

UNIVERSITY of TASMANIA



Institute for Marine and Antarctic Studies

Guide to the Environmental Monitoring and Assessment for Finfish Aquaculture

Jeff Ross, Flora Bush, Camille White, Jimmy Hortle,
Adam Davey, Jason Beard, Catriona MacLeod

May 2025



FRDC

Fisheries Research and
Development Corporation



Institute for Marine and Antarctic Studies,
University of Tasmania,
Private Bag 49, Hobart TAS 7001

This publication (and any information sourced from it) should be attributed to Ross J., Bush F., White C., Hortle J., Davey A., Beard J., and MacLeod C (2025). Guide to the Environmental Monitoring and Assessment for Finfish Aquaculture. Institute for Marine and Antarctic Studies, Hobart, Tasmania

This Field Guide was developed under FRDC project 2015-024

Enquires should be directed to:

Dr Jeff Ross
Institute for Marine and Antarctic Studies,
University of Tasmania,
Private Bag 49, Hobart, Tasmania, 7001, Australia

jeff.ross@utas.edu.au

Ph: (03) 6226 8281

The authors do not warrant that the information in this document is free from errors or omissions. The authors do not accept any form of liability, be it contractual, tortious, or otherwise, for the contents of this document or for any consequences arising from its use or any reliance placed upon it. The information, opinions and advice contained in this document may not relate, or be relevant, to a reader's particular circumstance. Opinions expressed by the authors are the individual opinions expressed by those persons and are not necessarily those of the Institute for Marine and Antarctic Studies (IMAS) or the University of Tasmania (Utas).

©The Institute for Marine and Antarctic Studies,
University of Tasmania 2024.

Copyright protects this publication. Except for purposes permitted by the Copyright Act, reproduction by whatever means is prohibited without the prior written permission of the Institute for Marine and Antarctic Studies.

Contents

Acronyms	05
1 Purpose and objectives	06
2 Background	07
3 Receiving Habitats	09
3.1 Soft sediment	09
3.1.1 Benthic Community Assessment	14
3.1.2 Abiotic measures	18
3.1.3 Visual assessment	24
3.2 Inshore Reef	28
3.2.1 Biodiversity surveys	30
3.2.2 Targeted assessment	31
3.3 Deep reefs	33
3.3.1 Deep reef quantitative biodiversity assessment	35
3.3.2 Deep reef qualitative assessment	36
3.4 Seagrass beds	36
3.4.1 Seagrass bed extent mapping	37
3.4.2 Seagrass health	37



4	Recommended techniques	39
4.1	Soft sediment sampling and analyses techniques	40
4.1.1	Benthic macrofauna sampling and analysis	40
4.1.2	Sediment core sampling	41
4.1.3	Sediment chemistry analyses	43
4.1.4	Visual assessment	45
4.1.5	eDNA	48
4.2	Inshore Reefs surveying and analyses techniques	49
4.2.1	Biodiversity survey	50
4.2.2	Targeted assessment	50
4.2.3	Remote assessments	55
4.3	Deep reefs surveying and analyses techniques	56
4.3.1	Deep Reef Quantitative Biodiversity Assessment	56
4.3.2	Deep reef Qualitative Assessment	58
4.4	Seagrass surveying and analysis techniques	59
4.4.1	Seagrass bed extent mapping	59
4.4.2	Seagrass health	60
	References	66
	Appendix A	70
	Appendix B	73

Acronyms

AUV	Autonomous underwater vehicle
C:N	Ratio of carbon to nitrogen
CATAMI	Collaborative and Automated Tools for Analysis of Marine Imagery
eDNA	Environmental DNA
EPA	Environment Protection Authority Tasmania
FRDC	Fisheries Research Development Corporation
GPS	Global Positioning System
HD	High definition
IMAS	Institute for Marine and Antarctic Studies
NAS	Nuisance algae assessment
POM	Particulate organic material
ROV	Remotely operated vehicle
RVA	Rapid Visual Assessment
SAOB	Sulphide antioxidant buffer
SMRCA	Sustainable Marine Research Collaboration Agreement
TOC	Total organic carbon
UHD	Ultra high definition
$\delta^{13}\text{C}$	A measure of the ratio of stable carbon isotopes ^{13}C : ^{12}C (‰)
$\delta^{15}\text{N}$	A measure of the ratio of stable nitrogen isotopes ^{15}N : ^{14}N (‰)

1 Purpose and objectives

This report has been produced to provide an up to date ‘how-to’ guide for monitoring the environmental effects of finfish aquaculture on marine habitats, based on the most recent research undertaken in Tasmanian coastal waters.

The ‘Guide to the assessment of sediment condition at marine finfish farms in Tasmania’ by Macleod and Forbes (2004) focused on methods for monitoring the effects of finfish farming on soft sediments. The current guide is extended to provide an overview of our current knowledge of interactions and monitoring methods for other key receiving habitats, including rocky reefs and seagrass beds.

This guide summarises the findings from numerous research projects conducted since the initial guide (Macleod & Forbes 2004) was published. Notably, Fisheries Research Development Corporation (FRDC) project 2015/024 (Managing ecosystem interactions across differing environments: building flexibility and risk assurance into environmental management strategies) and FRDC project 2018/131 (Storm Bay Observing System: Assessing the Performance of Aquaculture Development) (Strain et al. 2020, Strain et al. 2023, Ross et al. 2021). It then provides a detailed description of the most suitable monitoring design and techniques currently available for effectively carrying out environmental monitoring of impacts from finfish aquaculture on Tasmanian marine habitats.

While the methods detailed have been developed for application in finfish aquaculture environmental monitoring, it should be noted that they are equally relevant to any environmental monitoring program interested in understanding potential effects of organic enrichment and sedimentation on marine habitats more generally.

The production of this guide is supported by the FRDC and the Sustainable Marine Research Collaboration Agreement (SMRCA), which is a collaboration between the State Government and the University of Tasmania.

Report objectives

The main objective of this report is to produce an updated guide for the environmental monitoring and assessment of finfish aquaculture in Tasmania, based on the original work by Macleod and Forbes (2004):

- » to extend the scope of the guide to include additional receiving habitats: reefs and seagrass beds.
- » to provide an overview of our current knowledge of the different receiving environments (soft sediment, reefs and seagrass beds) in relation to their interactions with finfish aquaculture.
- » to provide clear methodology incorporating the most up-to-date research on design and techniques for environmental monitoring of marine habitats for the effects of finfish farming.

2 Background

The original ‘field guide’ written by Macleod and Forbes (2004) brought together the outputs of the Cooperative Research Centre project 4.1: The development of novel methods for the assessment of sediment condition and determination of management protocols for sustainable finfish cage aquaculture operations (Macleod et al. 2004), which was initiated to:

- 1 assess the rate of recovery associated with fallowing practices,
- 2 determine if (the then) current farming practices were sustainable, and
- 3 develop novel approaches for farm-based monitoring of environmental condition.

Since its publication in 2004, the finfish aquaculture industry in Tasmania has continued to intensify and expand into new areas. Significant research has been published and continues to explore how the maturing finfish aquaculture industry interacts with Tasmanian marine ecosystems:

FRDC PROJECT 2015/024

Managing ecosystem interactions across differing environments: building flexibility and risk assurance into environmental management strategies.

The aim of this research was to provide an improved understanding of the environmental interactions of salmon farming in Tasmanian marine waters and to provide recommendations to both government and industry on monitoring and management strategies that are appropriate to the level of risk associated with these interactions.

Due to changes in farming practices, innovations in technology and expansion of the industry into new areas, it was considered necessary to reassess and validate current approaches to monitoring and assessment of soft sediment habitats. To ensure these remain fit for purpose in different growing regions, extensive sampling and analysis was undertaken in Storm Bay and the southern D’Entrecasteaux Channel in the southeast, and in Macquarie Harbour on the west coast of Tasmania.

In addition, increasing community concern about the potential effects on reef systems led to the inclusion of inshore rocky reefs in the scope of the project. It was also recognised that modelling capabilities have significantly advanced since the original ‘field guide’ was published in 2004. As such, an important component of the project was to evaluate how recent modelling tools could support monitoring.



FRDC PROJECT 2018/131

Storm Bay Observing System: Assessing the Performance of Aquaculture Development

The aim of this research program was to provide a comprehensive overview of the potential interactions between salmon farming and the water column, soft-sediment, inshore reefs, deep reefs and seagrass habitats in Storm Bay.

Due to the expansion of finfish aquaculture into Storm Bay, the Storm Bay Observing System project was initiated to:

- » develop a robust monitoring program to assess the interactions between finfish farming and the receiving environment;
- » undertake mapping to provide a comprehensive map of the benthic habitats and bathymetry in the Storm Bay region and assess change at key focus areas;
- » develop and apply a lease-scale model for assessing the environmental footprint of dissolved and particulate farm inputs;
- » assess the interactions between finfish farming and the receiving environment; and
- » evaluate and review the implemented monitoring program.

While the original 'field guide' focused on interactions between finfish farming and soft sediments, these recent research projects (FRDC 2015/024 and FRDC 2018/131) have increased our understanding of how finfish aquaculture interacts with other receiving environments (reefs and seagrass beds) and have broadened the potential scope of environmental monitoring requirements for finfish aquaculture in Tasmania. As such, this updated 'field guide' has been initiated to amend content on soft sediment interactions and monitoring methodology as necessary, and to add new content around interactions with, and monitoring techniques for, additional receiving environments (reefs and seagrass) based on the best available research.

Another key component of the updated 'field guide' was to address assumptions made in the original guide around differences in depositional (low energy) and exposed (high energy) sites. The original field guide identified differences in the way environments responded based on site energy; however, the data was limited to only two sites. To verify this assumption, additional data was collected at a variety of sites exposed to different energy levels through FRDC 2015/024 (Managing ecosystem interactions across differing environments: building flexibility and risk assurance into environmental management strategies). Regionality was also considered, specifically comparing responses to organic enrichment in southeast Tasmania and Macquarie Harbour. Consequently, the updated 'field guide' aims to clearly set out key indices for assessing impact based on the energy of the environment, while also considering the potential for regional variability.

3 Receiving Habitats

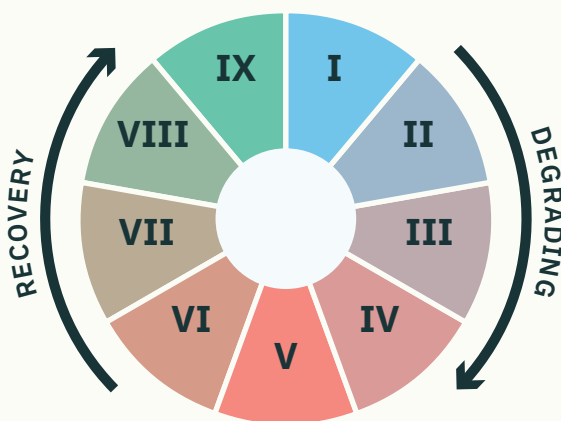
Sections 3.1 to 3.4 provide an overview of our current knowledge of the different receiving environments (soft sediment, inshore reefs, deep reefs and seagrass beds) in relation to their interactions with finfish aquaculture, based on the best available information to date.

3.1 Soft sediment

Monitoring soft sediment in proximity to finfish farms is a well-established method of assessing local-scale impacts from finfish farm discharges (e.g. Holmer et al. 2008, Macleod & Forbes 2004, Macleod et al. 2004, Keeley et al. 2015, Rector et al. 2021, Ross et al. 2021). Research from studies in Tasmania indicates a progressive and predictable environmental response to increasing enrichment and sedimentation

from finfish aquaculture (Figure 1, Table 1) (Macleod & Forbes 2004, Ross et al. 2021, Strain et al. 2023, Thompson et al. 2008, Volkman et al. 2009). Where sediment is given the opportunity to recover (i.e. where sites are fallowed or fish cages removed), recovery will also occur progressively and predictably, returning to minor effects (Figure 1).

