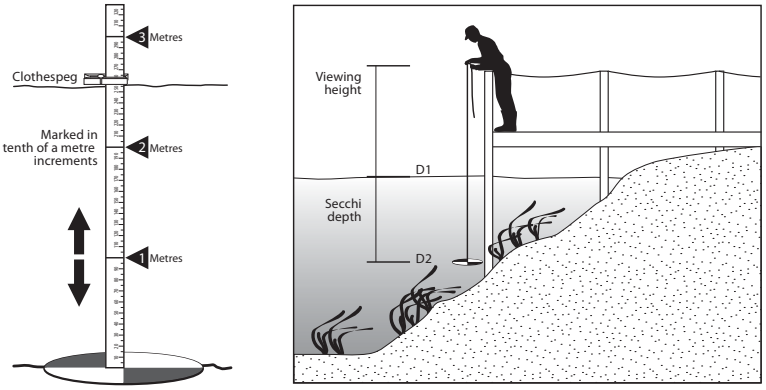
Reduced water clarity can affect marine plant growth and reduces the depth range and extent of seagrass and macrophyte beds.

Equipment: secchi disk, recording sheets

Procedure

1. Lower the secchi disk on its measuring cord until it touches the surface of the water. Record this length on your recording sheet as D1.
2. Continue to lower the secchi disk into the water until you can no longer see clearly.
3. Slowly raise the secchi disk until you can just distinguish the black and white markings on it. Record this length on your recording sheet as D2.
4. Subtract D1 from D2 to obtain the secchi depth.
5. If the disk touches the bottom of the estuary before becoming unclear, record the secchi depth as greater than the estuary depth.

Taking a secchi depth reading



D2 – D1 = secchi depth

Worked example

Kyle lowers his secchi disk from a jetty. When the disk touches the surface of the water, he records the distance as 1.6 m (D1). Kyle then lowers the disk into the water until he can no longer see the black and white markings. He slowly raises the secchi disk until he can just distinguish the markings. He notes the distance as 3.3 m (D2). Using the equation above, Kyle determines the secchi depth to be 3.3 – 1.6 = 1.7 m.

Kyle conducts the same test at another location, from a boat. He lowers the secchi disk over the side of the boat (D1 = 0 m), and he can still make out the secchi markings when the disk touches the bottom of the estuary. He records this depth as 2.4 m, so the secchi depth is recorded as > 2.4 m.

For the relationship between turbidity and secchi depth the following provides a rough guide in metres:
⩾4 m = 7 NTU 2 m =10 NTU 0.9 m = 25 NTU 0.2 m = 100 NTU

Collecting water samples for further analysis – *chlorophyll a* and *enterococci*

Some water quality tests cannot be done by community groups. They may require specialised equipment only available in laboratories, or tests may be dangerous or difficult to conduct. When community groups cannot test water themselves, they may still be able to assist by collecting water samples from their regular monitoring sites for further analysis elsewhere. Two such tests are those for chlorophyll *a* and enterococci. Some general information about these tests and how to collect samples is provided below.

Please contact your local Waterwatch Coordinator to arrange for laboratory testing, procedures for sample collection and further information about the chlorophyll *a* and enterococci tests.

Chlorophyll a – what is it and why is it important?

Chlorophyll *a* (Chl *a*) is the green, photosynthetic pigment found in plants, macroalgae and phytoplankton growing in estuaries. Chl *a* is essential to plants by being active in the capture of light energy for photosynthesis. Concentrations of Chl *a* can be used as an indirect measure of the concentration of phytoplankton cells in the water.

Phytoplankton thrives on nutrients from the surrounding water, and subsequently transforms light and nutrients into plant matter. As such it is the main contributor to primary productivity in the estuarine environment.

Phytoplankton concentrations are generally low in nutrient-poor estuaries and high in nutrient-rich estuaries. Therefore, measuring Chl *a* can provide a test for nutrient levels.

High concentrations of phytoplankton are detrimental to an estuary's functioning as they have an adverse effect on dissolved oxygen and water clarity, whilst some blooms may be directly toxic to other aquatic organisms.

Sampling method

There is no field test for Chl *a*; instead, samples are collected using a sterile sample bottle (1 litre). It is important that the turbidity or secchi depth is also measured at the same time.

When sampling for Chl *a*, avoid collecting surface water or water from the bottom near sediments. Sample from at least 1 m depth. If your waterway is shallow, aim for mid-depth. Wrap the bottle in foil immediately to prevent light penetration and keep it on ice for transport to a laboratory for analysis.

In the laboratory, specific chemicals are used to destroy the algal cells and dissolve the Chl *a* into a solution. This is then put into a spectrophotometer to determine the concentration of the pigment.

Though samples can be frozen for up to a month before analysis, the logistics of storage and transportation may make it difficult to conduct this test successfully, particularly in remote areas.

Source: Adapted from *Waterwatch Australia National Technical Manual, module 7*.

Enterococci – what are they and why are they important?

Bacteria are present in all environments, and form an essential part of the detrital cycle; however, not all bacteria are benign. Water contaminated with faecal matter from people or other warm-blooded animals like cattle can carry bacteria that pose a risk to humans through swimming or eating contaminated shellfish. Unfortunately, many of these pathogens (disease-causing bacteria and viruses) are very difficult to detect in water samples, as they may only be present in small numbers, yet still pose a danger to human health.

Fortunately, other organisms which live in faecal matter in huge numbers, can be used as 'indicators' of faecal contamination. Some of the more common bacteria may also cause disease themselves, but their main importance in waterway monitoring is that they warn us of the presence of more harmful pathogens.

The National Health and Medical Research Council's (NHMRC) 2008 guidelines advocate enterococci as the preferred indicator organisms for the detection of faecal contamination in recreational waters (see www.environment.nsw.gov.au/beach/thewaterqualityguidelines.htm).

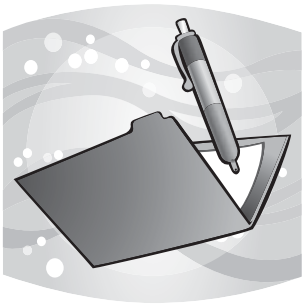
Enterococci are also found in the intestines of warm blooded animals and are present in very high numbers in raw sewage; (millions of enterococci bacteria can be present in just 100 mL). Studies have found a strong relationship between elevated levels of this bacteria and illness rates in swimmers. It is important to note that enterococci do not cause illness, rather, they are measured to detect the presence of sewage and hence the possible presence of harmful pathogens.

Sampling method

The in-field procedure for measuring coliforms such as enterococci is complicated and lengthy. Instead, we suggest that samples are collected using a sterile sample bottle (1 litre), then kept on ice for transport to a laboratory for analysis.

When sampling, avoid sampling surface water or water from the bottom near sediments. Sample from at least 1 m depth. If your waterway is shallow, aim for mid-depth.

Source: *Waterwatch Australia National Technical Manual, module 7* and DECCW Beachwatch, see www.environment.nsw.gov.au/beach/Monitoringtest.htm.



SECTION 3

Interpreting your results

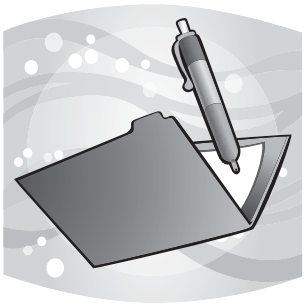
The Australian and New Zealand Environment Conservation Council (ANZECC) has developed guidelines for classifying the quality of water in rivers, lakes, estuaries and marine waters. Waterwatch has also developed guidelines for linking water quality parameters to the health of ecosystems.

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 estuary. 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.

Included in this section: *Page*

3.1 Water quality guidelines



Water quality guidelines are recommended values or ranges for a water quality parameter. They help to identify when changes in a parameter have the potential to cause an environmental problem. Trigger values are provided for parameters in the ANZECC guidelines. If a measured parameter reaches or exceeds the trigger value, risk of environmental or ecosystem harm increases. Waterwatch provides an assessment of water quality for each parameter at an estuary site, based on both water quality and ecosystem health.

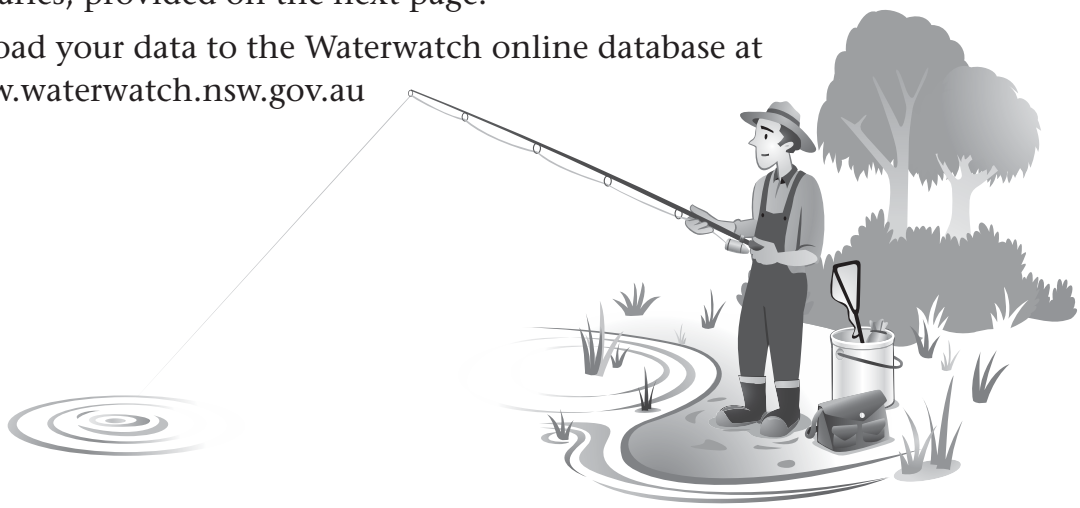
Estuaries	**Healthy**	**Fair**	**Poor**
Temperature °C	No trigger values (affected by tides)		
pH	7–9.0 Estuary >8.5 Marine	N/A	<7 or >9.0
EC µS/cm or mS/cm	No trigger values (affected by tides)		
Turbidity NTU	⩽10	>15–20	>20 (may be influenced by tides)
Avail. PO_4 mg/L	⩽0.02	>0.02–0.3	>0.3
DO % sat.	80–110	No trigger value	<80 or >110

Applying the ANZECC guidelines for estuaries

Step 1: Conduct water quality testing using the procedures and methods outlined in the *Waterwatch Field Manual* or *Junior Waterwatch Field Manual*.

Step 2: Analyse your results using the ANZECC guidelines and trigger values for estuaries, provided on the next page.

Step 3: Upload your data to the Waterwatch online database at www.waterwatch.nsw.gov.au

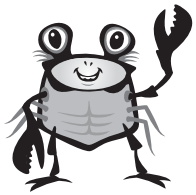


Analysing the results – ANZECC guidelines (2000)

Temperature **°C**	**pH**	**Electrical conductivity** **µS/cm or mS/cm**	**Turbidity** **NTU**	**Available phosphate (mg/L)**	**Dissolved oxygen** **(% saturation)**	**Enterococci** **(CFU/100ml)**
Stressor: change to indicator (+ or –)	**Stressor:** change to indicator (+ or –)	**Stressor:** increase to indicator (+)	**Stressor:** increase to indicator (+)	**Stressor:** increase to indicator (+)	**Stressor:** change to indicator (+ or –)	**Stressor:** increase to indicator (+)
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)	**Effect:** loss of native plants and animals
Trigger values do not exist due to tidal influence	**Trigger value:** **Estuaries:** 7.0–9 **Marine:** 8.0–9	Trigger values do not exist due to tidal influence eg expected to be salty	**Trigger value:** **Estuaries:** 10 NTU **Marine:** <10 NTU	**Trigger value:** (The figure below represents 3.06 times the filterable reactive phosphorus calculated within the ANZECC guidelines) **Estuary:** 0.02 mg/L	**Trigger value:** **Estuaries:** 80–110% **Marine:** 90–110%	**Trigger value:** **Estuary & marine** **Primary contact** <35 CFU /100ml **Secondary contact** <230 CFU /100ml

3.2 Junior estuary field result sheet

Turbidity:_____ NTUS



☐ Less than 10 NTU Healthy

☐ Greater than 10 NTUS
May affect aquatic plants and animals

Note: If secchi depth is use, ⩾4 m = 7 NTU, 2 m = 10 NTU, 0.9 m = 25 NTU, 0.2 m = 100 NTU.

Water quality trigger values

Trigger value: the point where a change in water quality can affect estuary health.

Parameter 	**Temp. °C**	**pH** (0 – 14) Increase or decrease may affect waterway health	**EC** µS/cm OR mS/cm	**Turbidity** (NTU) Increase may affect waterway health
Trigger value	No triggre value	7 – 9 (Estuary) 8 – 9 (Marine)	No trigger value	10 NTUs (Estuary) 10 NTUs (Marine)

Ref: ANZECC Guidelines 2000

Waterwatch NEW SOUTH WALES
Communities Caring for Catchments

Junior estuary field result sheet

Note: Only use this recording sheet for basic parameters in tidal streams and estuaries

Site name:_____

Date: _____ **Time:** _____

Type of site: (e.g. lake, lagoon, tidal stream):

_____

Number sampling:_____ **Time taken:** _____

Rainfall: (tick the box if rainfall occurred)

☐ Today ☐ Previous 24 hours ☐ Previous week

Observations at the site:

_____

_____

_____

_____

_____

Record your water quality results on the Waterwatch database as soon as possible after testing.

www.waterwatch.nsw.gov.au

waterwatch NEW SOUTH WALES
Communities Caring for Catchments

Temperature & tides:

Results: Water temp. _____ °C Air temp. _____ °C

Time: _____ AM/PM **Approx. depth sampled:** ____ m

Tide: Mark on the rectangle the height of the tide when testing.

Low [ ] High

← Going out Coming in →

Rate of flow: Tick the box that best describes rate of flow.

☐ Fast ☐ Slow ☐ Standing water

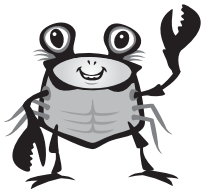
pH: _____

← →

1 Acid	Neutral 7	Alkaline 14

Results: ☐ Acid ☐ Neutral ☐ Alkaline

☐ 7–9 Healthy



☐ <7 or >9 Not healthy

EstuarySalinity: (use a high range EC meter)

The sample was ☐ Not diluted ☐ Diluted

For diluted samples:

The dilution factor = (one part sample water + number parts deionised water) = _____

Results: reading on screen (mS/cm) _____ x (dilution factor) = _____ x 1000 (convert to µS/cm) = _____ **µS/cm)**

Tick the box below that matches your result:

Freshwater

☐ <1500 µS/cm (<1.5 mS/cm)

Only very salt tolerant freshwater species as salinity levels rise.

Brackish (freshwater mixed with some salt water)

☐ 1510–4800 µS/cm (1.5–4.8 mS/cm)

Only very salt tolerant freshwater and estuary species.

Estuary

☐ >4800 µS/cm – 51,500 µS/cm (>4.8–51 mS/cm)

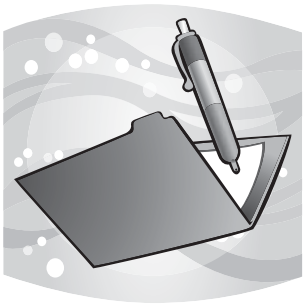
Estuary species and habitats.

Marine

☐ Greater than 51,500 µS/cm (>51 mS/cm)

Marine species and habitats.

3.3 Senior estuary field result sheet



Group: _____

Date sampled: _____ **Time sampled:** _____ AM / PM

Site name:_____

Number of volunteers: _____ **Time taken:** _____ (hours) _____ (minutes)

Tide: ☐ high ☐ low ☐ flood (coming in) ☐ ebb (going out)

Rainfall last week: ☐ within 24 hours ☐ 1–7 days

Rainfall description: ☐ light ☐ medium ☐ heavy

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

Calibration solution: High range meter:_____mS/cm

Note: In an estuary, salinity should always be measured using a high range EC meter.