Figure 1: Diagram showing impact and recovery stages for soft sediment in response to increasing enrichment and sedimentation from finfish aquaculture in Tasmania



Stage	Category	Description
I	Unimpacted	No evidence of farm impact
II	Minor effects	Slight infaunal and community change observed
III	Moderate effects	Clear change in infauna and chemistry
IV	Major effects (1)	Major change in infauna and chemistry
V	Major effects (2)	Bacterial mats evident, outgassing on disturbance
VI	Severe effects	Anoxic/abiotic, spontaneous outgassing
VII	Major effects	Monospecific fauna, major chemistry still clearly effected
VIII	Moderate effects	Fauna recovering, chemistry still clearly effected
IX	Minor effects	Largely recovered, although slight faunal/chemical effects still apparent

Reproduced from Macleod and Forbes (2004).



Table 1: Summary characteristics for impact stages associated with soft sediment degradation due to interactions with finfish aquaculture in Tasmania

Impact stage	Effect category	General description
Degrading ↓	I	No evidence of impact » No evidence of impact. » Environment showing no signs of enrichment in biotic, abiotic or visual indicators. Unimpacted indicator species present, showing range of functional groups. All indicators representative of reference site conditions.
	II	Minor effects » Environment showing signs of low level enrichment – zone of enhancement » Small scale community change may be observed with absence of larger, long lived species and unimpacted indicator species, however biotic indices (total abundance, species richness and Shannon index) may not differ significantly from reference site values or may show values greater than reference conditions (zone of enhancement). » Sediment chemistry is likely to remain unaffected with abiotic indicators (redox, sulphide concentration, stable nitrogen isotope) showing values comparable to reference site conditions. » Burrows and faunal tracks/ tubes are likely to be evident in visual assessment.
	III	Moderate effects » Environment showing clear evidence of enrichment. » Significant community change likely to be observed including shift to community structure dominated by deposit feeding polychaetes/ opportunists and an absence of filter and suspension feeders. Biotic indices may show signs of enrichment, with total abundance exceeding values observed in ‘Minor effects’, and species richness and Shannon index showing values less than reference conditions. » Change in sediment chemistry evident, with potential for reduced redox and elevated sulphide concentrations compared to reference site conditions. » Pellets and farmed fish faeces may be present in visual assessment.

Impact stage	Effect category	General description
Degrading ↓	IV	Major effects 1 » Environment showing strong evidence of major enrichment » Major community change will be observed in benthic community including monospecific dominance of opportunistic species (especially Capitellids or Dorvillieds). Biotic indices will likely show significant signs of enrichment with total abundance exceeding values observed in 'Moderate effects', and species richness and Shannon index showing values which may represent less than 50% of reference conditions. » Major changes in sediment chemistry will be observed compared to reference site conditions with reduced redox values, elevated sulphide values and stable nitrogen isotopes values (in south east Tasmania) likely to be reduced compared to reference site conditions. » Pellets and farmed fish faeces may be present in visual assessment.
	V	Major effects 2 » Environment showing strong evidence of major enrichment as per 'Major effects 1'. » In addition bacterial mats (e.g. <i>Beggiatoa</i>), pellets and farmed fish faeces may be present, sediments may appear blackened, and outgassing may occur from the sediment on disturbance.
	VI	Severe effects » Environment showing evidence of severe enrichment » Environment largely devoid of macrobenthic community due to abiotic conditions resulting in very low total abundance values. Spontaneous outgassing may occur from the sediment.

In areas of soft sediment, the dominant effect on the seabed occurs from the deposition of particulate organic material (POM) in the form of fish faeces and uneaten feed, which can result in localised over-enrichment. Other related effects on the seabed include those associated with biofouling, where both organic and inorganic material is either dislodged naturally or mechanically (through in situ net cleaning) from fish farm infrastructure and falls to the seabed, causing both smothering and enrichment (Bannister et al. 2019). The localised enrichment of the seabed can also attract local aggregations of scavengers and predators, including non-indigenous species (Woodcock et al. 2018).

It is important to note that in addition to the direct enrichment of sediments from particulate waste, there can also be consequences from the discharge of dissolved nutrient inputs (i.e. fish excretion) on soft sediments. Dissolved nutrient inputs are likely to stimulate production of POM (e.g. phytoplankton and bacteria) in the water column, and as such, the potential exists for this to be an indirect or secondary source of organic matter reaching the seabed. This is likely to cause a more diffuse source of POM, distributed across a broader spatial scale.



While diffuse effects may occur, the most significant benthic responses are typically seen directly beneath and adjacent to fish farm cages where most of the particulate waste is deposited (Ross et al. 2021). With increasing distance from the cage (i.e. the enrichment source), there is a strong gradient of decreasing impact (Ross et al. 2021). However, the intensity and spatial extent of the depositional footprint and intensity of benthic response is site specific, resulting largely from local hydrodynamics and the volume of depositional POM. Studies have found that high flow, deep water sites produce larger but more diffuse footprints than those located in lower flow sites with shallower water depths (Keeley et al. 2013a, Keeley et al. 2013b, Keeley et al. 2019). Mild effects can be detected up to 1,000 m from fish farms under some dispersive conditions, while in less dispersive areas, effects can be limited to within 10s of metres from farms (Keeley et al. 2013b, Keeley et al. 2019).

In Tasmania, the most significant benthic impacts have been shown to be largely restricted to within 35 m of cage edges; however, subtle effects have been evident out to 150 m at low flow, depositional sites (Edgar et al. 2010) and 200-500 m at more dispersive, high energy sites (Ross et al. 2021).

A number of measures of sediment condition can be used for monitoring effects on soft sediment, including:

- » visual assessment,
- » benthic community assessment, and
- » abiotic measures (e.g. redox potential, sulphide concentrations, carbon and nitrogen isotope signatures, sediment particle size, metals).

New Frontiers in monitoring – Environmental DNA (eDNA)

In addition to the established methods for monitoring, environmental DNA metabarcoding (eDNA metabarcoding) is now being used to identify the structure and diversity of biological communities in soft sediment (Keeley et al. 2018).

eDNA metabarcoding involves DNA sequencing of taxonomically informative genes present in DNA recovered from environmental samples (e.g. soil, water, sediment etc.) The resulting sequence data can be matched to reference databases to allow taxonomic identification (Taberlet et al. 2012). As such, DNA can be extracted from soft sediment samples to determine what taxa are present within the area, which like traditional benthic community assessment data, can be related to specific stages of organic enrichment. The method enables detection of a wider range of species than is possible using traditional benthic community assessment as it can be used to survey bacterial, foraminiferal and eukaryotic (including meiofauna and microscopic eukaryotes) communities (Keeley et al. 2018).

Several recent eDNA metabarcoding projects in the finfish farming context have developed biotic indices for environmental impact assessment (Aylagas et al. 2014, Aylagas et al. 2017, Keeley et al. 2018), cross-validating results with more traditional macrofauna and physicochemical metrics.

Measured using eDNA metabarcoding, bacterial assemblages around fish farms in New Zealand's Marlborough Sounds were shown to significantly differentiate organic enrichment gradients between reference sites, with repeatable and regionally transferable patterns (Keeley et al. 2018). Used in conjunction with the established eDNA-based bacterial index, which weights taxonomic units based on their tolerance to enrichment (Aylagas et al. 2021), results from this technique were able to assign organic enrichment impact levels just as well as traditional sample indicators (macrofauna and physico-chemical indicators) (Pochon et al. 2021). The eDNA-based bacterial index is now accepted and utilised as an alternative best practice to macrofauna sampling within soft sediment regulatory monitoring of the salmon farming industry in New Zealand (MPI 2023).

eDNA metabarcoding has been tested in Tasmania's Storm Bay to measuring both local-scale, farm-derived enrichment and broader-scale environmental change in soft sediment habitats, with results indicating bacterial communities also provide a sensitive measure of enrichment (Coutts et al. 2024). The approach has been recommended for testing more broadly in Tasmania.

For more information on the techniques used to sample for eDNA analysis see Section 4.1.5.

3.1.1 Benthic Community Assessment

The use of macrobenthic faunal community structure has been found to be a sensitive and reliable measure of sediment condition in Tasmania and overseas. While specific patterns of change in community structure are predictable and have been identified in response to specific stages of organic enrichment in soft sediments (Figure 1, Table 1) the species involved have been found to vary between leases and regions in Tasmanian coastal waters (Table 2, Table 3) (Macleod & Forbes 2004, Ross et al. 2021).

In southeast Tasmania, this largely relates to variations in the energy of the environment. Higher energy locations exhibit coarser sediment type, which generally favours higher proportions of mobile crustaceans. In contrast, lower energy environments presenting finer sediment types tend to be naturally associated with relatively lower proportions of crustaceans and higher proportions of polychaete worms (Macleod & Forbes 2004, Ross et al. 2021). Such variability affects the macrobenthic community response to sediment enrichment and recovery (Table 2) (Ross et al. 2021).

In southeast Tasmania, the most enriched sites were generally marked by a dominance of the opportunistic polychaete *Capitella* sp. in high energy environments; however, at high energy sites in the southern D'Entrecasteaux Channel, *Mediomastus* sp. was also locally abundant (Macleod & Forbes 2004, Ross et al. 2021). In low energy environments, *Capitella* sp., *Malacoceros tripartitus* and *Nebalia* sp. are indicative of high enrichment. At less enriched sites (e.g., cage sites after fallowing, or at sites 35 m from the cage), a greater variety of species, characterised by the ability to take advantage of increased food supply while also exhibiting sensitivity to highly enriched conditions, were identified.

In high energy environments, these typically include the polychaetes *Lagis portus*, *Pectinaria antipoda*, *Mediomastus* sp., *Paraprionospio coora* and *Perinereis* sp., heart urchin *Echinocardium cordatum*, amphipods from the family Phoxocephalidae, the dogwhelk *Nassarius nigellus* and the two introduced bivalves *Varicorbula gibba* and *Theora lubrica* (Ross et al. 2021). At lower energy sites, faunal communities associated with minor effects include *Nassarius* sp., *Variocorbula* sp., *Echinocardium* sp., Phoxocephalidae and Nemertea (Macleod & Forbes 2004). For sites largely unimpacted by organic enrichment, the faunal community was dominated by species which are highly sensitive to any level of enrichment. These include species like *Pratulum thetidis*, *Ampelisca cf. australis*, *Byblis mildura*, Isaeid spp., Aorid spp. and *Amphiura elandiformis* in high energy environments (Ross et al. 2021) while in lower energy environments they typically included taxa like *Amphiura elandiformis*, *Lysilla* sp., *Nucula* sp., *Thyasira* sp. (Macleod & Forbes 2004).

The benthic ecology in Macquarie Harbour on Tasmania's west coast was found to be distinctly different to that in the salmon growing regions of Tasmania's southeast. The sediment in Macquarie Harbour is inherently depauperate with all sites showing low faunal abundance, species richness and diversity relative to sites elsewhere (Ross et al. 2021). Sediment size at sites within Macquarie Harbour is also typically finer than that identified in the low energy environments sampled in the southeast of Tasmania. While reference site samples in the southeast maintained diverse faunal populations characterised by about 34 taxa per grab and a predominance of crustaceans, in Macquarie Harbour an average of five taxa were identified per grab, with a predominance of surface deposit-feeding polychaetes (which are usually associated with local scale organic enrichment) (Ross et al. 2021).

In addition to distinct differences in response to farming pressure compared to southeast Tasmania, there was also considerable variability in macrofaunal response between leases and surveys within Macquarie Harbour (Ross et al. 2021). This is thought to be the result of variation in local environmental conditions (i.e. proximity to the ocean) and farm management approaches (i.e. feed input and site history) across the harbour, affecting bottom water oxygen concentrations and sediment enrichment, respectively (Ross et al. 2021).

In contrast to the southeast of Tasmania, where *Capitella* sp. is the dominant species associated with organic enrichment, highly enriched sites in Macquarie Harbour are dominated by the opportunistic dorvilleid polychaetes *Schistomeringos loveni* and *Ophryotrocha shieldsi*, and the leptostracean crustacean *Nebalia* sp. (Ross et al. 2021). But note, *Capitella* sp. is becoming increasingly common at farm sites in Macquarie Harbour. These species are all characterised by an ability to quickly adapt to and exploit changes in organic loading due to short generation times, which give them the advantage over other species. In less enriched areas in Macquarie Harbour, macrofauna that are less tolerant of highly enriched conditions are characterised by a mix of sessile suspension feeders and surface deposit feeders, including the terebellid *Pista australis*, the spionid *Pseudopolydora* cf. *paucibranchiata*, the sabellid *Branchiommma* sp., the ampharetid *Samythella* sp., the echinoderm *Echinocardium cordatum* (Ross et al. 2021).