Flow: ☐ dry ☐ no flow or standing water ☐ low flow ☐ high flow ☐ flood

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

_____

_____

_____

_____

_____

_____

_____

_____

Test	**Units**	**Results**	**Result range***	**Guidelines** *(tick the box)*
Temperature	ºC		No trigger value	No trigger value
pH	1–14 pH units		<7	☐ poor
			7–9.0	☐ healthy
			>9.0	☐ poor
Electrical conductivity (salinity) (Reading from meter x dilution factor)	µS/cm		No trigger value	No trigger value
Turbidity**	NTUs		<10	☐ healthy
			10–20	☐ fair
			>20	☐ poor
Available phosphate	mg/L		<0.02	☐ healthy
			0.02–<0.3	☐ fair
			>0.3	☐ poor
Dissolved oxygen	mg/L		<80	☐ poor
	% saturation		80–110	☐ very good
			>110	☐ poor
Enterococci	colonies/100mL		<153	Primary contact
			<230	Secondary contact
Chlorophyll *a*	µg/L		⩽4	☐ healthy
			>4	☐ poor
Colour	Coordinates from chart		No applicable	

* Adapted from *ANZECC Guidelines 2000*

** Note: If secchi depth is used, ⩾4 m = 7 NTU, 2m = 10 NTU, 0.9 m = 25 NTU, 0.2m = 100 NTU

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

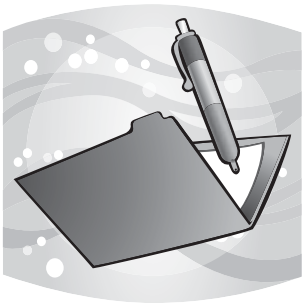
3.4 Continuous estuary result sheet – one site over time

Site name: _____

Date:						
Time:						
Rainfall last week: (nil, last 24 hours, in last week)						
Rainfall description: (light, medium, heavy)						
Observations: (including colour of water)						
Tide: (high, low, flood (coming in), ebb (going out))						
Flow: (dry, no flow, low, high, flood)						
Number of volunteers:						
Time taken monitoring:						
Air temperature: (°C)						
Water temperature: (°C)						
pH: (units)						
Electrical conductivity (EC)						
EC meter calibration: If meter is not calibrated results should not be recorded	☐ YES ☐ NO	☐ YES ☐ NO	☐ YES ☐ NO	☐ YES ☐ NO	☐ YES ☐ NO	☐ YES ☐ NO
EC meter reading: on screen (mS/cm):						
Dilution factor: (one part sample water + the number of parts deionised water)						
EC final result: (uS/cm) Meter reading (mS/cm) x dilution factor x 1000						
Turbidity: (NTU):						
Water clarity: (depth in metres)						
Available phosphates: (mg/L)						
Dissolved oxygen: (mg/L)						
Dissolved oxygen: (% saturation)						
Enterococci: (CFU/100mL)						
Chlorophyll *a*: µg/L						
Other comments:						

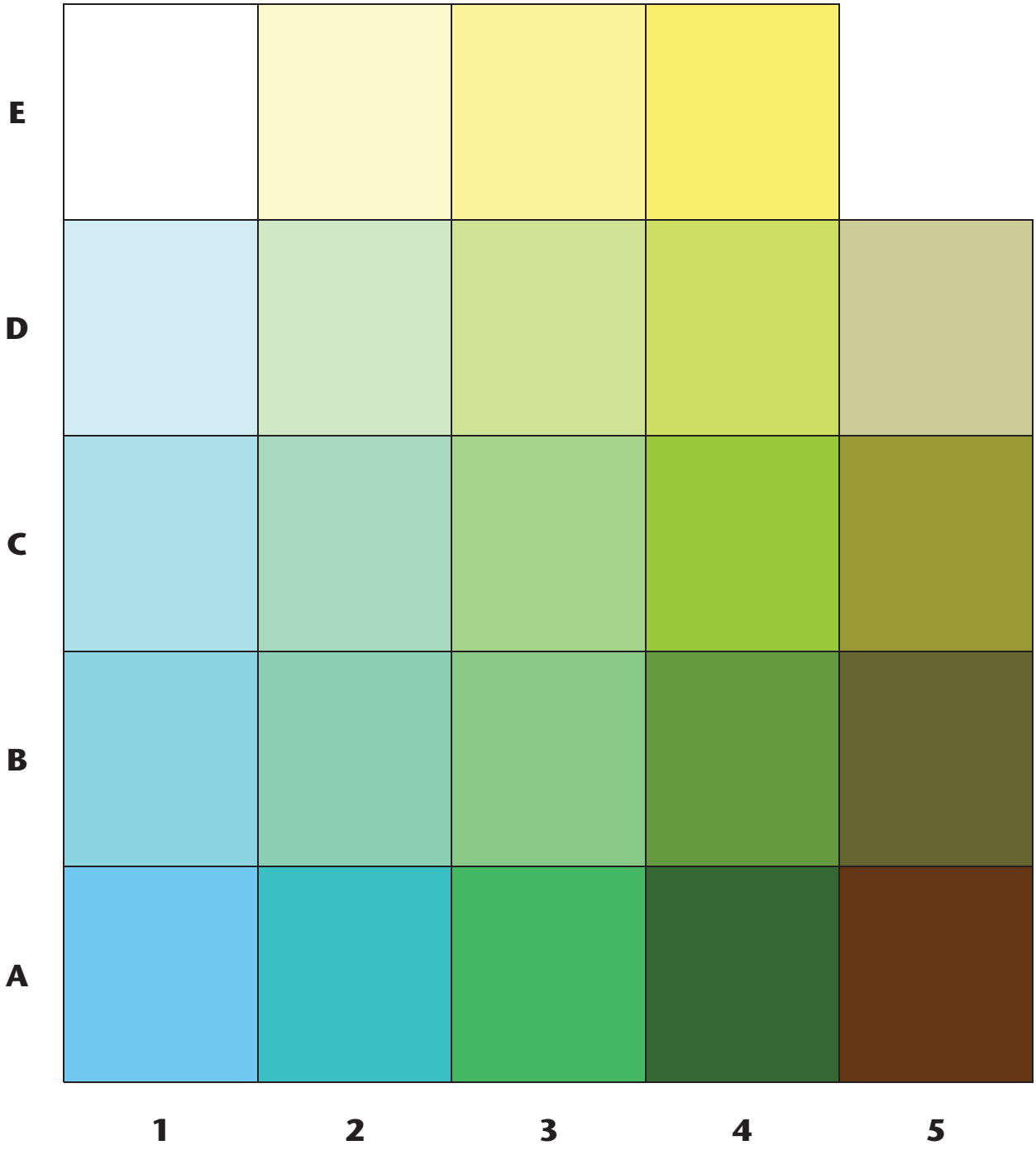
Upload your data as soon as possible after testing to www.waterwatch.nsw.gov.au.

3.5 Water colour chart



Compare and identify the colour of the water at your site with the colour chart provided. This information can be included as observations in your recording sheet.

Use the letters and numbers to identify the colour from the grid. For example, D5 (light olive green).



Note: This page will need to be printed in colour for the chart to be usable.

SECTION 4

Investigating habitats

Investigating estuarine habitats involves gaining an understanding of the influences and processes that have helped to shape the estuary. Habitat assessments build skills in observation and recording and involve measurements that help to track changes. Taking photographs or sketching details of flora and fauna provides qualitative data, which is also useful information for tracking changes over time and providing an indication of the health of an estuary.

Please remember to ensure you have minimal impact on the ecosystems you are investigating and 'tread lightly'.

This section explains the key skills and methods that can used to monitor habitats within estuaries. By mastering these skills and incorporating them within your monitoring plan you can make a valuable contribution to understanding ecosystem change and help manage the catchment.

Included in this section: *Page*

4.1 Essential skills: photopoints

Photos provide a visual record of a site over time and can show evidence of change. Make sure photographs are always taken in the same place, so they can provide a record of change over time. Ensure photopoints are upstream and downstream of monitoring sites, in different seasons, to record both natural and anthropogenic (human-caused) changes over time.

The way to achieve a meaningful photographic record is to choose a prominent landmark or structure (such as a fencepost) and include this in the photo, or clearly mark the photopoint with a star picket or other resilient structure. Close-ups can show more detail. Use the highest resolution on the camera, to allow magnification and good quality printing.

The *Waterwatch Field Manual* and the *Junior Waterwatch Field Manual* both provide worksheets with further information about establishing photopoints on site.

4.2 Essential skills: transects

Observing changes at your site can be done using a transect, which is a line between two points. This can be an extended tape measure, or the edge of a lake, stream or boardwalk.

There are two types of transect:

- A line transect is one where individual plants or invertebrates touching a measuring tape stretched across an area are recorded at regular intervals.
- A belt transect is one where a quadrat frame is placed next to the tape (see Section 4.3). This provides a closer look at species composition across your site.

Establishing 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. Lay out the transect, set the compass to the bearing and note a feature or landmark on the same bearing.
4. Tie the end of a tape measure to the transect marker and walk in a straight line with the tape following the bearing (walk directly towards the feature noted).
5. Stop when the length of the transect or the boundary of the study area has been reached.
6. If using quadrats, place them at fixed intervals to one side of the tape and walk along the opposite side recording the information contained within each quadrat.

When conducting transect surveys over a period of time you may either return to the same locations you previously conducted transects and place quadrats at the same intervals (plotted transects and quadrats), or you my place quadrats randomly within a given area (random quadrats within a set boundary).

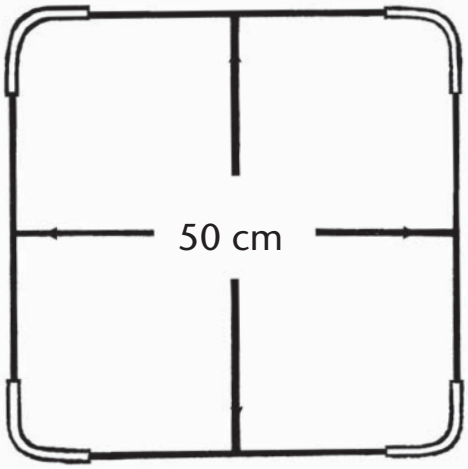
Adapted from: *Waterwatch Australia, National Technical Manual*, module 7, 2006

4.3 Essential skills: quadrats

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 is a square, the internal height and width of which is usually 50 cm. It is made using PVC pipe joined together with elbow connectors. When making the quadrat, the length of pipe will need to be slightly less than 50 cm to take account of the elbow connectors. Cut the pipe to ensure a finished internal size of 50 cm square.

Quadrat construction

A quadrat can also be made using a plastic hoop. These are light weight and inexpensive.



Quadrats can be used to determine:

- **Percentage cover:** Assuming there are 100 parts within a quadrat, the percentage cover is the number of parts covered.
- **Species composition:** This is the visible percentage of plant cover represented by each species within the quadrat; or the number of organisms of each species within the quadrat.

Photographing quadrats

- When photographing quadrats, remember to use a bird's eye view (looking down).
- Photographs should be taken from an angle as close to vertical as possible, and should include the entire quadrat frame with the tape measure beside it.
- Avoid having any shadows or reflection off any water in the field of view.

Quadrat studies

Studies of plants and other organisms in an estuary can be conducted effectively using a quadrat survey.

There are 2 ways to conduct these surveys:

- plotted transect and quadrats
- random quadrats within a set boundary.

Equipment: Quadrat (50 cm x 50 cm) or plastic hoop (roughly 120 cm diameter), 30–50 m fibreglass tape measure, field identification guides, recording sheets, camera, suitable footwear.

Plotted transect and quadrats

Step 1: Work in groups along a line about 10–50 m long.

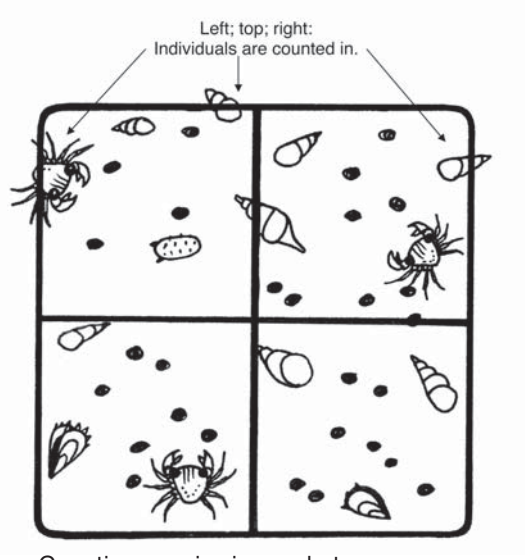
Step 2: Lay down quadrats to the right of the line at fixed intervals.

Step 3: On the recording sheet, enter the data relevant to your survey, such as:

- a count of the number of plant and animal types inside each quadrat
- other measurements relevant to your survey such as the height of the majority of the plants in each quadrat
- for plant studies, estimates of the percentage of plant matter (including dead plants) compared to bare ground in the quadrat.

Tips when counting species inside quadrats

- If part of a plant is inside the quadrat, the plant is included as part of the quadrat count.
- If counting animal species, only count living species.
- When determining the health of plant species, compare the health of plants to surrounding plants of the same species outside the quadrat.



Counting species in quadrats

Random quadrats within a set boundary

Step 1: Identify an area to be sampled. This should be representative of the estuary. Determine the boundary of the area using landmarks.

Step 2: Estimate the total area to be sampled (area= length x width).

Step 3: Lay down quadrats randomly within the area.

Step 4: On the recording sheet, enter the information required for your study, such as:

- a count of the number of different plant species and animal species inside each quadrat
- other measurements relevant to your survey such as the height of the majority of the plants in each quadrat
- for plant studies, estimates of the percentage of plant matter (including dead plants) compared to bare ground in the quadrat.

4.3 Essential skills: quadrats

Calculating the area of a quadrat

1. Calculate the area of each quadrat:

   Area of square quadrat = L^2 = _____ m (L = length)
   Area of circle = πr^2 = _____ m (r = radius)

2. To determine the total area assessed, multiply the area of the quadrat by the number of quadrats:

   Total area assessed = area of quadrat x number of quadrats = _____ m^2

3. Calculate species diversity

Species names	**How many of each species?**
1.	
2.	
3.	
4.	
5.	
6.	
7.	
8.	
9.	
10.	
Total number of species (A):	**Total number of organisms (B):**

Diversity = $\frac{\text{Total number of species counted (A)}}{\text{Total number of organisms counted (B)}}$

Your answer should come to between 0 and 1. The highest diversity is represented by answers close to 1 and the lowest diversity by answers close to 0.

Lowest diversity	Low – medium	Medium – high	High diversity	Highest diversity
0.1	0.25	0.5	0.75	1

4. Calculate **species abundance** =

   $\frac{\text{Number of species}}{\text{Total area assessed}}$ = species per m^2

i **A healthy estuary would generally have high levels of species diversity and species abundance, although this is also dependent on the type of habitat, e.g. saltmarsh, mangroves, intertidal mudflats etc.**

Estuary transect

SECTION 5

Habitat zones within estuaries

Estuaries have a unique combination of habitats that are influenced by both marine and freshwater. It is a dynamic zone where plant and animal species are adapted to daily variations in water levels and salinity.

This section provides an overview of habitat zones within estuaries and provides a range of assessments to monitor riparian vegetation and mangroves.