In addition to the presence or absence of indicator species, characteristics of the benthic community such as total abundance, species richness and species diversity (Shannon index), can also be used to assess sediment health (Ross et al. 2021). While these biotic measures can provide valuable insight into the benthic community structure, significant variation between leases and regions has been observed. This is particularly true for total abundance in high energy environments, where sites exhibiting comparable levels of organic enrichment have returned a wide range of abundance values (Table 2) (Ross et al. 2021).

The observed variability in biotic indices between leases and regions in Macquarie Harbour and southeast Tasmania highlights the importance of establishing local reference conditions through baseline surveys¹ and the ongoing monitoring of reference sites. This will underpin the development of performance indicators and their assessment. Table 2 provides a summary of the effect indices for degrading and recovering sediments in low energy sites and high energy sites in the finfish-growing regions of southeast Tasmania, while Table 3 provides a summary for Macquarie Harbour based on the best available information to date. ***Again, it is important to reiterate that the information within these tables needs be considered in conjunction with site specific information.***

¹ To ensure that reference conditions established prior to development are robust and account for temporal variation, we recommend that the 'baseline' is based on multiple surveys.



Table 2: Biotic finfish farm effect indices for low and high energy environments in the southeast of Tasmania, Australia

* Species indicative of a specific effect level in combination rather than individually.

** Southern D'Entrecasteaux Channel only.

† Species associated with net biofouling.

Impact stage		Degrading				
		I	II	III	IV	
Effect category		No evidence of impact	Minor effects	Moderate effects	Major effects 1	
Description		No evident change	Small scale community change. Zone of enhancement.	Significant community change: Rapid change in community mix.	Major community change including monospecific dominance	
Functional categories		Unimpacted indicator species present	Absence of larger, long lived species and pristine indicator species.	Deposit feeding polychaetes/ opportunist species dominate. Absence of filter/ suspension feeders.	Opportunist species (esp. <i>Capitella</i> sp./ <i>Nebalia</i> sp.) characterise the community	
Indicator species	Low energy	» <i>Amphiura elandiformis</i> » <i>Lysilla</i> sp. » <i>Mediomastus</i> sp.* » <i>Nuculanidae</i> * » <i>Thyasiridae</i> *	» <i>Capitella</i> sp. » <i>Nassarius nigellus</i> * » <i>Variocorbula gibba</i> * » <i>Echinocardium cordatum</i> * » <i>Phoxocephalidae</i> * » <i>Nemertea</i> *	» <i>Capitella</i> sp. » <i>Nebalia</i> sp. » <i>Corbula gibba</i> * » <i>Nassarius nigellus</i> * » <i>Neanthes</i> sp.*	» <i>Capitella</i> sp. » <i>Nebalia</i> sp. (dominant) » <i>Variocorbula gibba</i> * » <i>Nassarius nigellus</i> * » <i>Neanthes</i> sp.* » <i>Malacoceros tripartitus</i>	
	High energy	» <i>Apseudidae</i> » <i>Ampeliscidae</i> , » <i>Pratulum thetidis</i> » <i>Ampelisca cf. australis</i> » <i>Byblis mildura</i> » <i>Isaeidae</i> , <i>Aoridae</i> » <i>Amphiura elandiformis</i> **	» <i>Lyssianassidae</i> * » <i>Euphilomedes ferox</i> * » <i>Polydora cf socialis</i> * » <i>Phoxocephalidae</i> * » <i>Pectinaria</i> spp. » <i>Mediomastus</i> sp. » <i>Perinereis</i> sp. » <i>Echinocardium cordatum</i> » <i>Phoxocephalidae</i> » <i>Nassarius nigellus</i> » <i>Variocorbula gibba</i> » <i>Theora lubrica</i> » <i>Pagurixus handrecki</i>	» <i>Capitella</i> sp. (dominant); » <i>Mediomastus</i> sp.** » <i>Neanthes</i> sp. » <i>Corophium</i> sp.* » <i>Polydora cf socialis</i> * » <i>Tethygenia</i> sp.* » <i>Phoxocephalidae</i> * » <i>Pectinaria</i> spp. » <i>Perinereis</i> sp. » <i>Echinocardium cordatum</i> » <i>Nassarius nigellus</i> » <i>Corbula gibba</i> » <i>Theora lubrica</i> » <i>Paraprionospio coora</i> » <i>Edwardsia</i> sp. » <i>Pagurixus handrecki</i> » <i>Asterias amurensis</i>	» <i>Capitella</i> sp. (dominant) » <i>Neanthes</i> sp.* » <i>Dimorphostylis</i> * » <i>Mytilus galloprovincialis</i> † » <i>Jassa</i> spp.† » <i>Nebalia</i> sp. » <i>Pleurobranchaea maculata</i> » <i>Asterias amurensis</i>	
Total abundance (N)	Low energy	Same as ref		10 x ref	20 x ref	
	High energy	Same as ref		1 – 3 x ref	>2-3 x ref†	
Species richness (S)	Low energy	Same as ref	Same as ref	Reduction compared to ref; no. species < ref	Increased reduction; no. species < ref ^s	
	High energy	Same as ref	Same as ref	Reduction compared to ref; no. species < ref	Increased reduction; no. species < ref ^s	
Shannon Index (H')	Low energy	>2	>2	1-2; 0.5-1 x ref	<1; <0.5 x ref	
	High energy	>2.5	>2.5	1.5-2.5; 0.5-1 x ref	<1.5; <0.5 x ref	

‡ Significant variability depending on location, highlighting the importance of undertaking site specific baseline surveys. Sites in the southern D'Entrecasteaux Channel typically showed total abundance in the range of 8 x reference sites, while in Storm Bay they did not exceed 3 x reference total abundance at the most impacted sites.

§ Significant variability in species richness may be observed between surveys and sites. Change in species richness should be used in relation to change from reference sites and in conjunction with other indicators of effect.

Recovering					
V	VI	VII	VIII	IX	
Major effects 2	Severe effects	Major effects	Moderate effects	Minor effects	
Major community change with <i>Beggiatoa</i>	Devoid of macrobenthic community	Monospecific dominance	Zone of enhancement	Community largely recovered	
Opportunist species (esp. <i>Capitella</i> sp./ <i>Nebalia</i> sp.) dominate. Patchy <i>Beggiatoa</i> present	Abiotic conditions	Opportunist species (esp. <i>Capitella</i> sp.) dominate but numbers dropping and other species colonising.	Presence of transitional species with notable increase in epibenthic opportunist species	Diversification of community but absence of long lived species and pristine indicator species.	
» <i>Capitella</i> sp., » <i>Nebalia</i> sp. (extremely dominant) » <i>Malacoceros tripartitus</i>	Absence of species	» <i>Capitella</i> sp., » <i>Nebalia</i> sp. (abundant) » <i>Nassarius nigellus</i> * » <i>Neanthes</i> * » <i>Corbula</i> * » <i>Phoxocephalidae</i> * » <i>Malacoceros tripartitus</i>	» <i>Capitella</i> sp. » <i>Nebalia</i> sp. (decreasing abundance); » <i>Nassarius nigellus</i> * » <i>Echinocardium cordatum</i> * » <i>Phoxocephalidae</i> *	» <i>Nassarius nigellus</i> » <i>Corbula gibba</i> » <i>Neanthes</i> * » <i>Echinocardium cordatum</i> * » <i>Phoxocephalidae</i> * » <i>Nemertea</i> *	
» <i>Capitella</i> sp. (greatly dominant) » <i>Neanthes</i> sp. » <i>Nebalia</i> sp. » <i>Pleurobranchaea maculata</i> . » <i>Asterias amurensis</i>	Absence of species	» <i>Capitella</i> sp. (dominant) » <i>Neanthes</i> sp.* » <i>Corophium</i> spp.* » <i>Nebalia</i> sp.* » <i>Phoxocephalidae</i> *	» <i>Capitella</i> sp. (lower numbers) » <i>Mediomastus</i> sp. » <i>Euphilomedes ferox</i> * » <i>Polydora cf socialis</i> * » <i>Euchone</i> spp.* » <i>Paraprionospio coora</i>	Mix of species with increasing crustacea and decreasing annelids: » <i>Apseudes</i> * » <i>Polydora cf socialis</i> * » <i>Euphilomedes</i> * » <i>Nephtys</i> sp.* » <i>Pectinaria</i> spp. » <i>Mediomastus</i> sp. » <i>Perinereis</i> sp. » <i>Echinocardium cordatum</i> » <i>Phoxocephalidae</i> » <i>Nassarius nigellus</i> » <i>Corbula gibba</i> » <i>Theora lubrica</i>	
20 x ref	Not observed	20 x ref	10 x ref		
>2-3 x ref‡	Not observed	>2-3 x ref‡	1 – 3 x ref		
Further reduction; potentially <0.5 x ref§	Not observed	< ref; potentially <0.5x ref§	Reduction compared to ref; no. species < ref	no. species same as ref	
Further reduction; potentially <0.5 x ref§	Not observed	< ref; potentially <0.5 x ref§	Reduction compared to ref; no. species < ref	no. species same as ref	
<1; <0.5 x ref	Not observed	<1; <0.5 x ref	1-2; >0.5-1 x ref	>2	
<1.5; <0.5 x ref	Not observed	<1.5; <0.5 x ref	1.5-2.5; 0.5-1 x ref	>2.5	

Table 3: Biotic finfish farm effect indices for Macquarie Harbour, Tasmania, Australia

Impact stage	Degrading				
	I	II	III	IV	
Effect category	No evidence of impact	Minor effects	Moderate effects	Major effects 1	
Description	No evident change	Small scale community change. Zone of enhancement.	Significant community change: Rapid change in community mix.	Major community change including monospecific dominance	
Functional categories	Unimpacted indicator species present	Absence of larger, long lived species & pristine indicator species.	Deposit feeding polychaetes/ opportunist species dominate. Absence of filter/ suspension feeders.	Opportunist species (esp. <i>Capitellida</i> sp./ <i>Dorvillidae</i>) characterise the community	
Indicator species		» <i>Pista australis</i> » <i>Pseudopolydora</i> cf. <i>paucibranchiata</i> » <i>Branchioma</i> sp. » <i>Samyrella</i> sp. » <i>Echinocardium cordatum</i> » <i>Parathyasira resupina</i>	» <i>Pista australis</i> » <i>Pseudopolydora</i> cf. <i>paucibranchiata</i> » <i>Samyrella</i> sp. <i>Echinocardium cordatum</i>	» <i>Dorvillia</i> (<i>Schistomeringos loveni</i> [dominant], <i>Ophryotrocha shieldsi</i>) » <i>Nebalia</i> sp.	
Total abundance (N)*	Same as ref*	1-2 x ref*	2-10 x ref*	1-5 x ref*	
Species richness (S)*	Same as ref*	Same as ref*	1-3 x ref	1-2	
Shannon Index (H')*	0.5-2	> ref	> ref	0.3-1; <ref	

* Significant variability depending on lease location in the harbour and the environmental conditions (e.g. bottom water dissolved oxygen conditions) at the time of the survey. The metrics total abundance, richness and diversity metrics are typically reduced further into the harbour and the background environmental conditions have the capacity to override the typical patterns observed in response to enrichment, notably during periods of very low bottom water oxygen conditions (see Ross et al. 2021).

3.1.2 Abiotic measures

Abiotic indices remain a valuable monitoring tool in environmental monitoring of the marine environment, providing information on prevailing environmental and sediment condition (Environment Protection Authority (EPA) Tasmania 2020, Ross et al. 2021). A variety of abiotic indices are available for use, including redox potential, sulphide concentrations, carbon and nitrogen stable isotope signatures, particle size description and metals analysis. Several of these parameters (redox, sulphide, particle size analysis and stable isotopes) are

included in finfish aquaculture regulatory Broadscale Environmental Monitoring Programs (or BEMP) as part of Marine Farm Licence conditions, baseline assessments or when more detailed environmental surveys are required to be undertaken (EPA Tasmania 2020).