Included in this section: *Page*

5.1 The two main habitat zones found in estuaries

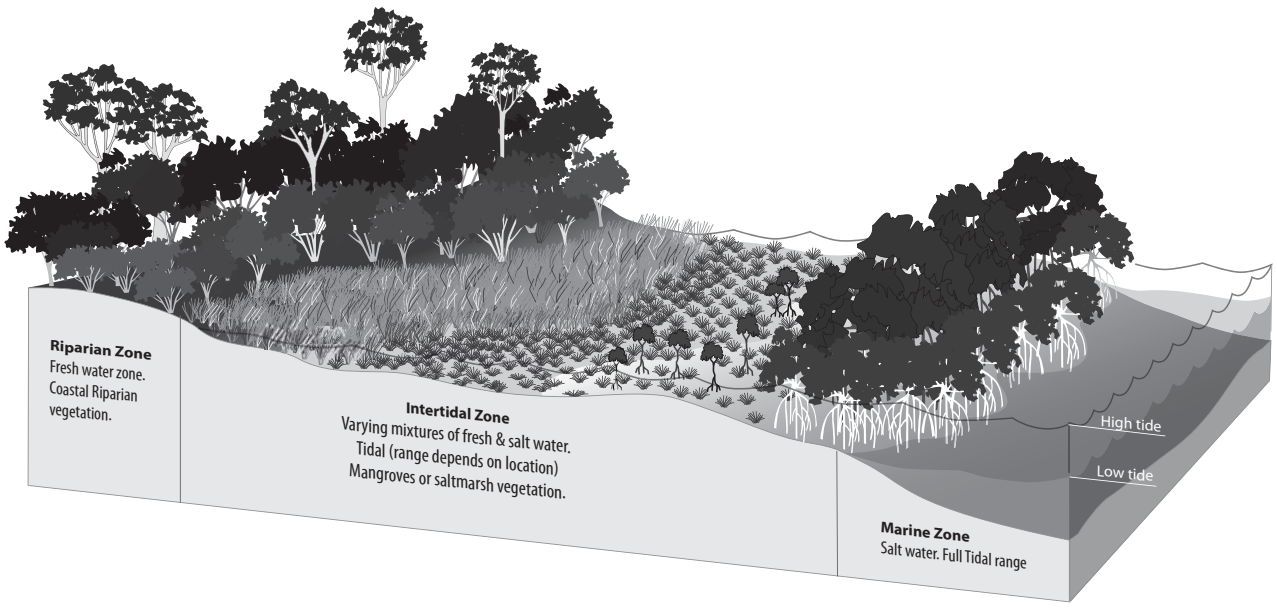
A habitat is a place that provides food and shelter for living things. In an estuary habitat changes occur due to the influence of fresh and salt water. The salinity of the water determines whether the habitat provides food and shelter for fresh or saltwater species, or organisms adapted to both. Hence, the influence of tides within estuaries affects the types of plant and animal species found there.

There are two main habitat zones within estuaries:

- the **riparian zone** located above the high tide level
- the **intertidal zone** between the high and low tide levels, which includes mangrove and saltmarsh habitats.

Habitat	**Features**
Intertidal zone	
Seagrass/mudflats	Covered by tides for most of the time. May be uncovered twice each day for short periods
Mangroves	Covered by tides twice each day and exposed for greater periods of time
Saltmarsh	Covered by tides for shorter periods less often
Riparian vegetation	Rarely covered by tides unless from an extreme weather event (floods)

Typical habitat zones within estuaries are illustrated in the cross-section below.



5.2 Riparian vegetation

'Riparian' means adjacent to, or in contact with a waterway. In an estuary, riparian vegetation is the vegetation above the high tide level. This means that the riparian area does not include mangroves and saltmarsh.

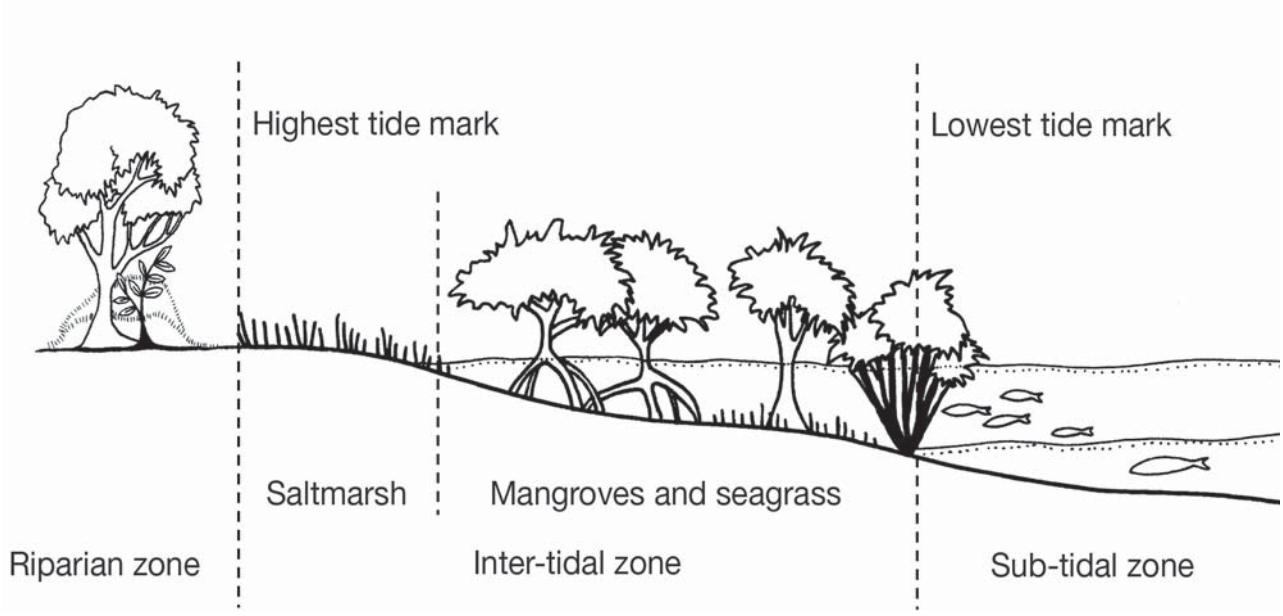
Riparian vegetation is important in estuaries. It prevents erosion, provides habitats for birds and animals and reduces the transfer of pollutants into the estuary. In coastal areas, the high level of human use has modified these areas causing the removal of riparian vegetation or the invasion of these areas by pest and weed species. This causes changes to the dynamic interactions within estuarine ecosystems.

Assessment of riparian vegetation provides valuable information about the health and stability of the estuary. This information can inform the decisions of natural resource managers and identify priority areas for protection, management or restoration.

Riparian condition assessment

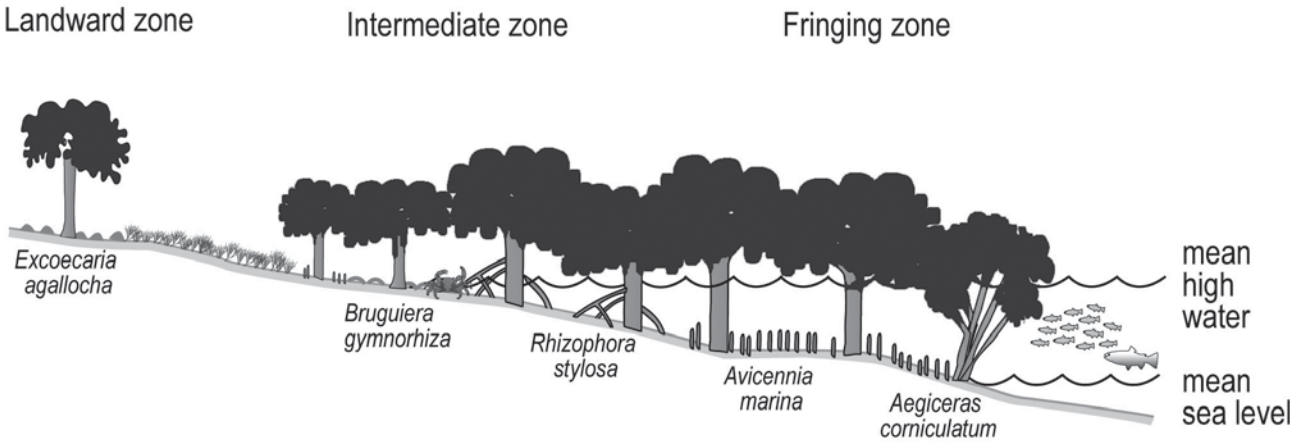
Level of difficulty: Challenging
Suitable for community groups

Conduct the riparian condition assessment as outlined in the *Waterwatch Field Manual* (Section 9). This assessment can be uploaded to the Waterwatch online database.



5.3 Intertidal habitats: mangroves

Mangrove forests vary considerably between different forest types, and between the same forest types in different locations. They are influenced by natural factors including climate, tidal inundation, soil pH and salinity, sediment type and the amount of freshwater, which can vary with rainfall and tidal limits.

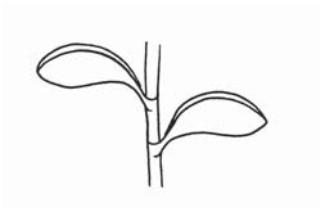


Source: NSW Department of Industry and Investment

Mangroves provide an indicator of estuary health as they respond slowly to changes within the estuary. Positive changes can result in greater forest vigour such as increased canopy cover and tree regeneration, while negative changes can stress the mangrove community, resulting in reduced canopy cover, lower canopy height or tree death. Monitoring changes in the structure of mangrove plant communities over time can provide an indication of the effectiveness of management actions and the impact of human activity on mangrove plant communities.

Monitoring mangroves may include the assessment of forest structure such as changes in canopy height, age, tree circumference and the species represented. These factors have been incorporated into a rapid site assessment that will enable Waterwatch groups to monitor key changes in forest structure and type over time as part of their monitoring plan.

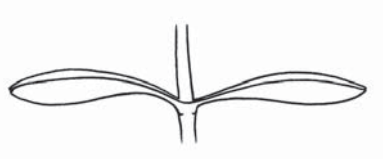
5.4 Key to common mangrove species



Mangrove type

Leaves alternate

Leaves opposite



Is sap milky?

Yes

Milky mangrove *Exoecaria agallocha*

No

Are salt crystals present on leaf surface? Is the leaf spoon shaped? Is length to width 2:1

Yes

River mangrove *Aegiceras corniculatum*

No

The tree you are trying to identify is not common to NSW

Are leaves grey on the underside

Yes

Grey mangrove *Avicennia marina*

No

Does the trunk have aerial prop roots? Are there black or brown spots on the underside of leaves?

Yes

Red or stilted mangrove *Rhizophora stylosa*

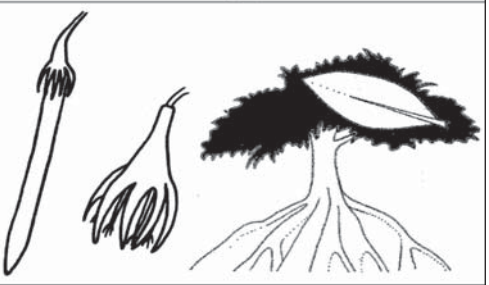
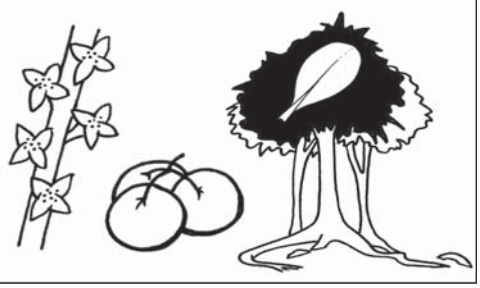
No

Is the bark hard and furrowed? Does the tree have knee roots?

Yes

Large leaved or orange mangrove *Bruguiera gymnorrhiza*

5.5 Mangrove identification chart

NAME	Distinguishing Features	Distribution in NSW	Examples
River Mangrove *Aegiceras corniculatum*	• Occurs in fringing tidal zone • Smooth dark brown bark • Shrub/multi-stemmed tree (1-3 metres) • No above ground roots • Alternate oval leavea (>7cm); rounded tips • Curved tapering fruit with pointed tip • Salt deposits on leaves	Tweed River to Merimbula River	
Grey Mangrove *Avicennia marina* (variety *australasica)*	• Occurs in fringing to intermediate tidal zone • Smooth, pale grey bark • Tree (2-12 metres) • Pneumatophores (vertical roots) • Opposite pointed leaves; pointed tip; pale grey undersurface • Small pale orange, scented flowers • Bulb like fruit (1-4cm)	Entire Coast	
Red/Stilted Mangrove *Rhizophora stylosa*	• Occurs in fringing to intermediate tidal zone • Grey/black rough bark • Tree (2-8 metres) • Prop/stilt roots • Opposite dk green glossy leaves (10-15cm); broad with sharp abrupt point • Yellow white flowers with hair petals • Long green/brown fruit; narrow; tapered (20-30cm)	Tweed River to Macleay River	
Large Leaved Mangrove *Bruguiera gymnorhiza*	• Occurs in intermediate to landward tidal zone • Dark brown rough bark • Tree (2-6 metres) • Buttress trunk and exposed knee roots • Opposite large leaves (10-20 cm) • Red flower; orange petals • Green cigar shaped fruit (10-20 cm)	Tweed River to Clarence River	
Milky/Blind-your-eye Mangrove *Excoecaria agallocha*	• Occurs in intermediate to land-ward tidal zone • Grey slightly rough bark • Tree (2-6 metres) • Emergent roots • Alternate leaves (6-10cm) • Exudes milky sap • Male and female flowers on different trees • Fruit has peppercorn like seeds	Tweed River to Manning River	

5.6 Field identification guide: common mangrove species

Grey mangrove *Avicennia marina*

Grey mangrove *Avicennia marina* pneumatophores

River mangrove *Aegiceras corniculatum*

Blind-your-eye or milky mangrove *Excoecaria agallocha*

Red or stilted mangrove *Rhizophora stylosa*

Large leaved mangrove (orange mangrove) *Bruguiera gymnorrhiza*

Photographs reproduced courtesy of the NSW Department of Industry and Investment.

5.7 Mangrove plant study (junior students)

Level of difficulty: Easy
Suitable for primary schools

Equipment: recording sheets, ruler or pencil

The mangroves at my site could be described as:

☐ trees (>2 metres)

☐ shrubs (<2 metres)

Estimate the height of mangroves:

_____ metres

Draw a picture of a mangrove at your site. Label the main features including the leaves and roots

Mangrove leaves

Draw the leaf of a mangrove.

Describe leaf colour on top:

_____

Describe leaf colour underneath:

_____

Using the information in Sections 5.4 to 5.6, name this mangrove:

_____

Mangrove roots

Why do you think some mangrove roots are called 'snorkel roots' or 'Pencil roots'?

Draw animals you can see living in and around mangroves.

What may happen to mangroves if sea levels rise?

5.8 Mangrove plant study (senior students)

Level of difficulty: Moderate

Suitable for secondary schools

Equipment: recording sheets, pen or pencil, mangrove key

Use the information in Sections 5.4 to 5.6 about common mangrove species to determine the type of mangroves at your site.

Mangrove type:

_____

Draw and label the leaves of the mangroves. Include colour and shape.

Describe the height and shape of mangroves at your site.

_____

_____

_____

Classify the visible roots as:

☐ pneumatophores ☐ arching prop roots ☐ knee roots

☐ trunk roots ☐ radial roots ☐ no visible roots

Prop Roots Trunk Roots Pneumatophores

Describe the health of mangroves at this site:

☐ healthy – dense canopy of green leaves

☐ unhealthy – tree foliage is sparse or yellowing. There may be evidence of canopy die back with bare sticks at the top of the crown

☐ dead or dying – no leaves or leaves are black

How has human activity impacted on mangroves?
(mention helpful and harmful impacts)

What other plant species live in the same habitat?

5.9 Quadrat study: counting mangrove pneumatophores

Level of difficulty: Easy

Suitable for primary and secondary school students

Mangroves have special Pencil-shaped roots called pneumatophores that can be seen above the soil and water. These roots support the plant and at high tide are an important habitat for fish and invertebrates such as crabs and snails.

Note: Mangroves are fragile areas. Take care and tread lightly!