While abiotic indices alone are not recommended for providing a complete picture of sediment condition in Tasmania, they are an important tool for understanding the processes occurring in soft

			Recovering		
	V	VI	VII	VIII	IX
	Major effects 2	Severe effects	Major effects	Moderate effects	Minor effects
	Major community change with <i>Beggiatoa</i>	Devoid of macrobenthic community	Monospecific dominance	Zone of enhancement	Community largely recovered
	Opportunist species (esp. <i>Capitella</i> sp./ <i>Dorvilliedae</i>) dominate. Patchy <i>Beggiatoa</i> , matts of <i>Beggiatoa</i> may also be present	Abiotic conditions. Extensive matts of <i>Beggiatoa</i> often present	Opportunist species (esp. Capitellids/ <i>Dorvilliedae</i>) dominate but numbers dropping and other species colonising	Presence of transitional species with notable increase in epibenthic opportunist species	Diversification of community but absence of long lived species and pristine indicator species.
	» <i>Beggiatoa</i> and <i>Dorvillied</i> (<i>Ophryotrocha shieldsi</i> [dominant], <i>Schistomeringos loveni</i>) » <i>Nebalia</i> sp. » <i>Capitella</i> sp. » <i>Charcotia australiensis</i>	Absence of species	» <i>Dorvillied</i> (<i>Schistomeringos loveni</i> [dominant], <i>Ophryotrocha shieldsi</i>), » <i>Nebalia</i> sp.	» <i>Pista australis</i> » <i>Pseudopolydora</i> cf. <i>paucibranchiata</i> » <i>Samythella</i> sp. » <i>Echinocardium cordatum</i>	» <i>Pista australis</i> » <i>Pseudopolydora</i> cf. <i>paucibranchiata</i> » <i>Branchiomma</i> sp. » <i>Samythella</i> sp. » <i>Echinocardium cordatum</i> » <i>Parathyasira resupina</i>
	0.5-5 x ref*	Fauna absent	1-5 x ref*	1-5 x ref*	1-2 x ref*
	< ref	Fauna absent	1-2	1-2 x ref	Same as ref*
	0.3-1; <ref	Fauna absent	0.3-1; <ref	> ref	> ref

sediment habitats, often adding to the interpretation of macrofaunal analysis (Ross et al. 2021). Variability in soft sediment macrofauna is often affected by naturally occurring variability in environmental conditions (e.g. sediment grain size and oxygen availability), in addition to anthropogenic pressures (e.g. organic enrichment or metal contamination). Monitoring both together provides a complete picture of the processes acting at a given location.

Table 4 and Table 5 provide a summary of the abiotic indices for degrading and recovering sediments in southeast Tasmania (low energy and high energy sites) and for Macquarie Harbour, respectively, based on the best available information to date. It should be noted, however, that these indices should be considered in conjunction with site specific data. Further details of the different abiotic indices commonly used are provided in the sections below.



Table 4: Abiotic finfish farm effect indices for low and high energy environments in the southeast of Tasmania, Australia

Impact stage		Degrading				
		I	II	III	IV	
Effect category		No evidence of impact	Minor effects	Moderate effects	Major effects 1	
Description	Low energy	No evident change	Sediment chemistry unaffected or with only very minor effects	Sediment chemistry affected	Major sediment chemistry changes	
	High energy					
Redox potential	Low energy	Typically >100mV	Typically, 0-100mV*; 0.5-1 x ref	Typically, 0-100mV*; 0.5-1 x ref	Typically <0mV	
	High energy	Typically >250mV	150-250mV*; 0.5-1 x ref	100-200mV*; 0.5-1 x ref	Typically -100-100mV*; <0.5 x ref	
Sulphide concentration	Low energy	<10µM	10-30µM*	30-50µM*, 1-3 x stage II	50-150µM*; 2-3 x stage III	
	High energy	<10µM	10-30µM*	30-100µM*, 1-3 x stage II	50-150µM*; 2-3 x stage II	
Stable nitrogen isotope	Low energy	Not assessed	Not assessed	Not assessed	Not assessed	
	High energy	>8 δ ¹⁵ N		7-8 δ ¹⁵ N	<7 δ ¹⁵ N	

* Variability depending on location, highlighting the importance of undertaking site-specific baseline surveys
e.g. sites in the southern D'Entrecasteaux Channel are typically at the lower end of the range relative to Storm Bay.

Table 5: Abiotic finfish farm effect indices for Macquarie Harbour, Tasmania, Australia

Impact stage		Degrading				
		I	II	III	IV	
Effect category		No evidence of impact	Minor effects	Moderate effects	Major effects 1	
Description	Low energy	No evident change	Sediment chemistry unaffected or with only very minor effects	Sediment chemistry affected	Major sediment chemistry changes	
	High energy					
Redox potential		-50 to 50mV*	-50 to 50mV ¹	-100 to 0mV*	-150 to -50mV*	
Sulphide concentration		<10µM		10-20µM**	10-40µM**	
Stable nitrogen isotope		2-5δ ¹⁵ N		3-6 δ ¹⁵ N	3-7 δ ¹⁵ N	

* Redox (mV) show high variability is space and time in Macquarie Harbour, and as such, are more reliably used to assess relative differences across an enrichment gradient at a particular lease location and survey.

** Sulphide concentrations in Macquarie Harbour are lower than expected, thought to be related to scavenging by metals present in sediment. Like redox, sulphide shows high variability is space and time in Macquarie Harbour, and as such, are more reliably used to assess relative differences across an enrichment gradient at a particular lease location and survey.

			Recovering		
	V	VI	VII	VIII	IX
	Major effects 2	Severe effects	Major effects	Moderate effects	Minor effects
	Outgassing on disturbance of sediment	Anoxic, spontaneous outgassing	Major sediment chemistry effects	Sediment chemistry still affected	Sediment chemistry still slightly affected
					Sediment chemistry recovered
	Typically <0mV	Conditions not observed	<0mV	0-100mV*; 0.5-1x ref	0-100mV*; 0.5-1x ref
	Typically -100-100mV*; <0.5 x ref		Typically -100-100mV*	100-200mV*; 0.5-1x ref	0-100mV*; 0.5-1x ref
	50-150µM*; 2-3 x stage III		>50µM	>20µM	>20µM
	50-150µM*; 2-3 x stage II		>50µM	>20µM	>20µM
	Not assessed				
	<7 δ ¹⁵ N				

			Recovering		
	V	VI	VII	VIII	IX
	Major effects 2	Severe effects	Major effects	Moderate effects	Minor effects
	Outgassing on disturbance of sediment	Anoxic, spontaneous outgassing	Major sediment chemistry effects	Sediment chemistry still affected	Sediment chemistry recovered
					Sediment chemistry still slightly affected
	-150 to -50mV*	-200 to -100mV*	-150 to -50mV*	-100 to 0mV*	-50 to 50mV*
	10-40µM**	10-40µM**	10-40µM**	10-20µM**	<10µM
	3-7 δ ¹⁵ N	3-7 δ ¹⁵ N	3-7 δ ¹⁵ N	3-6 δ ¹⁵ N	

Sediment redox potential

The analysis of sediment redox levels provides valuable information on the oxidation/reduction state of the sediment, which relates to the degree of sediment enrichment. While sediment enrichment can occur naturally in some situations, it is often associated with anthropogenic sources of organic matter discharge.

In Tasmania, sediment redox conditions show large variability between regions due to diverse environmental conditions. While sites in the southeast of Tasmania are characterised by oxidising conditions (redox potential typically >300 mV), in Macquarie Harbour sediment conditions are highly reducing (redox potential typically <50 mV) (Ross et al. 2021).

The reducing benthic environment in Macquarie Harbour is largely a function of the bathymetry of the harbour, which results in a highly stratified water column. A shallow and narrow entrance to the ocean restricts oceanic water entering the deep central basin of the harbour, and together with significant river inputs, causes bottom waters to be naturally depleted in oxygen (Ross et al. 2021). This results in a gradient in bottom water oxygen concentrations (and redox levels) with increasingly reducing conditions further from the harbour entrance (Ross et al. 2021). The naturally reducing conditions of Macquarie Harbour influence the benthic ecology and subsequently, the way in which macrofaunal communities respond to organic enrichment (Ross et al. 2021). In contrast, the sites in the southeast of Tasmania are typically located in more open, well-mixed areas and bottom waters are generally high in dissolved oxygen. The benthic macrofaunal communities at these sites are generally less tolerant of reducing conditions (Macleod & Forbes 2004, Ross et al. 2021). Therefore, understanding the context of redox potential measurements at any given site is an important component of understanding sediment redox levels and the macrobenthic response to any potential enrichment. This is particularly important in areas like Macquarie Harbour, where anaerobic respiration and reducing conditions naturally dominate (Ross et al. 2021).

Table 4 and Table 5 provide a summary of the sediment redox levels associated with different levels of enrichment for southeast Tasmania and for Macquarie Harbour, respectively, based on the best available information to date; however, this should be considered in conjunction with site-specific and/or baseline information.

Sulphide concentration

Like redox, the measurement of sulphide concentration in soft sediment can be used as a means of determining the enrichment of sediment (EPA Tasmania 2020, Keeley & Taylor 2015). In Tasmania, sulphide is considered a suitable monitoring tool, however, measured concentrations have been found to vary significantly with location, indicating that its use may be more suited to describing spatial patterns of enrichment rather than temporal change (Ross et al. 2021).

In Macquarie Harbour, however, sulphide concentrations are unexpectedly low given the known condition of benthic sediment and oxygen availability. This is thought to be related to scavenging of sulphide by iron minerals present in the sediment (Ross et al. 2021). As a result, sulphide concentrations in Macquarie Harbour need to be interpreted carefully, with particular consideration to background environmental conditions, potentially reducing their reliability as an indicator of sediment enrichment in this region (Ross et al. 2021).

Table 4 and Table 5 provide a summary of the sediment sulphide concentrations associated with different levels of enrichment for southeast Tasmania and for Macquarie Harbour, respectively, based on the best available information to date; however, this should be considered in conjunction with site-specific and/or baseline information.

Stable isotope signatures and organic content

Stable nitrogen and carbon isotope signatures of sediments can be a useful tool for providing insight into differences in benthic environments and sources of organic material at different sites/regions. Marine and terrestrial/freshwater sources of organic matter present different ratios of stable nitrogen isotopes (^{15}N : ^{14}N or $\delta^{15}\text{N}$) and stable carbon isotopes (^{13}C : ^{12}C or $\delta^{13}\text{C}$), which can be used to determine the source of potential organic enrichment. While marine organic matter usually has a $\delta^{15}\text{N}$ signature of 5-7‰ and a $\delta^{13}\text{C}$ signature of -22‰ to -18‰, terrestrial organic matter usually has a $\delta^{15}\text{N}$ signature of <4‰ and a $\delta^{13}\text{C}$ signature of -25‰ to -33‰ (Middelburg & Nieuwenhuize 1998).

As finfish feed now comprises a higher proportion of terrestrial-based products, finfish farm waste typically has a more terrestrial signature. As such, stable isotopes can be used to help identify organic enrichment from finfish aquaculture where prevailing background stable isotope signatures for an area are known (Ross et al. 2021). In Tasmania, the background environmental $\delta^{15}\text{N}$ and $\delta^{13}\text{C}$ signatures vary between regions with $\delta^{15}\text{N}$ of 8-9‰ and $\delta^{13}\text{C}$ -20‰ to -21‰ in Storm Bay and $\delta^{15}\text{N}$ of 8.5‰ and $\delta^{13}\text{C}$ -23‰ in the Southern Channel, indicating a mixed oceanic and marine signal (Ross et al. 2021). In Macquarie Harbour the signature is more terrestrial ($\delta^{15}\text{N}$ 3-4‰ and $\delta^{13}\text{C}$ -26‰ to -27‰) due to the influence of the Gordon River delivering fresh water to the harbour (Ross et al. 2021). While $\delta^{13}\text{C}$ isotopic signatures of sediment have not been found to be sensitive enough to independently identify enrichment levels in Tasmania, $\delta^{15}\text{N}$ isotopic signatures have been successfully applied (Ross et al. 2021).

Similarly, carbon/nitrogen molar ratios (C:N) can be used to provide evidence of enrichment sources where more depleted ratios are more typical of finfish feed inputs (Ross et al. 2021). However, application in Tasmania has shown mixed success depending on lease and region, with some leases showing clear effects with distance from cages while others do not (Ross et al. 2021).

In southern D'Entrecasteaux Channel, the C:N at sites closest to cages (e.g. at 0 m from cage) were indicative of feed inputs, however, sites further from cage edge did not generally show any clear evidence of enrichment (Ross et al. 2021). Similarly, sites in Storm Bay did not show any clear change in C:N ratio with distance from cage edge, suggesting that C:N ratios offer limited potential for monitoring impacts from finfish aquaculture in southeast Tasmania. In Macquarie Harbour, C:N ratios showed more potential in identifying a gradient of finfish impacts with distance from cage edge, however variations between leases were also evident (Ross et al. 2021).

Organic carbon (C) and organic nitrogen (N) content can also be used to provide information on the health of sediment, where more enriched sediments typically show higher C and N content. Gradients of decreasing C and N content have been observed with distance from cage edge in Macquarie Harbour and Storm Bay, however while organic nitrogen showed effects with distance in the Southern Channel, organic carbon was highly variable across sites (Ross et al. 2021).

While carbon stable isotope signatures, C:N ratios and organic C and N content all offer valuable information for establishing background environmental conditions in Tasmania, the variability observed between leases, regions and surveys highlights the importance of using them in conjunction with other biotic and abiotic measures to provide context when describing monitoring results. However, the $\delta^{15}\text{N}$ isotopic signature of sediments is considered to be a sensitive indicator of enrichment levels, particularly in the more marine locations of southeast Tasmania (Ross et al. 2021). Table 4 and Table 5 provide a summary of the $\delta^{15}\text{N}$ isotopic signature of sediments associated with different levels of enrichment for southeast Tasmania and for Macquarie Harbour respectively. This summary is based on the best available information to date, but should be considered in conjunction with site-specific and/or baseline information.



Sediment particle size

Sediment particle size influences both the type of organisms and the physico-chemical processes which act on the sediment, and consequently affects the response to organic enrichment (Ross et al. 2021). Therefore, while information on sediment particle size does not provide direct information on the health of soft sediment habitats, it offers important information that can help build context when interpreting other biotic and abiotic monitoring results.

Metals

Like sediment particle size, measuring concentrations of metals within sediment can provide important contextual information for interpreting the results of other biotic and abiotic measures (e.g. sulphide in Macquarie Harbour) (Ross et al. 2021). While use of copper-based antifouling is no longer common practice in Tasmania (Bannister et al. 2019), monitoring for metals may remain an important component of finfish aquaculture monitoring in areas where farm management activities release metals into the environment or where farming activities may influence their release from contaminated sediments (Macleod et al. 2014, Tett et al. 2018).

3.1.3 Visual assessment

Visual assessment using a Remotely Operated Vehicle (ROV) has been found to be a reliable, sensitive, rapid and cost-effective method for determining sediment health in all regions of Tasmania where site specific criteria and weightings are applied (Macleod & Forbes 2004, Ross et al. 2021). Specific and predictable patterns of change in visual appearance of the sediment have been identified for Macquarie Harbour and the southeast of Tasmania in response to increasing stages of organic enrichment and recovery (Table 6) based on a defined scoring system (Table 9).

Sediment health is assessed using a wide variety of easily identifiable visual indicators which are weighted in relation to their relative importance within an objective scoring system of sediment condition (Macleod & Forbes 2004, Ross et al. 2021). They include sediment characteristics, visible fauna, biofouling debris, pellets, faeces (for a complete list see Table 9 in Section 4.1.4). Enriched sites are more likely to exhibit darker sediment with visible bacterial (*Beggiatoa*) mats together with high numbers of opportunistic fauna, low numbers or absence of species sensitive to enrichment and evidence of finfish farm waste including fish feed pellets, faeces, biofouling debris. In situations where severe effects are observed, sediments may be devoid of any observable fauna.

Like benthic community assessments, the species observed in visual assessments varies between high and low energy sites in southeast Tasmania and Macquarie Harbour for different levels of enrichment. However, change in functional groups remains largely consistent across sites exposed to varying degrees of energy (Ross et al. 2021). In Macquarie Harbour, opportunistic species primarily consist of dorvilleid polychaetes, while in the southeast of Tasmania, enriched sites were generally characterised by polychaete species from the Capitellidae family, with *Capitella* sp. being the dominant species in both high and low energy environments. *Mediomastus* sp. were also identified in large numbers in the high energy sites of the southern D'Entrecasteaux Channel (Ross et al. 2021).

With increasing sediment health, the numbers of opportunistic species and presence of feed, faeces and biofouling debris are seen to decline and evidence of increasing faunal diversity, including a higher presence of species less tolerant of organic enrichment, is observed. Outgassing is likely to be absent and sediment colour should appear normal compared to reference sites. Visible signs of faunal activity including burrows, mounds, worm tubes, casts and faunal tracks are characteristic of this transitory stage (Macleod & Forbes 2004, Ross et al 2021). In Macquarie Harbour, this stage is characterised by Sabellids, Annelids, heart urchins (*Echinocardium cardatum*) and swarming epibenthic crustaceans, which can be identified by visual video assessment (Macleod & Forbes 2004, Ross et al. 2021). In southeast Tasmania characteristic fauna include brittle stars, squat lobsters (*Munida haswelli*) and dog whelk (*Nassarius nigellus*) in low energy sites, while Echiurans and Sipunculans are common in high energy environments (Macleod & Forbes 2004, Ross et al. 2021).

Where impacts of organic enrichment are largely absent, no evidence of change compared to reference sites is observed in visual assessment. This includes an absence of fish farm waste, sediment colour which appears normal when compared with reference conditions and the presence of unimpacted indicator species. In southeast Tasmania, these typically include macroalgae, echiurans and sipunculans in high energy environments, and brittlestars in low energy environments (Macleod & Forbes 2004, Ross et al. 2021).

While visual assessment using video with specific visual criteria and weightings offer a reliable method for determining sediment health between ‘moderate effects’ and ‘major effects’, it is less sensitive at discriminating between areas of ‘minor impacts’ and areas with ‘no evidence of impact’. Although identifying differences between impact stages is important across the whole spectrum of enrichment, discriminating between the more enriched stages is likely to be of greatest importance for day-to-day farm management. Visual assessment conducted using Remotely Operated Vehicle (ROV) footage based on the presence or absence of key criteria for ‘unacceptable impact’ is currently undertaken as part of ongoing regulatory monitoring by finfish aquaculture operators (EPA Tasmania 2020). While this method does provide relevant information on unacceptable impacts as determined by the regulator, the same video footage can be used to provide a more detailed and informative assessment of sediment health when it incorporates objective scoring of diverse criteria together with weightings, as described by Ross et al. (2021).

For use as a management tool, visual video assessments are best employed as part of an ongoing time series together with ongoing monitoring of reference sites. When applied in this way, interpretation of results is optimised, minimising the effect of variability in scores between sites and over time (Ross et al. 2021). Interpretation can be further aided by employing visual assessment in conjunction with other soft sediment assessment methods (i.e. benthic community assessment and abiotic measures) to provide a more detailed understanding of finfish farm effects (Ross et al. 2021).



Table 6: Visual assessment indicators for low and high energy environments in southeast Tasmania, and for Macquarie Harbour

Impact stage		Degrading				
		I	II	III	IV	
Effect category		No evidence of impact	Minor effects	Moderate effects	Major effects 1	
Description		No evident change	Zone of enhancement; slight community change observed	Significant community change: Rapid change in community mix.	Major community change and visible change in sediment characteristics	
Functional categories		Unimpacted indicator species present	Absence of larger, long-lived species and pristine indicator species.	Deposit feeding polychaets/ opportunist species dominate. Absence of filter/ suspension feeders.	Opportunist species (esp. Capitellids/ Dorvillies) characterise the community	
Video score	Low energy (incl. Macquarie Harbour)	>5	2.5 to 5	<2.5	Negative	
	High energy	>2.5	2.5 to 5	<2.5	Negative	
Video features	Low energy (incl. Macquarie Harbour)	» Brittlestars	» Prevalence of burrow/ faunal tracks/tubes » squat lobsters » dog whelk (<i>Nassarius</i> sp.)	» Squat lobsters » dog whelk (<i>Nassarius</i> sp.) » pellets » farmed fish faeces	» Continuous patches/ mats of <i>Beggiatoa</i> » gas bubbles » black sediments » pellets » farmed fish faeces	
	High energy	» Algae » Echiurans » Sipunculans	» Prevalence of burrow/ faunal track/tubes » Echiurans » Sipunculans	» Side Gill Slugs (<i>Pleurobranchia maculata</i>) » Pacific Seastar (<i>Asterias amurensis</i>) » pellets » farmed fish faeces » biofouling debris (mussel shells, ascidians, algae)	» Side Gill Slugs (<i>Pleurobranchia maculata</i>) » Pacific Seastar (<i>Asterias amurensis</i>) » Any evidence of <i>Beggiatoa</i> » gas bubbles » black sediment » pellets » farmed fish faeces » excessive biofouling debris (mussel shells, ascidians, algae)	

			Recovering		
	V	VI	VII	VIII	IX
	Major effects 2	Severe effects	Major effects	Moderate effects	Minor effects
	Major community change and visible change in sediment with <i>Beggiatoa</i>	Devoid of macrobenthic community with significant sediment change	Major community change (return to monospecific dominance) and visible change in sediment characteristics	Fauna re-establishing: zone of enhancement	Community largely recovered
	Opportunist species (esp. Capitellids/ Dorvillieds) dominate. Patchy <i>Beggiatoa</i> present		Opportunist species (esp. Capitellids) dominate but numbers dropping and other species colonising.	Presence of transitional species with notable increase in epibenthic opportunist species	Diversification of community but absence of long-lived species and pristine indicator species.
	Negative	Conditions not observed; < -10	Negative	<2.5	2.5 to 5 (stabilisation of cumulative scores within this range)
	Negative	Conditions not observed; < -10	Negative	<2.5	2.5 to 5 (stabilisation of cumulative scores within this range)
	» Continuous patches/mats of <i>Beggiatoa</i> » gas bubbles » black sediments	Conditions not observed	» Continuous patches/mats of <i>Beggiatoa</i> » gas bubbles » black sediments	» Squad lobsters » dog whelk (<i>Nassarius</i> sp.)	Prevalence of burrow/ faunal track/tubes; brittle stars, squat lobsters, dog whelk (<i>Nassarius</i> sp.)
	» Side Gill Slugs (<i>Pleurobranchia maculata</i>) » Pacific Seastar (<i>Asterias amurensis</i>) » Any evidence of <i>Beggiatoa</i> » gas bubbles » black sediment » excessive biofouling debris (mussel shells, ascidians, algae)	Conditions not observed	» Side Gill Slugs (<i>Pleurobranchia maculata</i>) » Pacific Seastar (<i>Asterias amurensis</i>) » any evidence of <i>Beggiatoa</i> » gas bubbles » black sediment » excessive biofouling debris (mussel shells, ascidians, algae)	» Side Gill Slugs (<i>Pleurobranchia maculata</i>) » North Pacific Seastar (<i>Asterias amurensis</i>) » biofouling debris (mussel shells, ascidians, algae)	Prevalence of burrow/ faunal track/tubes; Point at which sea slugs are displaced (temporal)