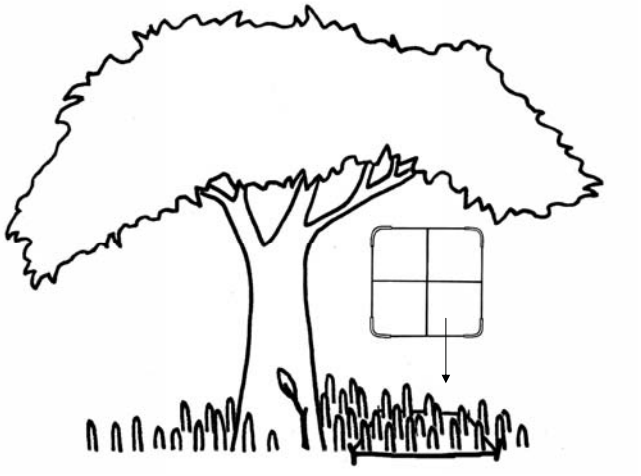
Equipment: quadrat square (or plastic hoop), magnifying glass, mangrove recording sheet

Procedure

Refer to Section 4.3 for information about setting up and recording information within quadrats.

1. Carefully place the quadrat (or hoop) over the pencil roots close to the base of the tree.
2. Count the number of pencil roots (pneumatophores) inside the quadrat.
3. Look carefully inside the quadrat. Count the small animals that live there.
4. Classify the animals using the information in Section 8 of this manual.
5. Fill in the recording sheet.

Draw **one** animal that lives in this habitat.



5.10 Mangrove recording sheet

Site name:

Date: **Time:**

Tide *(tick)*: ☐ high ☐ low ☐ coming in ☐ going out

Method of collecting data *(tick the box below)*:

☐ Plotted transect/quadrats
Transect length (metres):_____ Number of quadrats _____
Quadrat spacing (metres): _____

☐ Random quadrats within a set boundary: Number of quadrats _____
Area within boundary (metres square):_____

Shape of quadrat *(tick)*: ☐ square quadrat ☐ round (hoop) quadrat

Quadrat	Count the numbers within each quadrat					
	Pneumatophores	Saplings	Crab holes	Molluscs	Woody debris/ leaf litter	Human litter
1						
2						
3						
4						
5						
6						
7						
8						
9						
10						
TOTALS						
Total m^2						

Refer to Section 4.3 for information about calculating the area of a quadrat and reporting numbers per square metre (species diversity).

5.11 Mangrove assessment

Level of difficulty: Moderate

Suitable for community groups and secondary students

Background

This assessment is used to identify the features of mangroves at the site and the risks to mangrove condition.

The assessment is based on observations that assess mangrove species composition, cover, regeneration and health whilst recording the features of mangroves.

By assessing these features over time, changes to the character and structure of mangrove forests can be identified.

Step 1: *Site selection*

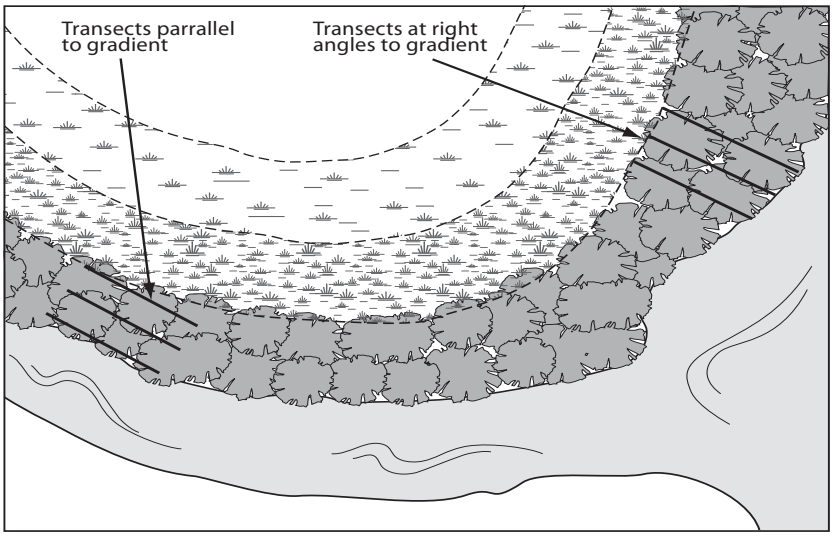
Select sites that have the same physical attributes and are representative of the mangroves in the area.

Conduct an assessment from the seaward side of the mangroves to the landward edge to identify the characteristics of mangroves at the site (i.e. at right angles to the gradient)

OR

Conduct an assessment parallel to a coastal stream or estuary (parallel to the gradient).

Diagram of mangrove transects



Step 2: *Canopy dominance*

Using the information in Sections 5.4 to 5.6, identify the dominant mangrove type:

☐ Grey mangrove ☐ River mangrove ☐ Large leaved mangrove

☐ Blind-your-eye or milky mangrove ☐ Red or stilted mangrove

Step 3: *Mangrove form and height*

Identify the form of the dominant mangrove species at your site.

Form:

☐ Tree >2 metres

☐ Shrub <2 metres with a bush appearance (greater width and more foliage to the ground)

Height:

Estimate the average height of the mangroves.

> **These estimates can be made using the simple method outlined in the *Waterwatch Field Manual*, Section 8.3.**

Note: For a more accurate measurement of height, a clinometer is recommended.

Classify the average mangrove height as:

☐ <2 metres ☐ >2–6 metres ☐ >6–8 metres ☐ >8–12 metres

Step 4: *Root type*

Classify the mangrove roots as:

☐ pneumatophores or vertical roots ☐ prop or stilt roots ☐ knee roots

☐ roots that cannot be seen ☐ radial roots ☐ trunk roots

Step 5: *Soil texture/type*

Collect a sample of the soil from each site where assessments are made. Rub it between your fingers. Record the sediment type based on its feel (e.g. sand, mud, sand/mud, mud/sand):

☐ sand ☐ mud/sand ☐ sand/mud

☐ mud/silt ☐ clay ☐ organic matter

The description below may assist to identify the soil texture.

Soil Type	Description
Sand	Coarse texture and sand grains visible
Muddy sand	Mostly smooth mud with some sand grains visible
Sandy mud	Mostly coarse texture due to sand mixed with a small amount of mud
Mud/silt	Smooth texture with no coarseness or visible sand grains
Clay	Smooth sticky texture; mottled grey colour
Organic matter	Black sediment that is spongy and crumbles easily

Step 6: *Estimate percentage canopy cover*

Assess the percentage cover provided by the mangrove canopy. (This is the amount of sky that is blocked by the mangrove canopy).

Use the estuary plant percentage cover diagram in section 6.6 of this manual.

Tick the box that bes describes the percentage canopy cover of the mangroves at your site:

☐ 0–5% ☐ 5–25% ☐ 25–50% ☐ 50–75% ☐ 75–100%

Step 7: *Mangrove health*

Tick the box that best describes mangroves at your site:

☐ healthy – dense canopy of green leaves

☐ unhealthy – tree foliage is sparse or yellowing. There may be evidence of canopy die back with bare sticks at the top of the crown

☐ dead/dying – no leaves or leaves are black.

Step 8: *Stem circumference*

Classify the diversity of mangroves by stem circumference:

☐ high diversity of stem sizes with evidence of mangrove regeneration

☐ some diversity of stem sizes but few seedlings or saplings

☐ low diversity of stem sizes with little or no evidence of seedlings or saplings.

Note: Stem circumference should be estimated and measured at chest height (or 1.3 metres above the ground).

Step 9: *Regeneration*

Tick the box that describes the regeneration of mangroves at your site:

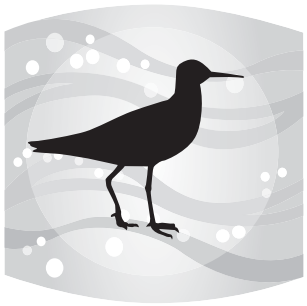
☐ few seedlings and saplings

☐ a significant number of saplings (1–2 metres with thin stem)

☐ a significant number of seedlings (<1 metre with thin stems)

Note: This can be estimated by marking a 10 metre square inside a representative area and counting the number of adult trees, bushes, saplings and seedlings.

SECTION 6



Saltmarsh monitoring

This section provides assessments for monitoring saltmarsh. Investigations of plant cover and species dominance can provide useful information about the health of saltmarsh communities within an estuary.

Included in this section: *Page*

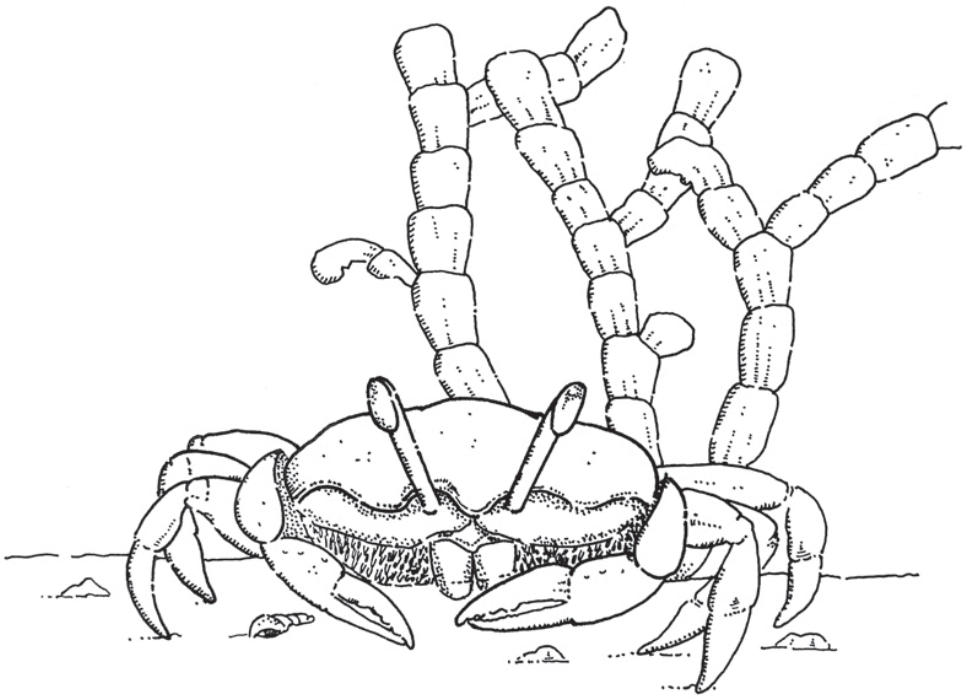


Illustration: Hawkesbury-Nepean CMA, used with permission

6.1 Saltmarsh communities

Saltmarsh plant communities are dominated by herbs, sedges, grasses and low shrubs. The main saltmarsh species found in New South Wales are samphire, salt couch and rushes, such as sea rush, and weeds *Juncus kraussii* and spiny rush, *Juncus acutus*.

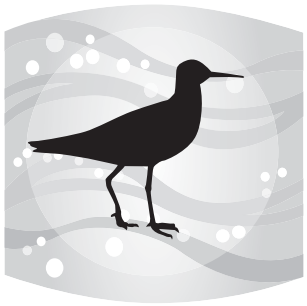
Saltmarsh is generally flooded with salt water during high tide. At low tide, land becomes exposed and it is possible for freshwater to flow into the saltmarsh. This means that creatures living in the saltmarsh are exposed to variations in temperature and moisture and they are also subjected to large changes in salinity. Species that live in this zone have adapted to both aquatic and terrestrial conditions, as changes can occur within the same day.

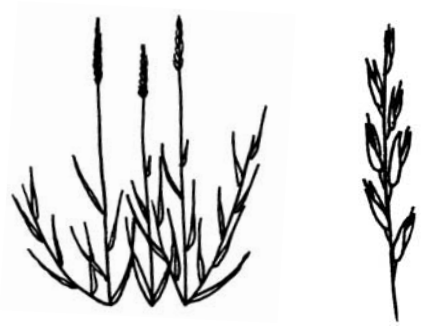
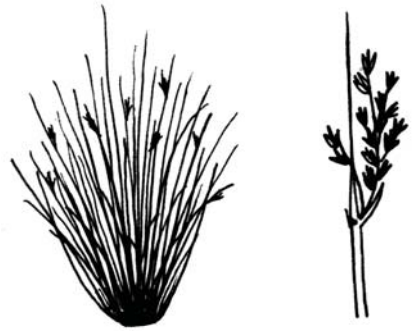
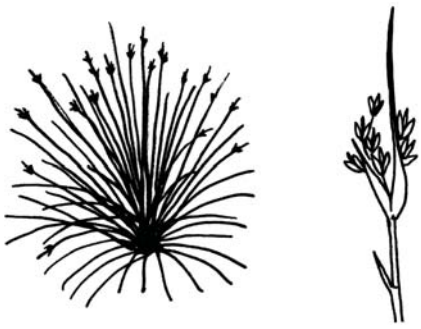
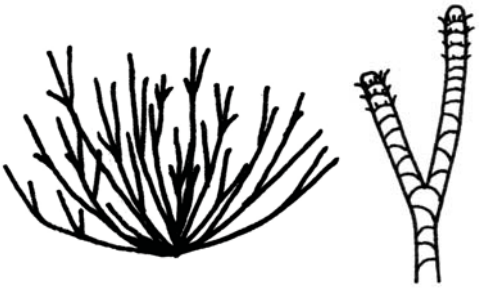
Changes to the distribution and abundance of species within saltmarsh communities can take place due to fluctuations in water quality, soil salinity or flow patterns that may be influenced by structures such as flood gates. A decline in species number and/or distribution may therefore indicate stress, so monitoring change is very important.

Investigations of plant cover and species dominance along a transect line can provide useful information about the health of a saltmarsh.

Illustration: Hawkesbury-Nepean CMA, used with permission

6.2 Saltmarsh identification chart



NAME	Distinguishing Features	Appearance
Salt/Marine/ Sand Couch *Sporobolus virginicus*	• Perennial ground cover grass • Forms dense mat/tufts often spreading over large areas • Grey/green to yellow - greener in the summer months	
Sea Rush *Juncus kraussii*	• Tussock forming perennial • Leaves 40-50cm long • Green with golden/brown sheath • Clustered flowers 4-20cm long: straw to reddish brown	
Spiny Rush *Juncus Acutus* (Introduced)	• Large clumping perennial • Grows to 120 cm tall • Tough leaves and flower stems tapering to sharp points	
Red Samphire *Sarcocornia quinqueflora*	• Perennial herb/shrub (0.1-.05 metres) • Grows in sand; sandy loam; clay; moderately saline soils • Found in swamps; estuar-ies; salt lakes	

6.3 Field identification guide: common saltmarsh species

Samphire *Sarcocornia quinqueflora*

Photograph: Hunter-Central Rivers Waterwatch, used with permission

Marine couch *Sporobolus virginicus*

(Also called sand couch, salt couch, saltwater couch)

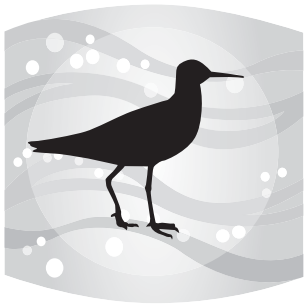
Sea rush *Juncus Kraussii*

Spiny rush *Juncus acutus*

(Introduced species)

Photographs: NSW Department of Industry & Investment, used with permission

6.4 Saltmarsh quadrat survey



Level of difficulty: Moderate

Suitable for secondary students and community groups

Selecting a site

Select a site which is typical of the whole area. This is called a representative site.

Conduct a quadrat survey

Step 1:

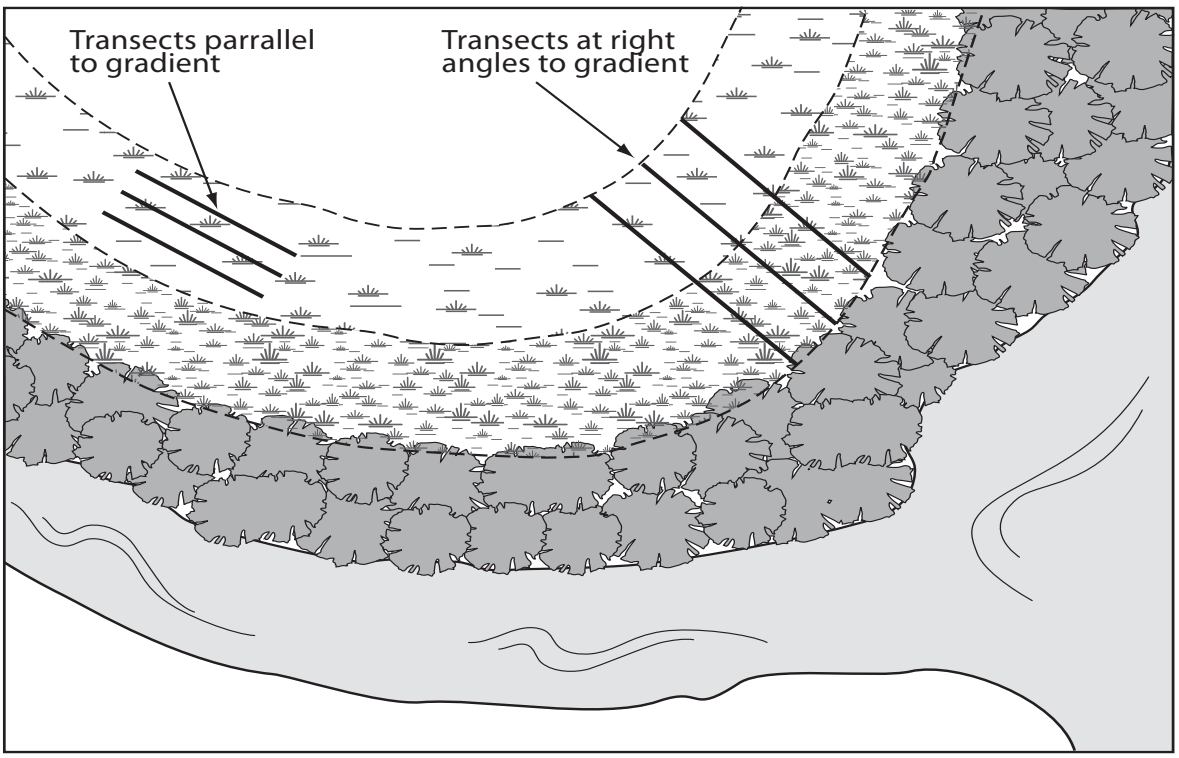
Conduct a quadrat survey following the procedures for plotted transects (see Section 4.3 in this manual).

Note: Conducting this investigation in more than one location will increase the validity of your results. These are called replicates.

The position of the transect will be determined by the purpose of the monitoring:

- **Investigating homogeneous units:** a longitudinal assessment parallel to the edge of the saltmarsh community
- **Investigating a diversity of habitats:** transect is placed perpendicular to the edge of the saltmarsh, beginning at the outer edge and moving inwards.

Diagram of Saltmarsh Transect



Random quadrats in a fixed area can be used as an alternative method for conducting this survey. Procedures for this method are also provided in Sections 4.2 and 4.3.

Step 2:

Count the number of different plant species inside each quadrat.

Measure the approximate height of the majority of the plants in each quadrat.

Do **not** measure the tallest plants. Measurements should be taken of the height of the majority of plants within each quadrat.

Step 3:

Estimate the percentage of plant matter (including dead plants) compared to bare ground in the quadrat.

Note: Look down over the quadrat and use the % saltmarsh cover diagrams to help you.

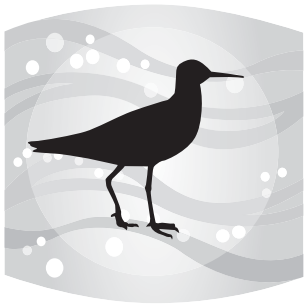
Step 4:

Classify the plants as healthy, crushed or broken, unhealthy, dead. Looking at surrounding plants may assist with this classification.

Plant health	**Features**
Healthy	Upright plant; healthy leaves, dense foliage
Broken or crushed	Leaves lying flat on the ground; broken stems
Unhealthy	Withered leaves; colour dull or yellowed; leaf tips dead
Dead/dying	All stems and leaves brown, withered or decaying

Compare the information for each quadrat.

6.5 Saltmarsh quadrat recording sheet



Group name:	**Date:**	**Time:**
Tide (tick): ☐ high ☐ low	☐ coming in (flow)	☐ going out (ebb)

Method of collecting data: (select either plotted or random):

☐ **Plotted transect/quadrats**

Transect length (metres): Quadrat spacing (metres):

☐ **Random quadrats within a set boundary:**

Area within boundary (metres square): Number of quadrats:

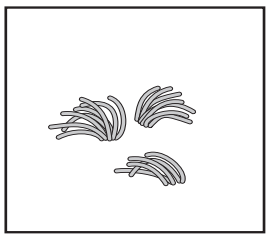
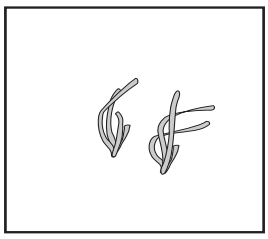
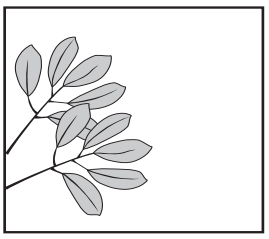
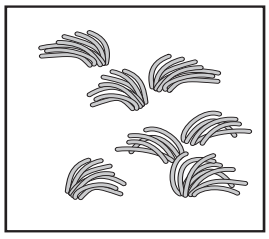
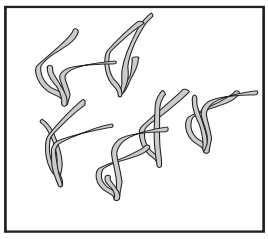
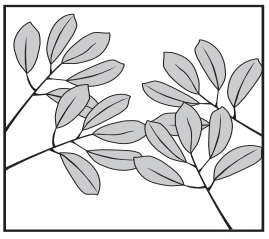
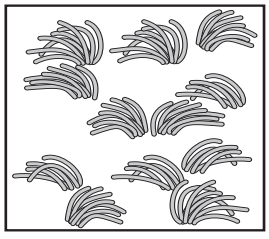
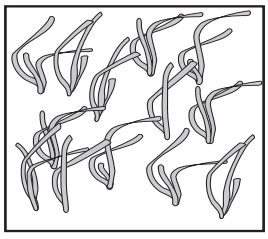
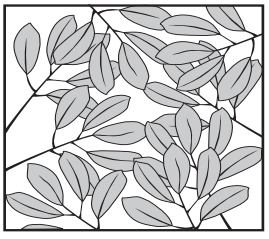
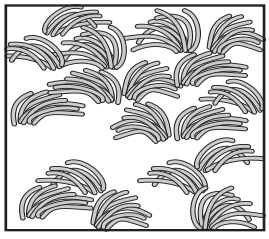
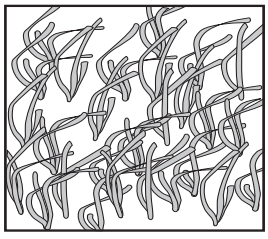
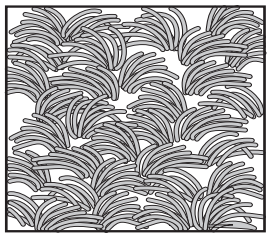
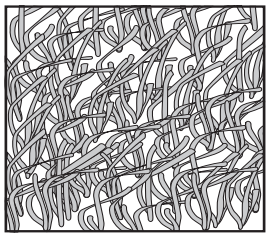
Shape of quadrat: ☐ Square quadrat ☐ round (hoop) quadrat

Quadrat number	**% Cover**	**Average plant height (cm)**	**Plant species within the quadrat**	**Species health (% of cover) Healthy, crushed or broken, unhealthy**
1				
2				
3				
4				
5				
6				
7				
TOTAL (per square metre)				

6.6 Estimating saltmarsh cover

Refer to the relevant diagram below to estimate the percentage cover of saltmarsh.

Estuary plant percentage cover diagrams

Saltmarsh	Seagrass	Mangroves
0 -5%	**0 -5%**	**0 -5%**
		
5 -25%	**5 -25%**	**5 -25%**
		
25 - 50%	**25 - 50%**	**25 - 50%**
		
50 -75%	**50 -75%**	**50 -75%**
		
75 -100%	**75 -100%**	**75 -100%**
		

SECTION 7

Seagrass monitoring

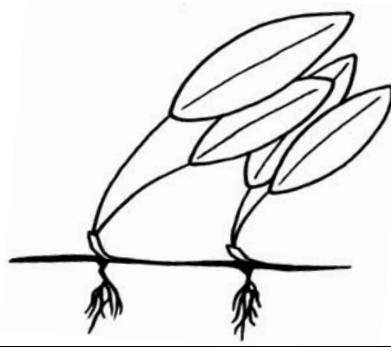
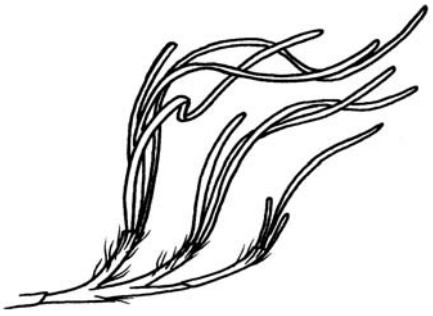
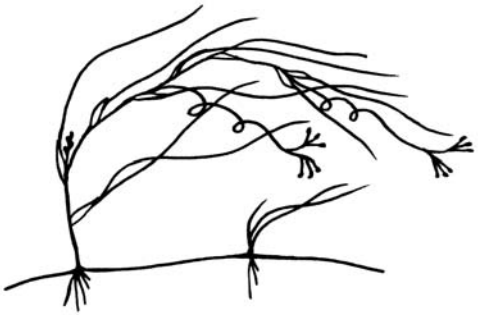
Seagrass is a flowering plant that lives in marine and brackish water. It is usually found in sheltered bays, lagoons, lakes and estuaries. It needs sunlight, good water quality and nutrients to grow. There is an abundance and diversity of species dependant on seagrass beds.

Monitoring seagrass is a procedure that requires specialised training. Waterwatch groups can monitor some aspects of seagrass health that do not involve this specialised training, as part of their Waterwatch Plan.

This section includes some background information about seagrass and a series of field activities suitable for Waterwatch groups to incorporate within their monitoring plans.

Included in this section: *Page*

7.1 Seagrass identification chart

NAME	Distinguishing Features	Appearance
Eelgrass *Zostera sp.*	• Leaf with 3-5 parallel veins • Cross veins form boxes • Leaf tip smooth and rounded, may be darker at the tip • Rhizome usually brown or yellow in younger parts • Leaf size can vary • Colour varies from bright green to brown • Leaves rise directly from rhizome	
Paddleweed *Halophila ovalis*	• Small seagrass with upright stems • Rounded leaves in pairs • Light green leaves (4 cm long; 2 cm wide) • Stems and leaves come off runners • Runners occur below sediment surface and also bear roots	
Strapweed *Posidonia sp.*	• Large strap like leaves - thick and stiff with rounded tips • Leaves are 45cm long; 15-20mm wide • 14-20 longitudinal veins • Leaves generally bright green • 3-5 leaves rising from leaf base at rhizomes	
Sea Tassel *Ruppia sp.*	• Long slender leaves (0.5-2mm wide) • Highly branched stem • Leaves are 50-200mm long • Generally dark green	

7.2 Field identification guide: common seagrass species

Common seagrass species

Eelgrass (*Zostera* species)

Description

- Leaf with 3–5 parallel veins
- Cross veins form boxes
- Leaf tip smooth and rounded, may be dark at the tip
- Rhizome usually brown or yellow in younger parts
- Leaf size can vary; colour can vary from bright green to brown
- Leaves always arise directly from rhizome

Photograph: Danny Roberts and Shane Murray, NSW DI&I, used with permission

Paddleweed (*Halophila ovalis*)

Description

- Small seagrass with upright stems
- Rounded leaves in pairs
- Leaves light green and grow up to 4 cm long and 2 cm wide
- Stems and leaves come off runners
- Runners occur below sediment surface and also bear roots

Photograph: David Harasti, NSW DI&I, used with permission

Strapweed (*Posidonia* species)

Description

- Large strap-like leaves which are thick and stiff with rounded tips
- Grows to 45cm, 15–20 mm wide with 14–20 longitudinal veins
- Leaves generally bright green in colour with ±3–5 leaves arising from the leaf base at the rhizomes

Photograph: Danny Roberts and Shane Murray, NSW DI&I, used with permission

Common seagrass species *continued*

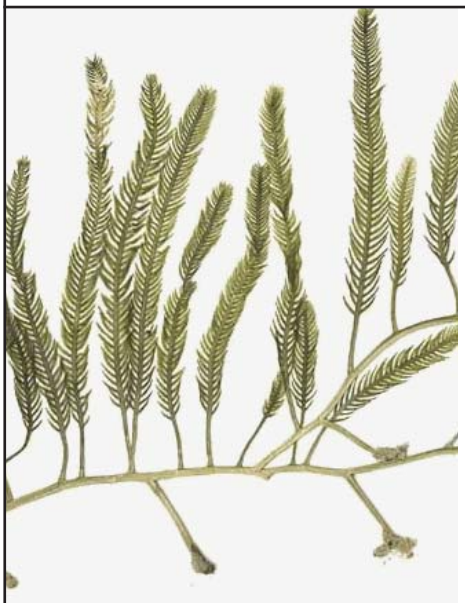
Sea tassel (*Ruppia* species)

Description

- Long, slender leaves
- Narrow (0.5–2 mm) highly branched stem
- Leaves vary from 50–200 mm long
- Leaves generally dark green in colour

Photograph: NSW DI&I, used with permission

Introduced species and pest *Caulerpa taxifolia*



Description

- Fronds would sit flat on table
- Fronds slightly narrowed at the base
- Fronds upwardly curved, tapering at tip
- Tips pointed, never spherical or club-shaped
- Frond length increases with depth
- The horizontal branches have an average diameter 1.5–1.8 mm
- Can be highly variable in appearance

Note: There are native Caulerpa species that are not a threat to ecosystems.

Photograph: Alan Millar, Royal Botanical Gardens, Sydney, used with permission.

Paddleweed, eelgrass and strapweed

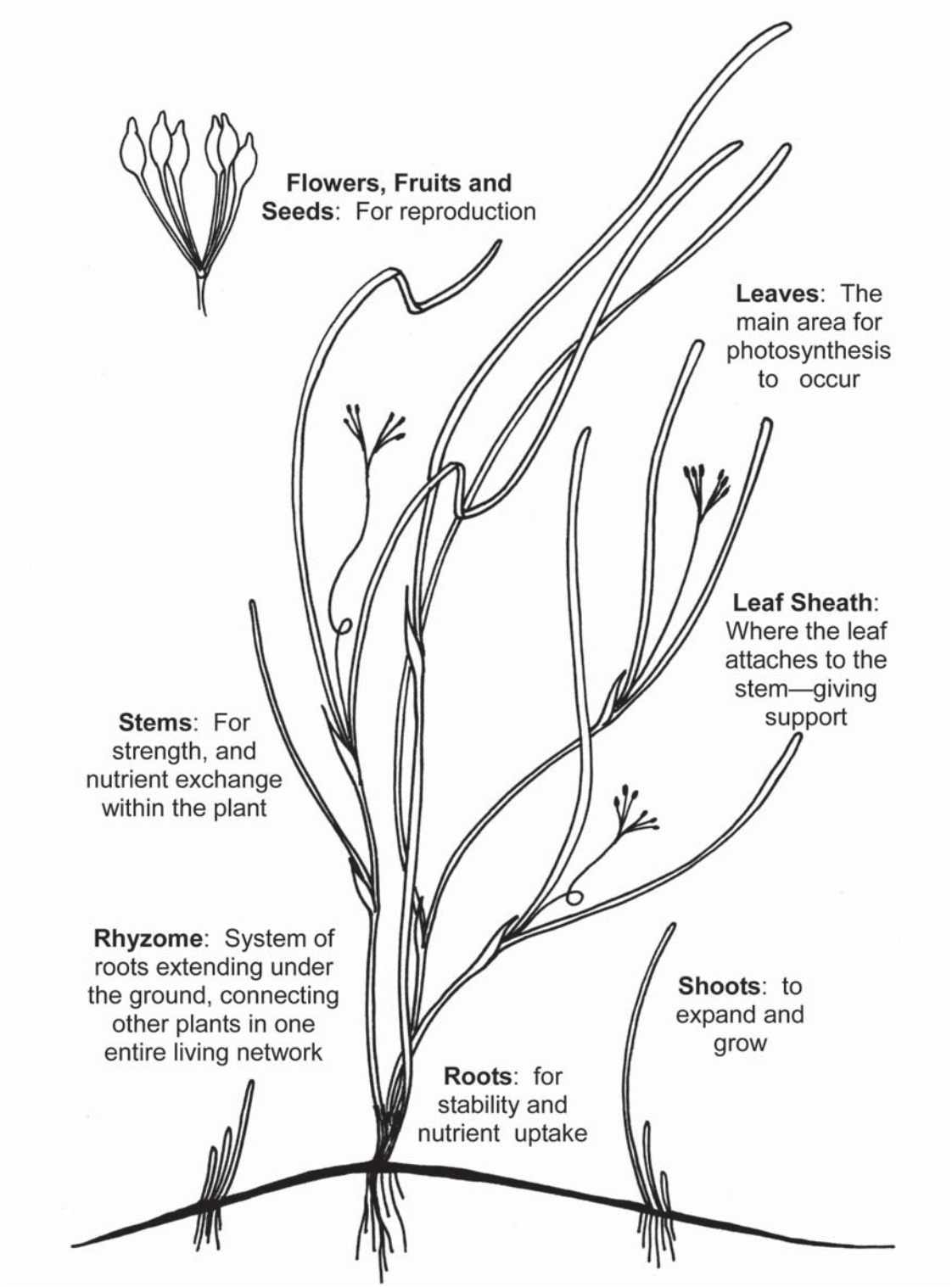
Photograph: Justin Gilligan, NSW DI&I, used with permission.