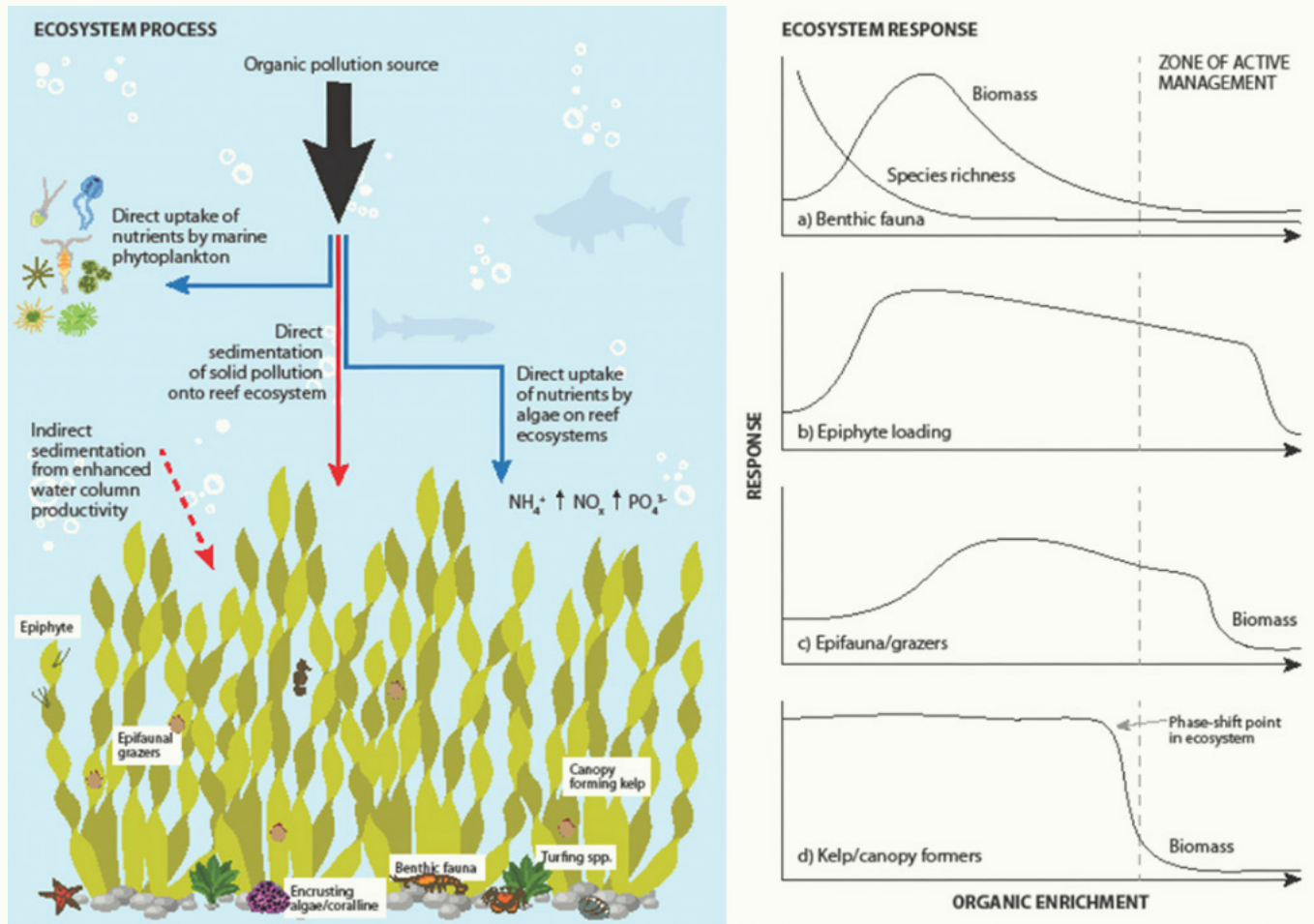


3.2 Inshore Reef

“Inshore reefs” refer to the hard substrate communities in the shallow subtidal area that are generally dominated by macroalgae. As the finfish aquaculture industry has intensified and expanded, there is an increased potential for interactions with inshore rocky reefs resulting in a need to develop effective methods to identify potential impacts from the industry (EPA Tasmania 2020, Ross et al. 2021). In Tasmania, dedicated research over the last decade has made significant advances in inshore reef monitoring methods (Ross et al. 2021, White & Brasier 2021, White et al. 2021). Outputs from FRDC 2015/024 (Managing ecosystem interactions across differing environments: building flexibility and risk assurance into environmental management strategies) and FRDC 2018/131 (Storm Bay Observing System: Assessing the Performance of Aquaculture Development) have resulted in new methods for monitoring inshore reefs specifically designed to detect signs of nutrient enrichment, particularly from finfish aquaculture (Ross et al. 2021, Strain et al. 2023). In addition, the Edgar-Barrett reef biodiversity method provides a comprehensive and standardised underwater visual census method for monitoring inshore reef biodiversity that can also be applied to monitoring effects from industry (e.g. Edgar & Barrett 1997, Edgar & Barrett 1999).

In Tasmania, direct sedimentation of farm waste onto rocky reefs is unlikely to occur as there are no finfish farms located directly over rocky substrate. The main interactions between finfish farms and inshore reefs in Tasmania are therefore likely to occur through both direct and indirect processes occurring in the water column (Ross et al. 2021, White et al. 2025). For instance, organic enrichment can occur directly from dissolved and particulate inputs (e.g. urea, uneaten feed or faeces), which increase the dissolved nitrogen in the water column and stimulate productivity of macroalgae on reefs. Indirect pathways include the stimulation of primary productivity in the water column (i.e. farm nitrogen is taken up by marine phytoplankton), which in turn can cause increased sedimentation and/or nutrient supply to inshore reefs (Ross et al. 2021, White et al. 2025). In dynamic environments, deposited sediment may also be resuspended and transported in the water column resulting in increased water column turbidity and sediment deposition on reefs. Both direct and indirect pathways are unlikely to operate in isolation and have the potential to result in complex and dynamic ecosystem responses (Figure 2).

Figure 2: Summary of the interaction pathways of organic pollution on inshore rocky reefs (from White et al. 2025)



The process pane outlines the direct and indirect pathways of organic enrichment of temperate reef ecosystems. The response pane highlights the complexity of response in a multi-level ecosystem:

- a)** an adaptation of the standard Pearson and Rosenberg (1978) model for organic enrichment for benthic infauna;
- b-d)** represent the hypothetical responses of different functional groups in a three-dimensional/ multi-trophic temperate reef system.

Research from overseas has shown that when farms are located directly above inshore reefs, sedimentation from fish farm waste (e.g. uneaten feed, fish faeces and biofouling) can also act as a food source for secondary consumers with the potential to attract aggregations of scavengers that may impact rocky reefs (Fernandez-Jover et al. 2011, Sanz-Lazaro et al. 2011, White et al. 2017, White et al. 2018, Woodcock et al. 2018).

Unlike soft sediment environments, which display largely progressive degradation with increasing impacts, inshore reefs appear to maintain their overall integrity until ‘tipping points’ are met, after which major canopy loss can occur (Connell et al. 2008, Teagle et al. 2017, White et al. 2025). This can result in phase shifts from canopy dominated systems to turfing ecosystems (Connell et al. 2008, Connell & Ghedini 2015). Recovery to canopy dominated systems is unlikely where environmental pressures are sustained, and where it is possible, may only occur over long timescales (Connell & Ghedini 2015).

While canopy loss on reefs has been observed in Australia and globally, to date these have not been associated with finfish aquaculture (Connell et al. 2008). In Tasmania, low levels of sustained organic enrichment can lead to increases in opportunistic (‘nuisance’) species productivity (e.g. epiphytic and filamentous algae) on inshore reefs, with the canopy remaining intact (Oh et al. 2015, Ross et al. 2021). With an expanding finfish aquaculture industry, the potential for interactions with inshore reefs is increasing, particularly in association with other anthropogenic pressures (e.g. climate change and urbanisation) (Ross et al. 2021, White et al. 2025). It is therefore essential to undertake effective monitoring programs that not only monitor reef biodiversity, but also provide information on reef resilience to support sustainable management practices and ensure that inshore reefs do not exceed their ‘tipping points’ (Ross et al. 2021, White et al. 2025).

Methods developed and tested in Tasmania for monitoring effects of finfish aquaculture on inshore reefs include:

- » Biodiversity surveys (full visual census of reef species, abundance and diversity)
- » Targeted assessments (assessment of main functional groups of macroalgae)
- » Remote assessment (assessment of spatial extent of enrichment effects)

3.2.1 Biodiversity surveys

Biodiversity surveys developed by Edgar and Barrett (1997) and the Reef Life Survey (Edgar & Stuart-Smith 2014) are widely used across Australia and globally to collect high quality biodiversity data on temperate reef ecosystems (Ross et al. 2021). They involve a full visual census of all fish, invertebrate and algal species and their associated abundances. These standardised surveys are an important tool in providing understanding of ecosystem structure and trophic interactions in rocky reefs where there is the potential for effects of organic enrichment (Ross et al. 2021, White et al. 2025).

Biodiversity surveys can be used to undertake baseline surveys of inshore rocky reefs to help understand the way in which they may respond to environmental pressures and can be used as a benchmark from which to assess future change (Ross et al. 2021). Biodiversity surveys can identify changes in population numbers, size structure and ecosystem effects resulting from both natural and anthropogenic change. They can also capture data across the entire ecosystem and be used to assess unanticipated future change or impact (Ross et al. 2021).

Inshore reefs are dynamic ecosystems over time and space, with the assemblage of species often site-specific and driven by a wide range of environmental factors (Ross et al. 2021). In Tasmania, inshore reefs are typically characterised by a dominant canopy forming algae species with a diversity of subcanopy algal species, fish and invertebrates (Ross et al. 2021). In southeast Tasmania, the dominant canopy algae varies from site to site. Along the east coast of North Bruny and the D'Entrecasteaux Channel, the canopy consists of either *Phyllospora comosa* (crayweed), *Ecklonia radiata* (common kelp), or *Durvillaea potatorum* (bull kelp), while at the Acteons, *Macrocystis pyrifera* (giant kelp) dominates. In the southeast of Tasmania, fish assemblages on inshore reefs are generally characterised by large numbers of wrasse (Labridae) and leatherjackets (Monocanthidae), while *Heliotis rubra* (black-lip abalone) and purple urchins (*Heliocidaris erythrogramma*) are common invertebrate species.

Species richness is seen to vary from site to site and may be influenced by a number of locally variable environmental conditions including site exposure, nutrient availability, depth, light availability, sea surface temperature, rugosity and the influence of different water masses (Ross et al. 2021). Therefore, to understand local dynamics and to effectively inform management decisions it is important to undertake site specific biodiversity surveys (Ross et al. 2021).

3.2.2 Targeted assessment

Targeted assessments aim to detect a loss of ecosystem resilience due to organic enrichment, before widespread loss of canopy can occur (Valentine et al. 2016). In Tasmania, a rapid visual assessment (RVA) technique for monitoring the functional response of inshore rocky reefs to nutrient enrichment was developed to complement biodiversity surveys (Ross et al. 2021).

This technique has been designed specifically to provide targeted information on functional change and loss of resilience due to organic enrichment (Strain et al. 2020, Ross et al. 2021). Due to its design, the RVA can provide information on reef health more rapidly than Edgar-Barret biodiversity surveys, making it suitable for ongoing monitoring. As the parameters monitored through RVA are simplified and targeted compared to biodiversity surveys, shifts in reef function caused by nutrient enrichment are easily detected using this method.

RVA surveys are intended to be undertaken biannually to improve the ability to identify change in reef function/health. Increasing the number of observations within a long time series allows for better distinction between short term change (e.g. in response to pulses of nutrients into coastal waters) and more chronic, long-term change often associated with anthropogenic pressures (Ross et al. 2021).

Of the 15 parameters assessed through RVA, ten measure broad structural elements associated with reef function while five are indicative of enrichment responses (Ross et al. 2021, White et al. 2021). The broad functional parameters assessed include percentage total canopy cover, sub-canopy brown, green and red algae cover, turfing algae cover, pink and red encrusting algae cover, sponge cover, levels of encrusting fauna, and numbers of the dominant mobile invertebrates. Enrichment parameters include percentage cover of epiphytic and filamentous algae, cover of opportunistic ('nuisance') green and red algal species, together with the level of 'dust' (sedimentation or flocculant material) covering the algae (White et al. 2022). For more information on how these functional parameters are likely to be affected by organic enrichment and associated pressures, see Table 7. The parameters chosen are relatively easy to observe and are assessed against defined criteria to improve consistency between surveys (see Table 11 in Section 4.2.2).