Anemone with strapweed and paddleweed

Photograph: Justin Gilligan, NSW DI&I, used with permission.

7.3 Anatomy of seagrass

Seagrasses, like land plants, are structurally complex, possessing the following structures:



7.4 Seagrass wrack investigation

Level of difficulty: Easy

Suitable for all ages.

Seagrass wrack is the leaves of seagrass which have been detached from the original plant. This is due to a biological response to season and increased hydrodynamic forces due to winter swell. Seagrass is naturally buoyant and floats to the surface and is pushed to the foreshore by wind and tidal currents. Seagrass wrack on the shore provides microhabitat, nutrients and shading for species, e.g. saltmarsh plants.

Groups will need initially to look through the seagrass wrack to identify species occurring in their local area.

Procedure

1. Look for seagrass wrack washed up on the foreshore. Refer to the photo below.
2. Walk the perimeter of the foreshore and randomly throw/place a quadrat or hula hoop onto the wrack.
3. Identify and count invertebrates within the wrack. See Section 8.7 of this manual for instructions and use the recording sheet in Section 8.16.
4. Note that seagrass wrack is often intermixed with macroalgae (seaweed) species.

Important note:
You must not remove any seagrass or wrack without a permit.

7.5 Measuring seagrass distribution: distance to the nearest edge

Level of difficulty: Easy

Suitable only for community groups due to the Occupational Health and Safety risks of students entering water.

The shallow edge of the seagrass bed is defined as the onshore edge of the continuous seagrass cover. The last shoot is the deepest occurring seagrass shoot.

Equipment: GPS unit, boat, kayak or pier, 100 metre measuring tape, bathyscope, seagrass identification sheet.

Step 1: Choose a fixed starting point (permanent structure) on shore or locate the starting point with a GPS unit or compass.

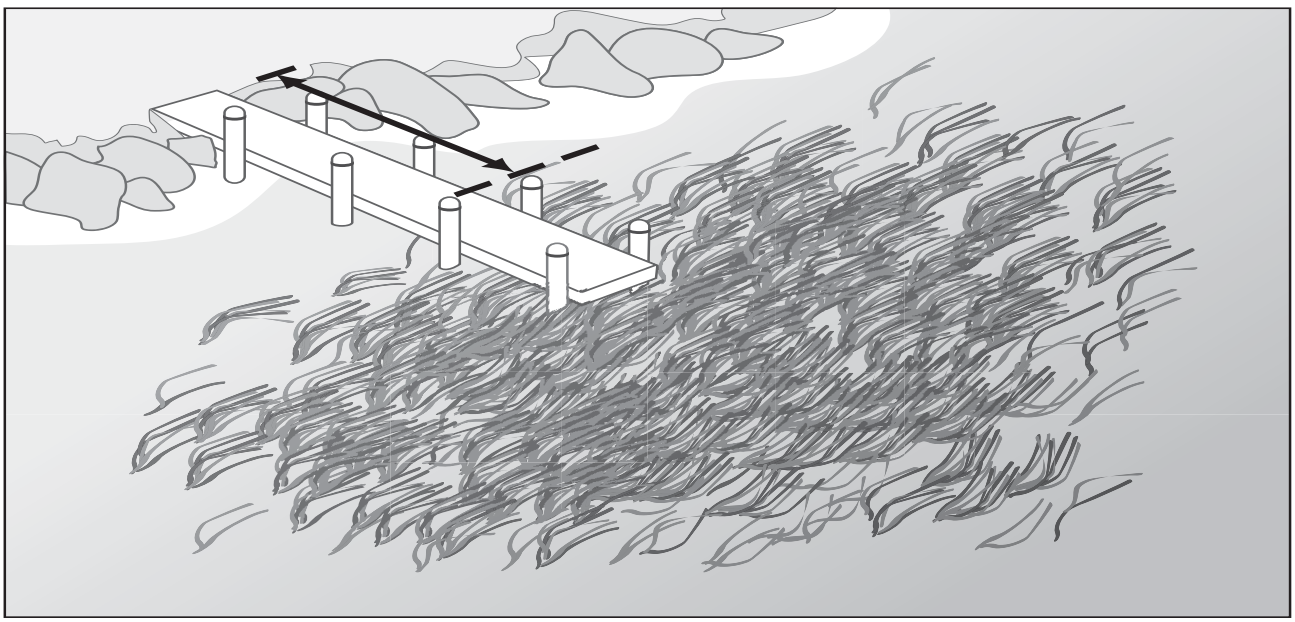
Step 2: Measure the distance to the shallowest seagrass bed in metres.

Step 3: Record this distance in metres on the recording sheet (see Section 7.8), to the nearest tenth of a metre.

If the edge of the seagrass has contracted inside the fixed starting point (since the first survey was taken), record the distance as negative.

Step 4: On the back of the recording sheet, provide a map and site description specifying where the inner and outer edges of the meadow are located.

Distance from Shoreline to nearest edge of seagress bed



7.6 Measuring seagrass continuous cover: water-based monitoring group

Level of difficulty: Moderate

Suitable for community groups.

Other: OHS issues.

Measure the continuous seagrass cover from the shallow to the deepest edge. This will require monitoring in a boat or along a jetty.

Equipment: GPS unit, boat, kayak or pier, 100 m measuring tape, secchi disk, bathyscope (underwater viewer), seagrass identification sheets.

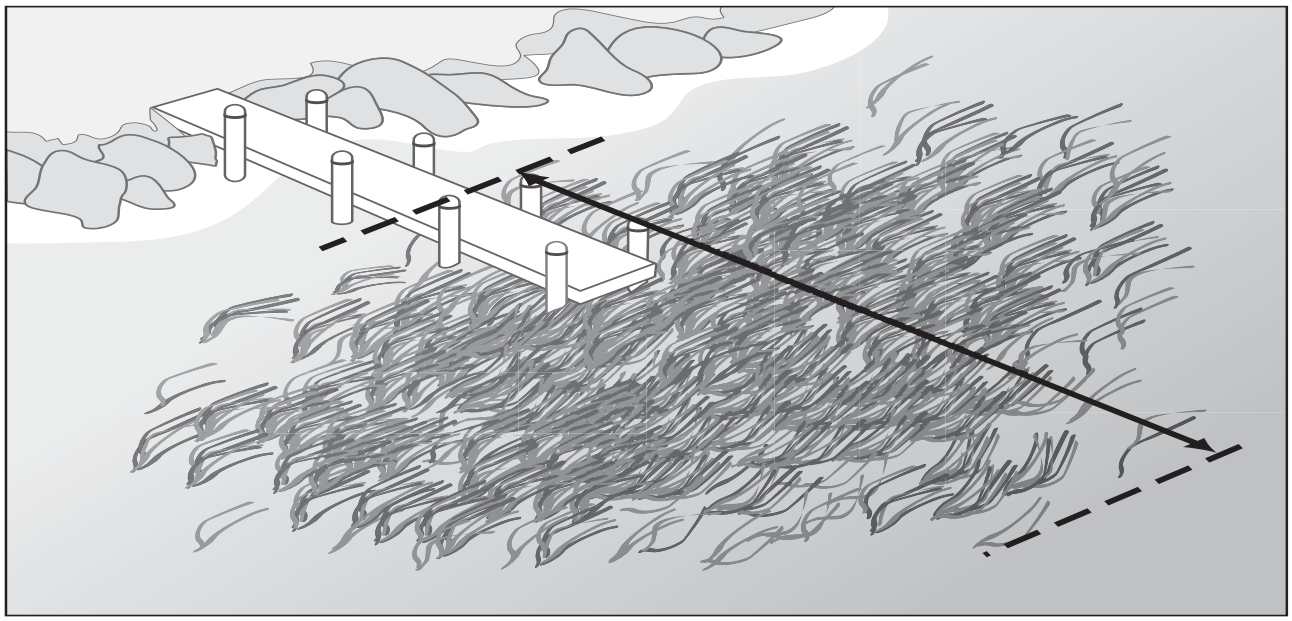
Procedure

Note: Before beginning this activity, complete the field activity in Section 7.45 to measure the seagrass distance from a fixed point. Enter this distance on the recording sheet in Section 7.8.

Step 1: Using a 100 m tape, measure the distance to the limit of seagrass from the shallow edge to the deep edge.

Record the distance of continuous seagrass cover.

Distance from nearest edge to furthest edge of seagrass bed.



Step 2: Using a bathyscope, identify the seagrass species.

> **Tip: If unsure of the seagrass species take photos and use a seagrass identification guide.**

Step 3: Estimate the percentage cover of seagrass in the area using the relevant estuary plant percentage cover diagram – in Section 6.4.

> **Note: Seasonal measurements can be recorded and compared over time.**

7.7 Seagrass perimeter measurements: water-based monitoring group

Level of difficulty: Moderate

Suitable for community groups.

Other: OHS issues.

Prepare a sketch map of the seagrass distribution within a defined area by mapping the boundaries using GPS coordinates.

This information can provide a basis from which loss or gain in seagrass habitat is monitored. If it is conducted during low tide when the seagrass is exposed you can wade or walk the perimeter. Other options include swimming, snorkelling, kayaking or boating.

Choose the appropriate method for your site and the conditions to establish the perimeter of the seagrass bed.

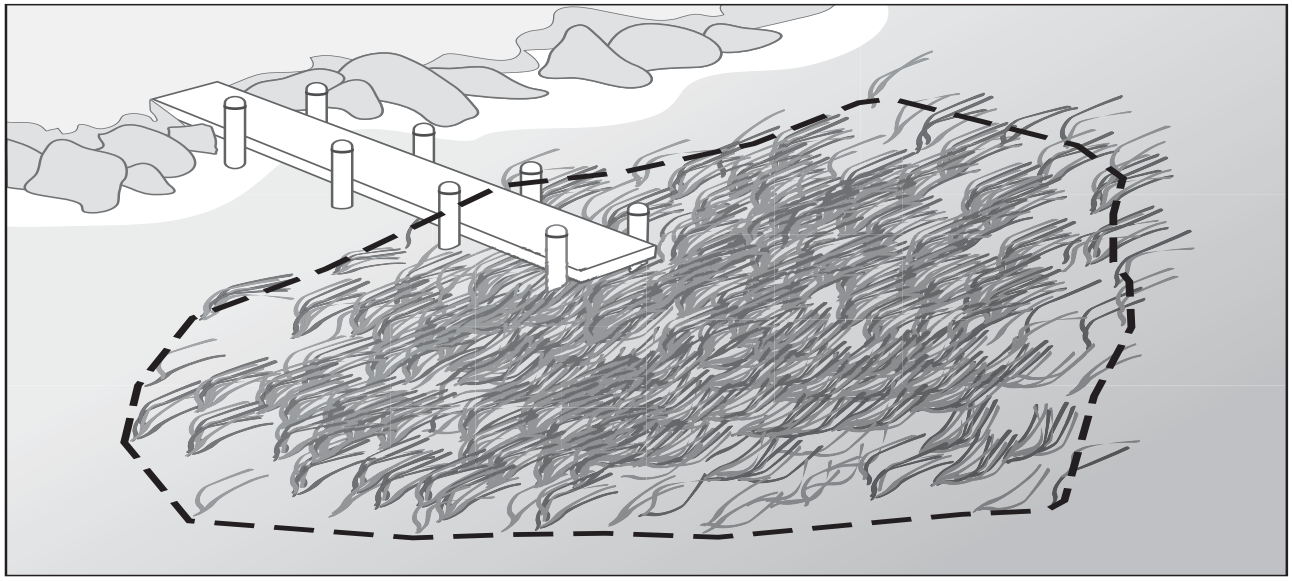
Note: Do not walk through the seagrass as this is a very fragile environment.

Equipment: GPS unit, boat, kayak or jetty, recording sheets, seagrass identification guide and seagrass percentage cover guide.

Procedure

Step 1: From the nearest edge of the seagrass bed take your first GPS reading and then every 5–25 metres (depending on size of the area and time available).

Distance around circumference of seagrass bed.

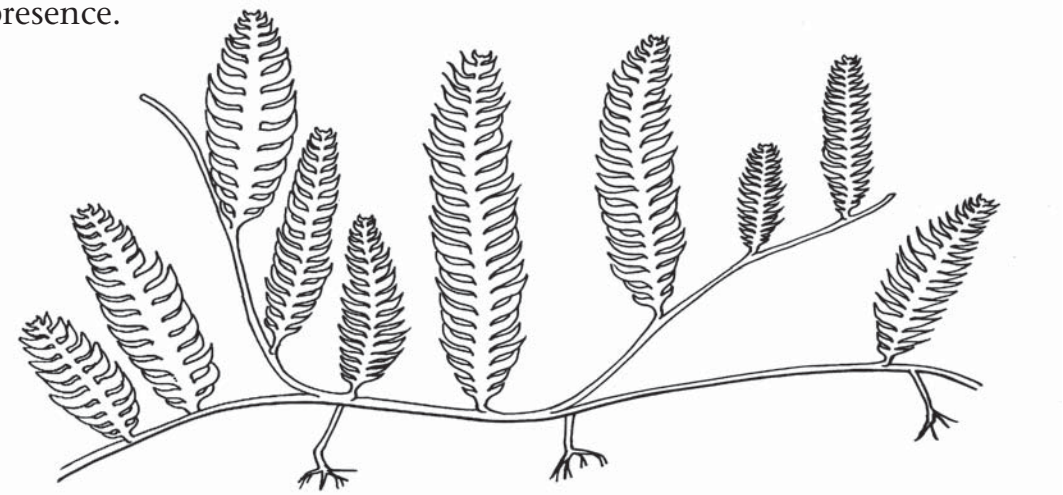


Step 2: Draw a sketch map of the area you are mapping and include landmarks to assist to identify the mapped area in the future.

Step 3: Comment on the shape of the continuous seagrass bed.

Step 4: Estimate the seagrass percentage cover by referring to the seagrass cover guide.

Step 5: Mark the location of Caulerpa taxifolia on the sketch map if you identify its presence.



Sketch map of seagrass area

7.8 Seagrass recording sheet

Group name:	**Date:**	**Time:**
Tide (tick): ☐ high ☐ low	☐ coming in (flow)	☐ going out (ebb)
Location land marker:	OR	
GPS Reading:	Latitude	Longitude
Method of collecting data: (tick the box below): ☐ land ☐ boat ☐ jetty ☐ other (e.g. snorkelling, wading)		

Distance from land marker to shallow seagrass edge (metres)	Distance from shallow to deep edge	Percentage cover of seagrass	Dominant seagrass species	Shape of seagrass bed

Perimeter location measurements

GPS location	Latitude	Longitude
1 Shallow edge		
2		
3		
4		
5		
6		

Length of perimeter:

Calculate the area of seagrass: (A=L x B)

Caulerpa taxifolia

If *Caulerpa* is identified while conducting seagrass monitoring, note its location as accurately as possible on the data sheet and notify local fisheries officers. Do not remove Caulerpa and take care not to disturb it.

Tick: ☐ present ☐ absent

If present, take a GPS reading: Latitude: Longitude:

Mark the distribution on the sketch map of the seagrass bed.

SECTION 8

Investigating salt water invertebrates

Estuaries are home to many small animals without a backbone, called invertebrates. They include crustaceans, molluscs and worms.

The presence and abundance of species adapted to different saltwater habitats provides an indication of the health of the ecosystem.

This section provides activities for investigating salt water macroinvertebrates.

Included in this section: *Page*

8.1 Measuring the abundance of organisms

Abundance is the number of organisms represented in a particular ecosystem.

The abundance of each type of organism is recorded on the Waterwatch data sheet for each sample, using the following rating:

1 = uncommon (<5 individuals)

2 = common (5–20 individuals)

3 = abundant (>20 individuals)

Interpretation

There has not yet been enough research into estuary macroinvertebrates to provide values to determine sensitive and tolerant species. All sampling should be related to the specific site and will provide an indication of change over time that may be caused by natural or human changes.

Each invertebrate is either present or absent at a site for a combination of reasons. Take into account lifecycle, seasons, water flow, riparian vegetation, habitats, etc. to try to explain why each animal is living at a site and how it is interacting with the environment and other organisms.

Some invertebrates can be classed as either sensitive or tolerant based on their response to specific changes in the water conditions. You can compare your data from a site at different times of the year, or with other sites, or with other water bodies to provide an indication of these sensitivities.

8.2 Common invertebrates in estuaries

Arthropods
Animals with hardened skin and jointed appendages

Crustaceans
Animals with crust-like skin

crabs & crayfish

prawns/shrimp

barnacles

isopods & amphipods

Molluscs

gastropods (snails)

bivalves
(clams, mussels & oysters)

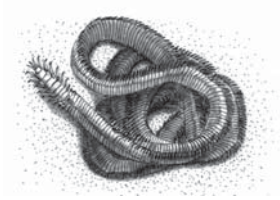
nudibranchs (e.g. sea slugs)

cephalopods
(octopus, squid & cuttlefish)

Worms

Segmented worms

Annelida



beach worms/rag worms

tube worms

Unsegmented worms

Platyheminthes

flatworms

Nemeteans

ribbon worms

8.3 Salt water bug sampling: core sampling

Level of difficulty: Easy

Suitable for all ages.

Salt water bugs can provide an indication of the health of the estuary. This is a way to find out what lives in the sediment. A sample is taken from the sediment to a known depth. A number of core samples can be taken in different parts of the mudflats and the results compared.

Equipment: Spade, core sampler or pipi pump, sieve (mesh 0.5 mm), white sorting tray, pipettes, ice cube trays, bucket, magnifying glass, recording sheet, and estuary detective guide.

Locating the sampling site

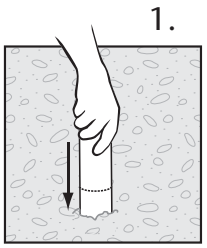
Random core sampling: Select an area close to the bank at low tide. As a group, dig a hole in the sediment to expose the different coloured layers. Note the colours of the top and bottom sediments (fawn or light coloured layers indicate the presence of oxygen; brown or grey layers usually do not have oxygen).

Sampling along a transect: Select core sampling sites along a transect line. The aim of the investigation determines whether the transect should be parallel or vertical to the edge of the waterway.

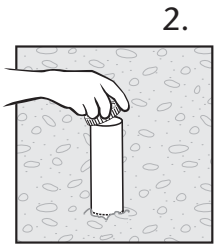
Procedure

1. Working in groups, select sampling sites.
2. Insert the corer into the sediment without the lid on.
3. When the corer reaches the desired depth, put the lid on.
4. Remove the core sample from the sediment.
5. Sieve the core sample over a bucket using water from the estuary.
6. Place the contents of the sieve into the sorting tray.
7. Sort the water bugs into groups and place in different compartments in the ice block tray.
8. Use ID sheets to identify the catch.
9. Return all sediment and bugs to the sampling site.
10. Rinse all equipment, making sure that all organic matter is removed from the sampler.
11. Repeat the sample at a number of locations.

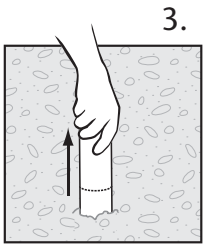
Locating the sampling site



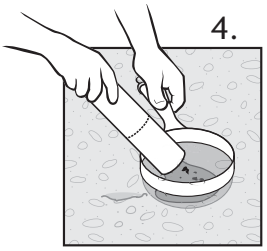
Push the tube into the soil



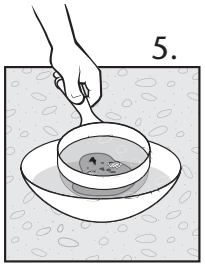
Place on the cap



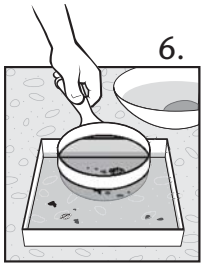
Pull out the tube



Tip sample into a sieve



Rinse away the soil



Place the organic matter that is left in an assessment tray with water

Making a core sampler

A simple corer for sampling can easily be constructed from a length of 50 mm diameter PVC pipe (at least 50 cm long) and a PVC cap.

Cut the PVC pipe to a length of approximately 50 cm using a hacksaw.

Clean the cut edges with a light sandpaper. At 20 cm from one end of the PVC pipe (or the depth that you intend to insert the corer), make a mark on the outside using a hacksaw. Continue the mark around the entire pipe. The corer will be inserted to this line.

8.4 Core sampling recording sheet

The estuary bug recording sheet provided in Section 8.16 should be used to record species types. The recording sheet below can be used to record **total numbers of invertebrates.**

Group Name:

Date: **Time:**

Tide *(tick)*: ☐ high ☐ low ☐ coming in (flow) ☐ going out (ebb)

Method of collecting data: *(select either plotted or random core samples)*

☐ Plotted transect
Transect length (metres):_____ Number of core samples: _____

☐ Random core samples within a set boundary:
Area within boundary
(metres square): _____ Number of core samples: _____

Core sample	Number of species types	Total number of individuals	Species abundance rating (number of individuals per type)		
			Abundant (>20)	Common (5-20)	Uncommon (<5)
1					
2					
3					
4					
5					
6					
7					

Abundant species types:

Common species types:

Uncommon species types:

8.5 Species abundance: counting surface snails

Level of difficulty: Easy

Suitable for all ages.

Surface snails can be used to assess habitats in mud or sand flats due to their ease of detection.

Surface snails provide food for a wide variety of larger species such as crabs, birds and fish, particularly wading birds that inhabit tidal flats.

Snails are affected by a number of natural changes within estuaries:

- changes in water flow
- increased nutrients
- increased sedimentation
- sulphide rich mud build-up
- changes to water temperature or salinity
- stormwater discharges
- predatory animals or introduced marine pests.

Frequency of monitoring

Seasonal fluctuations – monthly or quarterly monitoring

Long-term changes – 6 monthly or yearly monitoring

Selecting a site

Select a site which is typical of the whole area. This is called a representative site.

Procedure

Step 1: Follow the procedure for a plotted transect or random transects within a set boundary in Section 4.3 of this manual.

Step 2: On the recording sheet, enter the following dat

- the number of snails inside each quadrat
- the number of types of snails
- a description of each snail type.

8.6 Quadrat study: snail recording sheet

Group Name:

Date: **Time:**

Tide *(tick)*: ☐ high ☐ low ☐ coming in (flow) ☐ going out (ebb)

Method of collecting data: *(select either plotted or random)*

☐ Plotted transect/quadrats:
Transect length (metres):_____ Quadrat spacing (metres): _____

☐ Random quadrats within a set boundary:

Area within boundary
(metres square): _____ Number of quadrats:_____

Shape of quadrat: ☐ square quadrat ☐ round (hoop) quadrat

Quadrat	Snail count	Species types (whelk, snail, air breather)	Description (colours and shapes)
1			
2			
3			
4			
5			
6			
7			
8			
TOTALS			
Number of species/square metre:			
Number of snails/square metre:			

Calculations: **Refer to Section 4.3 for information related to recording results as a number per square metre.**

8.7 Monitoring salt water bugs in seagrass wrack

Level of difficulty: Easy

Suitable for all ages.

Some seagrass species (Zostera) shed some of their leaves annually, just like trees. Seagrass wrack refers to the shed or detached leaf material of seagrass plants.

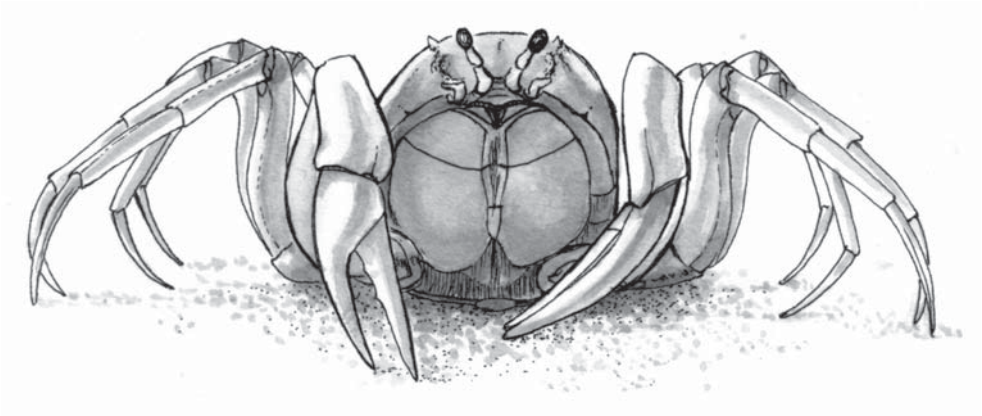
Equipment: Sieve, white sorting tray (or basin or bucket), white ice cube tray or specimen containers, magnifying glass, tongs, plastic spoon, thick gloves, pen, clipboard, estuary recording sheet, and estuary detective guide.

Procedure

1. Choose an area of foreshore with plenty of seagrass wrack that has not been walked on too much. The thick mats of fresh seagrass wrack at the high tide mark are best.
2. Put some clean water from the estuary into the white sorting tray/basin/bucket. Fill to about 2 cm deep.
3. Put on gloves. Place the sieve over the sorting tray/basin/bucket with water in it and use the tongs to pick up 3 or 4 clumps of the seagrass wrack. Try to get the damp wrack under the surface layer. Place the wrack in the sieve.
4. Shake the sieve several times so that any animals in the sieve fall through the holes into the sorting tray/basin/bucket. You can stir the wrack with your tongs.
5. Identify seagrass types found in your sieve (using local seagrass identification guides). Check for any large animals and shells left in the sieve. Make note of any other material in your sieve (e.g. rubbish).
6. Remove any floating plant material from the water in the sorting tray/basin/bucket with your fingers (so animals can be more easily seen).
7. Put some clean water from the estuary into the ice cube tray or specimen containers.
8. Let the water become calm in the sorting tray/basin/bucket and look for animals crawling on the bottom as well as things swimming around. Use the spoon to lift the bugs from the sorting tray/basin/bucket to the ice cube tray or specimen containers for closer inspection.

Procedure

9. Have a look at the bugs through the magnifying glass.
10. Compare your bugs with the pictures in the Estuary Bug Detective Guide.
11. Sort the bugs into groups (e.g. put all the sand hoppers together) and place in separate compartments in the ice cube tray or in specimen containers (a separate container for each bug type). Note: a specimen container would be better as sand hoppers can jump quite high and can easily jump out of ice cube trays!
12. Record the number and diversity of species on the Waterwatch estuary bug recording sheet (Section 8.16). You should only count up to 20 bugs of the one type as everything over this has a constant weighting factor.
13. Draw conclusions about the health of the site based on your results.
14. High diversity and species numbers may indicate a healthy site.
15. Low numbers and few species may indicate problems with the ecosystem.
16. Return bugs to the sampling site (gently).
17. Rinse all equipment, making sure that all bugs, organic matter and sediment is removed.
18. Repeat the sample at a number of locations.



8.8 Field identification guide: common crab species

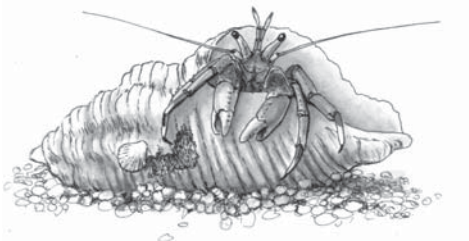
Large crabs

Swimming crab, *Portunus pelagicus*
Description: Their last pair of legs are modified as swimming paddles
Maximum size: Up to 350 mm
Habitat: Soft sediment and mud

Mud crab, *Scylla serrata*
Description: Dark brown or dark green mottled crab
Size: Large crab – can grow to more than 240 mm
Habitat: Mud, mangroves

Medium-sized crabs

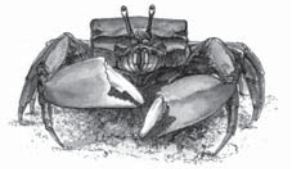
Hermit crab, *Pagurus sinuatus*
Description: Crab that uses the shell of another creature for its home
Size: 80–150 mm
Habitat: Sand or mudflats and mangroves



Spotted shore crab,
Paragrapsus gaimardii (Grapsidae)
Description: Very large purple-brown claws and grey or yellow flecked carapace
Maximum size: Up to 30–35 mm
Habitat: Mud; mangroves

Smooth handed crab,
Pilumnopeus serratifrons
Description: Purple colour and hairy legs
Size: 25–30 mm
Habitat: Mud; mangroves

Semaphore crab,
Heloecius cordiformis
Description: Rectangular shaped crab, greenish grey with light coloured limbs
Maximum size: 30–40 mm wide
Habitat: Different species in mud, shallow pools, mangroves, seagrass



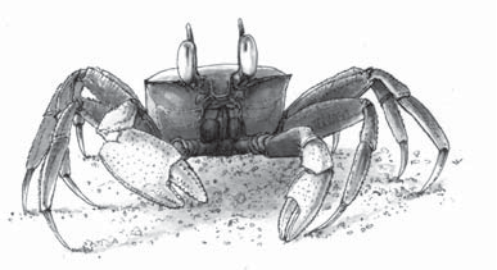
Medium-sized crabs

Ghost crab, *Ocypode cordimana*

Description: Light grey in colour, rounded shell, large eyes at the end of short eyestalks

Maximum size: Up to 35–50 mm

Habitat: Dry sand near estuary entrance or sandy estuarine beaches



Sentinel crab, *Macrophthalmus setosus*

Description: Rectangular shaped crab, greenish grey with light coloured limbs

Maximum size: 30–40 mm wide

Habitat: Different species in mud, shallow pools, mangroves, seagrass

Smooth shore crab, *Cyclograpsus granulosus*

Description: Purple, dark grey or purplish grey, scavenger eats whatever is around

Size: 30–40 mm

Habitat: Sheltered bays and estuaries in mud areas and saltmarsh flats

Red fingered marsh crab, *Sesarma erythrodactyla*

Description: Green carapace and red-tipped claws

Maximum size: 20–30 mm across with brown hairs on legs

Habitat: Mangroves, saltmarsh and river banks

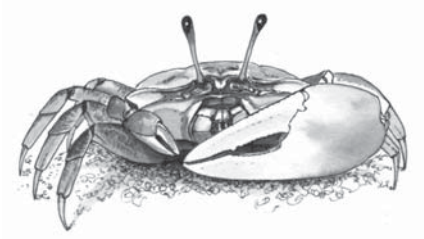
Small crabs

Fiddler crab, *Uca (Decapoda)*

Description: Has one large claw

Maximum size: 20–30 mm

Habitat: Mudflats, mangroves



Soldier crab, *Mictyris longicarpus*

Description: Burrows into sand in corkscrew manner if frightened

Maximum size: 12–15 mm

Habitat: Estuaries sand/mud

8.9 Crab species recording sheet

Level of difficulty: Medium

Suitable for secondary students

What species of crabs did you see?