Table 7: Common responses associated with organic enrichment and sedimentation observed in the functional parameters assessed within the RVA for inshore reefs (Ross et al. 2021)

Functional parameter	Likely response to organic enrichment/sedimentation
Canopy cover	Decline
Sub-canopy cover (red, brown, green macroalgae)	Depending on severity of enrichment, may see decline in brown sub-canopy and an increase in green sub-canopy.
Epiphytic algae	Increase in epiphytic algae
Filamentous algae	Increase in filamentous algae
Opportunistic algae	Increases in opportunistic green and red algae
Turfing algae	Likely to see an increase in turfing species
Encrusting fauna	Likely to see an increase with increasing sedimentation
‘Dust’ on algae	Likely to see an increase with increasing sedimentation

In Tasmania, canopy and subcanopy cover have been found to be largely stable at sites monitored using RVA, although observations of epiphytic, filamentous and opportunistic algae indicates evidence of organic enrichment in some locations (Ross et al. 2021, White & Brasier 2021, White et al. 2021). In the southeast of Tasmania, the opportunistic algae species include *Ulva* (green), *Cladophora* (green), *Chaetomorpha billardieri* (green) and *Asparagopsis armata* (red).

Stability in canopy and sub-canopy species is indicative that rocky reefs in Tasmania remain relatively resilient to temporary episodes of organic enrichment and associated increases in fast-growing algae species (i.e. epiphytic, filamentous and opportunistic species) (Ross et al. 2021). While organic enrichment on Tasmanian inshore reefs has largely been observed as a temporary condition, indicators of sustained enrichment have also been observed (Ross et al. 2021, White et al. 2021). Further research is needed to better understand the effects of sustained organic enrichment on Tasmanian inshore reef resilience (Ross et al. 2021).

Remote assessments

Remotely operated vehicles (ROVs), autonomous underwater vehicles (AUVs) or towed camera systems offer robust survey tools for quantifying temperate reef habitat in a way that is not constrained by diving limitations such as depth or bottom time (Sward et al. 2019, Sward et al. 2022). Although taxonomic resolution and the capacity for consistent species identification is reduced compared to diver-based surveys (Guinda et al. 2014), they can dramatically increase the spatial area that can be covered in a survey (Parravicini et al. 2009, Guinda et al. 2014).

Remote assessments have been developed alongside RVAs as a means of determining whether evidence of functional change observed by RVA surveys is a localised or widespread phenomenon. This method of assessment has been designed using towed camera, drop camera, ROV or AUV to quickly survey a larger spatial area of rocky reef than is possible if relying solely on divers. Analysis of subsequent images can be done using one of several different methods, depending on the research question of interest.

These methods include:

- » Opportunistic algae and/or canopy assessments
- » Presence/absence assessments
- » Algal biodiversity and/or ecological assessments

Where significant changes are observed, a more detailed and comprehensive biodiversity survey can be undertaken to determine the severity of reef impacts and to assess biodiversity change compared to baseline conditions. Technological advances are expected to further enhance the capability of remote assessment platforms for temperate reef monitoring into the future.

3.3 Deep reefs

In the context of salmon farming in Tasmania, deep reefs refer to hard substrate habitat that extends from the deeper subtidal through to the upper-mesophotic (low light) zone. As a general rule, canopy-forming macroalgae are absent from these systems, with green and red algal communities dominating in the upper depths and sponges dominant at deeper depths where light levels no longer support the growth of foliose algae. As the industry plans to move further offshore and into more exposed environments, interactions may occur with these systems for the first time. Storm Bay is home to an extensive deep reef system and the consequences of interactions with the salmon industry are, at this stage, largely unknown. Consequently, new monitoring requirements have been included in environmental licence conditions (EPA Tasmania 2020) and methods for monitoring interactions between finfish aquaculture and deep reefs have been recently developed and implemented (Strain et al. 2020).

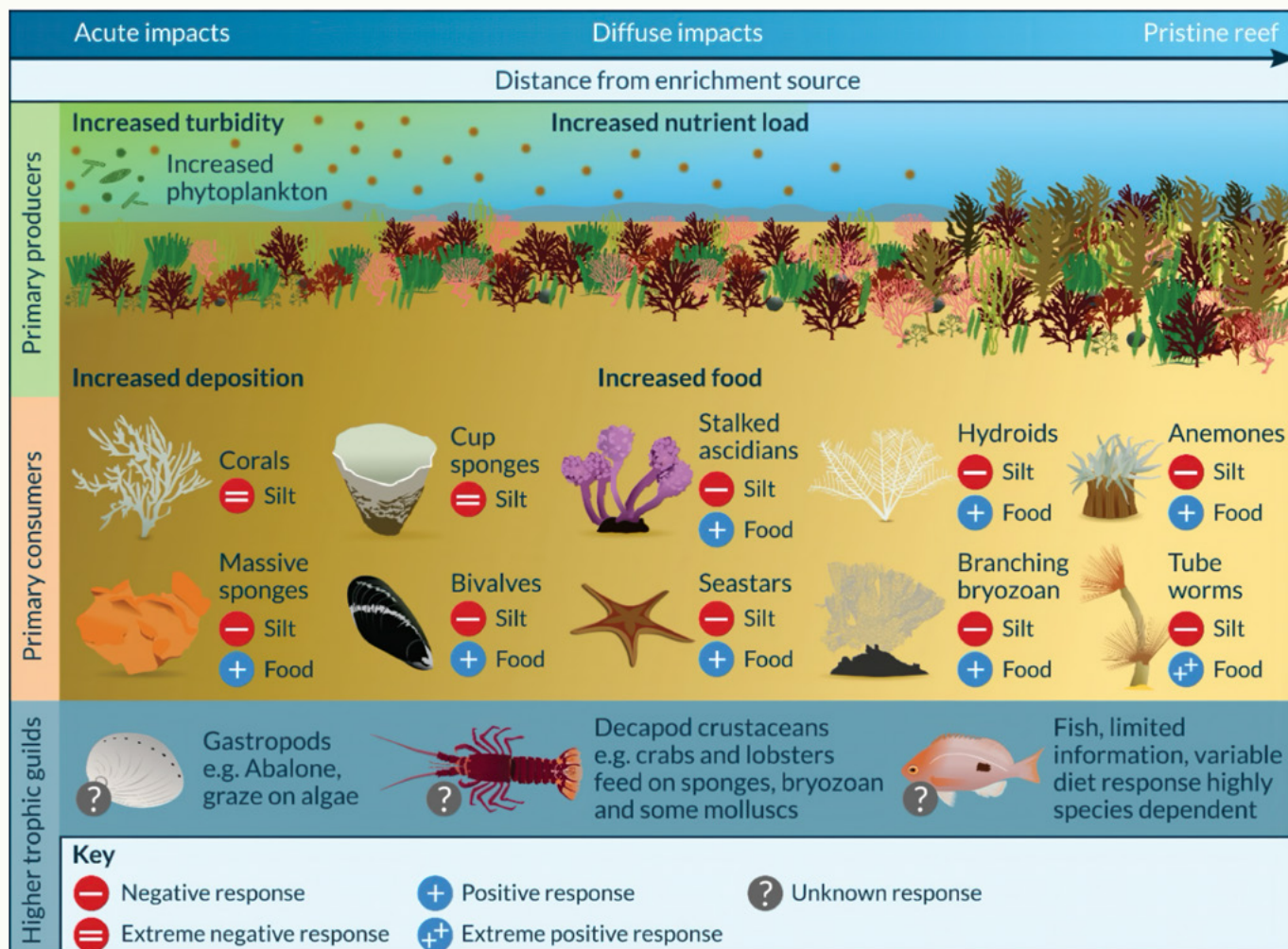
In Tasmania, no farm is located directly over hard substrate and therefore any observed impact on deep reefs is likely to be diffuse rather than direct. As with inshore reefs, enrichment can occur directly from dissolved and particulate farm waste (i.e. uneaten feed, fish faeces, urea and biofouling discharge) or indirectly by stimulating primary productivity (planktonic and epiphytic algae), which can then result in increased nutrient supply to reef environments as it decays (Ross et al. 2021).

The way in which different taxa in deep reef environments respond to increased nutrient and organic matter availability depends on the degree of

enrichment and their physiology, with some species likely to be negatively affected while others may experience a net benefit (Figure 3) (Strain et al. 2020). As light levels are lower in the deeper waters, many macroalgae species (e.g. larger brown algal species) exist on the threshold of their environmental limits. This can cause a complex dynamic between light, nutrient availability and the interplay between these factors under various organic enrichment scenarios.

For example, increased nutrient availability may favour growth of some green algae species (e.g. *Caulerpa* in southeast Tasmania), various red algae species and turfing species (Strain et al. 2020). Longer term, these species may gain the competitive advantage over brown algae, resulting in a change to species assemblage (Connell et al. 2008). Similarly, increases in phytoplankton in the water column as a result of organic enrichment can reduce light availability to deep reefs, which can affect the growth and survival of algae and phototrophic sponges (Strain et al. 2020). It may also result in increased sedimentation on deep reefs (Figure 3) (Strain et al. 2020). Low levels of increased sediment deposition may stimulate productivity in some species (e.g. erect and massive sponges, mussels and ascidians); however, high levels of deposition are likely to result in negative effects on biota sensitive to smothering, such as corals or cup sponges (Strain et al. 2020). Given the complexity and the lack of knowledge regarding system response, monitoring programs need to have the capacity to capture inherent variability and unforeseen change within these systems.

Figure 3: Potential impact pathways from organic enrichment on deep reef ecosystems in Storm Bay, Tasmania (Strain et al. 2020).



Methods used for monitoring interactions between finfish aquaculture and deep reefs in Tasmania include:

- » Deep reef quantitative biodiversity assessment
- » Deep reef qualitative assessment

Note that these methods are still under development, with extensive testing and understanding of limitations yet to occur with regard to monitoring for effects of finfish aquaculture.

Ongoing monitoring of deep reef systems in Tasmania will increase our understanding of the interactions between finfish farms and deep reefs and can be used to inform future survey designs here and elsewhere. While methods are presented for monitoring deep reefs within this field guide (Sections 3.3.1, 3.3.2 and 4.3), they have not been extensively tested for this purpose and may be subject to change as new research becomes available.

3.3.1 Deep reef quantitative biodiversity assessment

Biodiversity surveys have long been used in monitoring inshore rocky reefs where SCUBA divers are able to collect data in situ (see Section 3.2.1). For deeper systems that are inaccessible by diver, biodiversity surveys have only been made achievable more recently by advances in digital imaging technologies that enable high quality photographic and video footage to be collected and analysed (Strain et al. 2020).

Biodiversity surveys in deeper waters will generate a comprehensive dataset of all flora and fauna present in these systems, providing information on species assemblage, trophic interactions and potential interactions with finfish aquaculture or other anthropogenic impacts (e.g. climate change). Parameters monitored to characterise deep reef biodiversity include identification and abundance of fish (including fish length), sessile invertebrates (e.g. sponges, bryozoan and zoanthids), and algae and substrate, from which percentage cover is estimated.

Techniques for monitoring deep reef systems have been developed through the National Environmental Science Program (NESP) Hub, with quantitative biodiversity surveys to assess interactions with finfish aquaculture largely adopting these methods (Monk et al. 2020, Barrett & Monk 2021, Perkins et al. 2022). Quantitative biodiversity assessments for deep reefs rely on the ability to capture high quality, quantifiable, georeferenced footage using both forward-facing and downward-facing camera systems (Strain et al. 2023). Limitations associated with undertaking visual census using digital imaging technologies include difficulty in taxonomic identification to species level and observing cryptic species (e.g. mobile invertebrates). The development of a standardised method for identifying and classifying morphospecies has facilitated the collection of high-quality data using repeatable measures (Althaus et al. 2015), although true species estimates may be different to that indicated by morphology alone.