If you don't know the species describe each one in the table below and conduct further research to identify them:

Group Name:

Date: **Time:**

Tide *(tick)*: ☐ high ☐ low ☐ coming in (flow) ☐ going out (ebb)

Method of collecting data: *(select either plotted or random)*

☐ Plotted transect/quadrats:
Transect length (metres):_____ Quadrat spacing (metres): _____

☐ Random quadrats within a set boundary:

Area within boundary
(metres square): _____ Number of quadrats:_____

Shape of quadrat: ☐ square quadrat ☐ round (hoop) quadrat

Specimen	Species (if known)	Small or large	Colour	Shape	Special features
Crab 1					
Crab 2					
Crab 3					
Crab 4					

Assessing your results

Changes in crab numbers can indicate pollution at the site. Crabs accumulate toxins that can have an impact on their survival. Some crabs can cope with more toxins than others.

Number: _____

Diversity (number of different types): _____

Introduced species

Green shore crabs are an introduced species that are becoming an increasing problem in estuaries. Did you see any green shore crabs?

☐ Yes ☐ No

Take care: One hole does not always indicate one crab.

8.10 Abundance and diversity: mudflat crab hole count

Level of difficulty: Easy

Suitable for all ages.

Although crabs are quite mobile, they do not stray far from their burrows. They are thought to accumulate toxins, indicating pollution in the environment and a decline in environmental health conditions if their numbers decline.

Procedure

Step 1: Conduct a quadrat survey using the procedures described in Section 4.3 for:

- fixed transects
- random quadrats in a defined area.

Step 2: Record where each count is taken using a simple map.

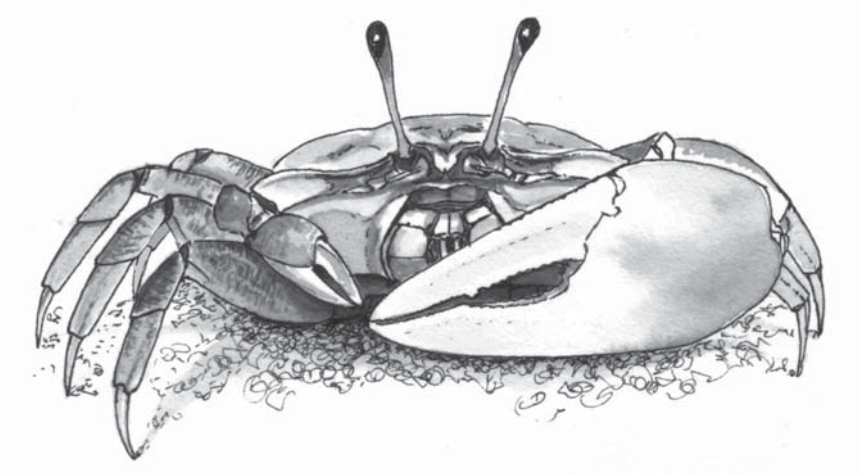
Step 3: Record the number of crab holes on the recording sheet. If there are too many to count in each quadrat, divide your quadrat into 4 and multiply your answer by 4.

Step 4: Measure the difference in the crab holes as this may indicate different species.

Step 5: Make observations of any crabs you see and identify them.

Step 6: Discuss what you have found out about crabs at this location.

Note: Refer to Section 4.3 for procedures for setting up a quadrat.



8.11 Mudflat crab hole count recording sheet

Location of each quadrat

Draw a map showing the location of your quadrat in relation to other features at the site:

Crab hole numbers recording sheet

Group Name:

Date: **Time:**

Tide *(tick)*: ☐ high ☐ low ☐ coming in (flow) ☐ going out (ebb)

Method of collecting data: *(select either plotted or random)*

☐ Plotted transect/quadrats:
Transect length (metres):_____ Quadrat spacing (metres): _____

☐ Random quadrats within a set boundary:

Area within boundary
(metres square): _____ Number of quadrats:_____

Shape of quadrat: ☐ square quadrat ☐ round (hoop) quadrat

	Quadrat 1	Quadrat 2	Quadrat 3	Quadrat 4	Quadrat 5	TOTAL
Number >1cm						
Number <1cm						
TOTAL crab holes						
Number of crabs seen						

Notes:

8.12 Fact sheet: green shore crab (Carcinus maenas)

This crab has been introduced to Australia and poses an extreme threat to coastal ecosystems.

It is a voracious predator with a broad diet and has been implicated in the decline of native shellfish populations. It consumes a wide variety of native species, outcompeting most for food and habitat. In Tasmania, Carcinus maenas has been present for about 15 years and is a major cause of mortality in native crab and mollusc populations.

Description: A medium-sized crab, it has five distinct spines on the outside edge of the carapace on either side of the eyes. The colour of the upper surface in adults is distinctively green but tends to red-orange on the under surfaces of larger animals. Juvenile crabs are generally lighter in colour than adults. The ends of the fourth walking legs are slightly flattened with acute tips

Size: Adults are usually 60 mm across.

Diet: Eats anything including oysters, mussels, marine worms, small crustaceans and shellfish.

Habitat: Extremely tolerant and hardy species, showing few limitations in the type of habitat it prefers. It is found in both the intertidal and shallow subtidal zones of bays and estuaries rather than exposed, rocky or sandy open coasts.

What do you do if you see a green shore crab?

According to the NSW Department of Industry and Investment (I&I), you should 'learn to recognise, and be aware of, the European green crab so that you can report any suspected new sighting to I&I NSW'.

Report any suspected new sightings to I&I NSW on the 24 hour recorded hotline: 02 4916 3877, or email aquatic.pests@industry.nsw.gov.au

If you are unsure of the species please take a photo or collect a sample (freeze crab in plastic bag) and send to I&I NSW Aquatic Biosecurity Unit, including a description of where it was found and GPS points or a map if possible.

If you see this pest in New South Wales, please report it immediately:

- noting the exact location
- if possible, taking a photo and/or collecting a sample and freezing it in a plastic bag.

Source: Department of Industry & Investment – www.dpi.nsw.gov.au/fisheries/pests-diseases/marine-pests/species/european-shore-crab

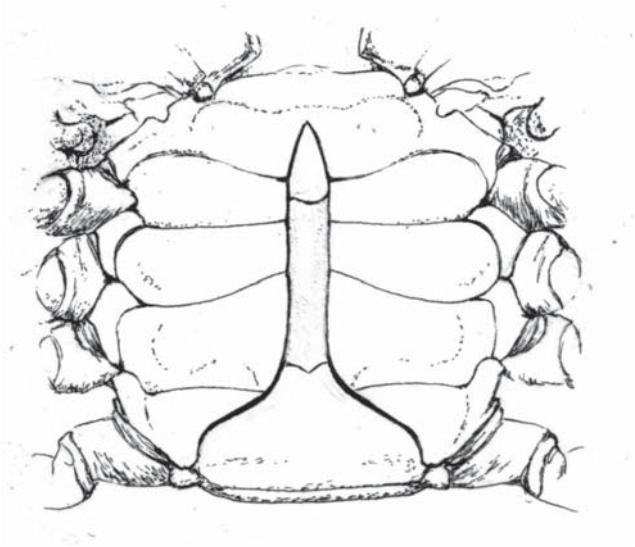
8.13 Observing crabs

Male and female crabs

Crabs have an abdomen which is folded up tightly underneath their body to form an abdominal flap.

When you turn a crab over and look underneath, you can tell a crab's sex by looking at its abdominal flap. A male crab has a small triangular flap, while a female crab has a broad oval-shaped abdominal flap.

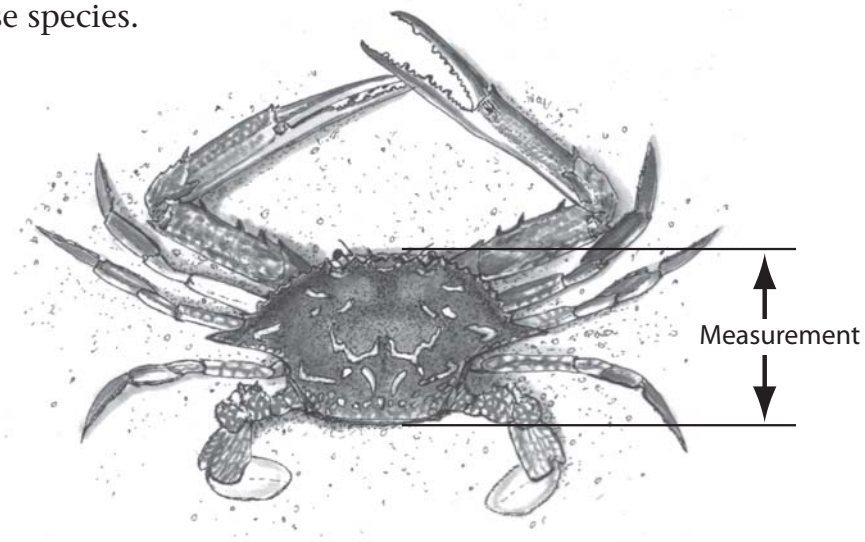
Female crab



Male crab

Measuring crabs

Crabs are measured along the body of the crab from the longest points. This is important to determine NSW Department of Industry & Investment bag limits for catching these species.



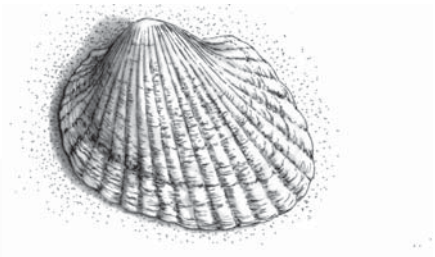
8.14 Abundance and diversity: shell collecting

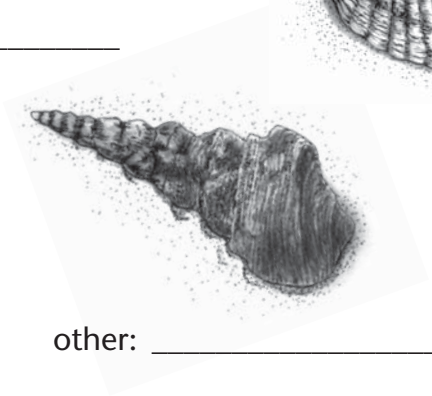
Level of difficulty: Easy

Suitable for primary students.

Many salt water bugs have shells. Shells are like fingerprints – they provide important clues about where they live and how they survive.

Look for shells of different shapes and sizes.





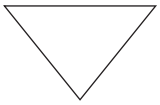
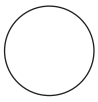
Select a favourite shell

What colour is my shell? _____

How big is my shell?

☐ big ☐ small ☐ tiny

What shape is my shell?


other: _____

Is my shell rough or smooth?

☐ rough ☐ smooth

Where did I find my shell? _____

Can my shell move around or is it fixed to something?_____

This is a drawing of my shell:

8.15 Abundance and diversity: salt water bug survey

Level of difficulty: Easy

Suitable for all ages.

Water bug surveys can be conducted in the seagrass, saltmarsh pools and freshwater rivers.

You will require a permit from the Department of Industry and Investment to net in these areas.

Equipment: Dip nets, sorting tray, ice block trays, pipettes, plastic spoons, magnifying chart and ID sheet.

Procedure

1. Assess the salinity at the site by using your EC (high range) meter and the field procedure for testing salinity. Salinity will influence the dominance of fresh and saltwater species.
2. Assess the impact of salinity and tides on the site: in brackish water, salt tolerant freshwater species may exist with saltwater species. The most salt tolerant freshwater species are crustaceans: shrimp, crayfish, etc.
3. Conduct your water bug survey using the appropriate ID and recording sheets.
4. Record the number and diversity of species.
5. Assess your results and draw conclusions about the health of the site: 0.
    - High diversity and species number indicates a healthy site.
    - Low numbers and few species indicates problems with the ecosystem.

Saltwater survey methods

A water bug survey helps students to investigate water bugs that live in their local creek or river and the features and needs of these organisms.

Sampling methods for netting salt water bugs are the same as for freshwater. The procedure is included in both the *Waterwatch Field Manual* and the *Junior Waterwatch Field Manual*.

Take care when sampling in seagrass as it is a fragile environment – sample at the edge of the seagrass. Move your net through the seagrass in the same direction as the seagrass is lying.

For more information check the website: www.waterwatch.nsw.gov.au

8.16 Estuary bug recording sheet

Group Name:

Date: **Time:**

Location of sample:

☐ mudflats ☐ seagrass ☐ saltmarshes

☐ mangrove ☐ other: _____

Note: This is a general recording sheet for all estuary habitats. Some of these species will **not** be found in all habitats. Record what you catch.

Abundance rating: Total number of invertebrate types	
Abundant (A)	More than 20
Common (C)	5–20
Uncommon (U)	Fewer than 5

Invertebrate groups	**Invertebrate Types**	**Number of individuals collected**	**Abundance**		**Diversity**
			Total number of individuals collected	**Abundance rating: A >20 C 5–20 U <5**	**Number of invertebrate types**
Molluscs	**Gastropods (snails)**				
	Whelks				
	Snails				
	Jelly egg sac				
	Bivalves				
	Cockles				
	Mussels				
	Oysters				
	Razor clams				
	Pipis				
	Others				
	Nudibranchs				
	Sea slugs				
	Cephalopods				
	Cuttle fish				
	Squid				
	Octopus				
TOTAL for page 1 (add all numbers in each column)					

Invertebrate groups	Invertebrate Types	Number of individuals collected	Abundance		Diversity
			Total number of individuals collected	Abundance rating: A >20 C 5–20 U <5	Number of invertebrate types
TOTAL from Page 1					
Crustaceans	**Shrimp**				
	Shrimp				
	Prawns				
	Barnacles				
	6/4 plated barnacles				
	Isopods				
	Marine slater				
	Pill bug				
	Sand hopper				
	Skeleton shrimp				
	Crabs				
Decreasing size ↓	Swimming				
	Mud				
	Hermit				
	Ghost				
	Smooth shore				
	Spotted smoth shore				
	Sentinel				
	Smooth handed				
	Semaphore				
	Fiddler				
	Red fingered marsh				
	Soldier				
	Other				
TOTAL for page 2 (add all numbers in each column, including totals from page 1)					

Invertebrate groups	Invertebrate Types	Number of individuals collected	Abundance		Diversity
			Total number of individuals collected	Abundance rating: A >20 C 5–20 U <5	Number of invertebrate types
TOTAL from Page 2					
Worms	**Worms**				
Annelida (segmented worms)					
Polychaetes	Rag worms				
	Tube worms				
Unsegmented Worms					
Platyhelminthes	Flat worms				
Nemerteans	Ribbon worms				
TOTAL (add all numbers in each column, including totals from page 1 and 2)					

8.17 Fish species recording sheet

Note: These are not invertebrates, but their numbers and diversity may provide an indication of changes within estuaries.

Fish	Fish Type	Number of individuals collected	Abundance		Diversity
			Total number of individuals collected	Abundance rating: A >20 C 5–20 U <5	Number of fish types
	Pipe fish				
	Seahorses/sea dragons				
	Whiting				
	Mullet				
	Puffer fish (toad fish)				
	Fortescue				
	Leatherjacket				
	Flathead				
	Gudgeons				
	Goby				
	Other				
TOTAL (add all numbers in each column)					

Image by: John Lugg, DECCW

www.waterwatch.nsw.gov.au