Results from FRDC 2018/131 (Storm Bay Observing System: Assessing the Performance of Aquaculture Development) indicate fish assemblages are dominated by schooling planktivorous fish such as butterfly and barber perch, with diverse leatherjacket and wrasse communities often associated with the shallower sites where macroalgae dominated benthic assemblages (Strain et al. 2023). The biodiversity observed in the benthic communities was very high, even compared to neighbouring marine parks, with a large array of depth strata covered across the survey area (Monk et al. 2016, Perkins et al. 2021). An amazing diversity of sponges was recorded, along with encrusting coralline algae, zoanthids, bryozoans, sea whips, gorgonian corals, stalked ascidians and feather stars (Strain et al. 2023). Canopy forming brown algae was present only in shallower depth strata, with understorey brown algae rarely found in deeper waters.

While the surveys undertaken through FRDC 2018/131 (Storm Bay Observing System: Assessing the Performance of Aquaculture Development) increased our understanding of deep reef systems, limitations to the tested approaches were also highlighted. For example, while sampling effort was adequate to capture fish biodiversity, analysis of data suggested that sampling effort was insufficient to capture the full diversity of the benthic community. There are also several approaches involving key species or functional groups that are worth investigating into the future (Bastiaansen 2020, Strain et al. 2023). Like inshore reefs, deep reefs show variability in benthic assemblages, with depth, substrate, rugosity, exposure and slope all likely to influence the biota observed in any given location (Barrett et al. 2001, Aquenal Pty Ltd 2019, IMAS 2020). Further research of deep reefs will help improve our understanding of their variability and change over time and space, both as a result of natural dynamics and finfish aquaculture interactions.



3.3.2 Deep reef qualitative assessment

While qualitative assessments of deep reefs rely on the same digital imaging footage as quantitative assessments, they consist of a simpler and quicker assessment. The methodology records the presence/absence of all major taxonomic groups, and categorical abundance estimates of fish and mobile invertebrates, visibility, depth and substrate type. Results from FRDC 2018/131 (Storm Bay Observing System: Assessing the Performance of Aquaculture Development) indicate that this method of scoring is only reliable for detecting a very large ecosystem shift (i.e. catastrophic change) (Strain et al. 2023).

Further research is recommended to support the development of a simple, yet robust method for deep reef monitoring. This method could focus on a more functional analysis similar to RVA surveys conducted on shallow reef systems, which focuses on known enrichment pathways and key indicator species (White et al. 2021).

3.4 Seagrass beds

The monitoring of seagrass beds has only recently become a requirement of Environmental Licences issued to marine farming operations in Tasmania (EPA Tasmania 2020) as a response to increasing community concern, particularly around the proliferation of nuisance algae in these systems. As such, research on seagrass beds in Tasmania is growing (Strain et al. 2020).

Pathways of interaction between seagrass beds and salmon aquaculture are relatively simple, with light limitation being the major factor inhibiting growth (Burkholder et al. 2007, O'Brien et al. 2018, Lapointe et al. 2020). This can occur directly by shading from aquaculture infrastructure (Strain et al. 2020) and increased turbidity (Lapointe et al. 2020), or indirectly as a result of stimulation of epiphytic and macroalgal growth (Burkholder et al. 2007) or phytoplankton blooms (Burkholder et al. 2007, Lapointe et al. 2020) in response to increased nutrients.

In addition, where sediments become anoxic and enriched in sulphide, seagrasses can experience sulphide invasion, increasing mortality and reducing seabed resilience (Frederiksen et al. 2007).

While interactions between finfish aquaculture and seagrass beds are evident, most research has been undertaken on the Mediterranean seagrass *Posidonia oceanica*, a slow growing species (Apostolaki et

al. 2007, Frederiksen et al. 2007, Diaz-Almela et al. 2008, Pérez et al. 2008, Rountos et al. 2012). Different seagrass species have different physiologies and therefore the potential to respond to nutrient enrichment and sedimentation in different ways (Strain et al. 2020). Little is known about the response of Tasmanian seagrass species to nutrient enrichment from finfish farms. In the finfish aquaculture farming areas of southeast Tasmania, the *Zostera tasmanica* complex is the only group of seagrass species present. The *Zostera tasmanica* complex shows higher growth rates in leaves, roots and rhizomes and produces seeds in much higher densities than *Posidonia* spp., resulting in a higher natural turnover of plants (Waycott et al. 2014, Sherman et al. 2018). However, *Amphibolis antarctica* is the dominant species in north-west Tasmania and is a slow growing species with lower tissue turnover, more similar to *Posidonia oceanica*. Given differences in ecology, the resilience of *Zostera tasmanica* to nutrient enrichment from finfish aquaculture is likely to be quite different to that observed in *Posidonia* spp. in the Mediterranean, or *Amphibolis antarctica* in north-west Tasmania (Strain et al. 2020). Therefore, the species of seagrass in the location of interest, along with the implied resilience of the ecosystem must be taken into consideration when determining a monitoring program.

A number of methods for monitoring seagrass were trialled on Zosteraceae beds through FRDC 2018/131 (Storm Bay Observing System: Assessing the Performance of Aquaculture Development). From this, potential methods which can be employed for monitoring effects on seagrass beds include:

- » Seagrass bed extent mapping
- » Monitoring seagrass health/ecological status

3.4.1 Seagrass bed extent mapping

The spatial extent of seagrass beds can be highly dynamic over time, with large shifts in both seagrass bed size and distribution (Ball et al. 2014). To understand any potential responses to anthropogenic pressures, it is necessary to understand the spatial variability of seagrass ecosystems in the area of interest. Studies from Port Philip Bay, Victoria have shown that seagrass beds can expand and contract significantly over time and space (Ball et al. 2014). Maintaining an awareness of the spatial extent is therefore a critical aspect of monitoring seagrass and must be carried out at a regional scale to ensure comprehensive mapping of existing seagrass beds.

In a Tasmanian context, it is important to establish an understanding of the dynamics of seagrass beds over time in any areas with potential for interactions with finfish aquaculture. By understanding seagrass extent, it may be possible to identify any potential reductions in seagrass extent due to nutrient enrichment and/or sedimentation prior to widespread ecosystem decline. As mentioned above, the stability of seagrass beds is likely to be strongly linked to the dominant species. For example, given the higher regenerative capacity of *Zostera tasmanica*, beds dominated by this species are likely to be much more dynamic than those dominated by *Amphibolis antarctica*.

3.4.2 Seagrass health

In addition to monitoring the extent of seagrass beds, it is also important to monitor their health or ecological status. Monitoring for signs of stress on the ecosystem may help to identify areas affected by nutrient enrichment and/or other interactions with finfish aquaculture prior to any significant shifts in ecosystem function (e.g. significant decline in seagrass beds).

Due to ecological differences between seagrass species (e.g. leaf and rhizome growth rates), the response of different species to the same environmental pressures may vary.

As such, it is important to build a picture of species-specific response pathways to interactions with finfish aquaculture. This is foundational in understanding ecosystem resilience and building effective long-term monitoring and management systems.

In Tasmania three key methods for monitoring seagrass health have been developed. All three methods focus on different aspects of environmental assessment and are intended to be used in association with each other to target different research questions as necessary:

- 1 Ecological assessment of seagrass bed & associated biota:** a comprehensive census of seagrass ecosystems that can be used when a full understanding of seagrass bed dynamics is needed (e.g. baseline surveys). These surveys incorporate information on fish abundance (and size), mobile invertebrates, seagrass bed composition, seagrass condition, benthic infauna and epifauna, and sediment chemistry.
- 2 Quantitative bed condition assessment:** a simpler survey focused on providing measurable information on spatial and temporal change in seagrass beds at specific sites to be undertaken as part of ongoing monitoring programs. These surveys assess percentage cover of seagrass, epiphytic algae on seagrass, *Caulerpa* spp., wrack algae (brown, green and red) and other algae (brown, green and red), and information on substrate and fauna to assess bed condition using photoquadrats (see Section 4.4.2 for more detail).
- 3 Qualitative bed condition assessment:** a survey methodology designed to assess the broadscale spatial extent of impacts (where they have been observed in quantitative bed condition assessments). These surveys use video footage to assess epiphytic algae cover, which is used as a proxy for overall seagrass bed health.

For more information on the key parameters used in monitoring seagrass health and how they relate to potential impact, see Table 8.

Table 8: Key parameters used to assess seagrass health

Parameter	Relevance to impact
Seagrass cover (e.g. <i>Zostera</i> spp., in southeast Tasmania)	A measure of seagrass bed density where patchiness and thinning are indicative of a bed decline.
<i>Caulerpa</i> spp. cover	<i>Caulerpa</i> spp. are a group of green seaweeds which can replace seagrass when it gains the competitive advantage (typically due to nutrient enrichment). Therefore, changes in <i>Caulerpa</i> cover over time can be used as a metric for nutrient enrichment.
Epiphytic algae (inc. filamentous) cover and species present	Epiphytic algae (including epiphytic filamentous algae) are a group of algae which grow on other plants or objects within the marine environment. Epiphytic algae grow on seagrass, particularly where nutrient availability is high, reducing seagrass resilience by limiting light availability. As such, changes in epiphytic algae growth in seagrass beds can provide information on seagrass resilience and nutrient enrichment.
Presence of macroalgae (brown, green and red inc. nutrient enrichment indicator species)	The presence of other macroalgae species may relate to seagrass bed resilience, where higher proportions of algae species can indicate a shift in seagrass bed composition. The presence of some species is indicative of nutrient enrichment (e.g. <i>Chaetomorpha billardierii</i> , <i>Chaetomorpha coliformis</i> , <i>Ulva</i> sp., and <i>Enteromorpha</i> sp. in southeast Tasmania).
Presence or number of any major invertebrates, infauna and epifauna	Used to provide information on the overall health and functioning of the seagrass ecosystem.
Fish abundance and size	Used to provide information on the overall health and functioning of the seagrass ecosystem.
Sediment chemistry	Used to provide information on the overall health and functioning of the seagrass ecosystem.

4 Recommended techniques

There are a wide variety of techniques that can be employed to provide information on the state of the marine environment, and more specifically relating to effects of finfish aquaculture.

These vary in their sensitivity and complexity, and while some can provide definitive information on the health of the environment, others are used in combination to develop an understanding of the environmental context within which to interpret other monitoring data.

The types of techniques employed vary markedly depending on the receiving habitat of interest and while all monitoring techniques will have specific requirements, there are a number of important considerations which are generally applicable to all:

- » It is always important to ensure that particular attention is given to maintaining comprehensive metadata including site locations, site names, sampling dates (and times where relevant) in addition to any relevant sampling notes/ observations that may affect analysis results and interpretation, as well as any sampling specific information.
- » It is essential that a consistent and thorough sample labelling system is adopted.

- » A suitable storage facility for both data (e.g. database) and physical samples is necessary so that information is not lost as it may need to be revisited sometime after the sampling event.
- » Positioning equipment is necessary to identify and/or record sampling locations using a standard datum (e.g. Geographic Datum of Australia (or GDA) 1994 or 2020) and while any form of Global Positioning System (GPS) can be used, differential GPS units generally provide greater accuracy than standard units.
- » Risk assessments should be undertaken for all field work, following all relevant occupational health and safety regulations/guidelines.
- » Vessel requirements are likely to vary between monitoring techniques, with consideration given to the availability of deck space, storage facilities and lifting facilities when considering a vessel's suitability for a given technique.

In addition, when considering undertaking a monitoring program, it is important to ensure that the chosen survey design and methods will provide suitable statistical power to answer the intended research questions (Ross et al. 2021).

4.1 Soft sediment sampling and analyses techniques

4.1.1 Benthic macrofauna sampling and analysis

Sampling soft sediment for benthic macrofauna can be undertaken using grabs deployed from a boat, or by diver using a handheld sediment corer. While there are a wide variety of grabs available on the market, for the benthic conditions in Tasmania, Van Veen grabs have been found to be the most successful (Figure 4a).

To ensure a robust sampling design, triplicate samples of similar sediment volume should be taken at each site. Where samples are taken by diver, a wide diameter sediment corer (Figure 4b) should be used (150 mm diameter x 100 mm depth).

Figure 4:



a) Van Veen grab in open position



b) diver operated sediment corer

After collection, benthic samples should be sieved through a 1 mm sieve using a low-pressure seawater supply to ensure that fauna are not forced through the mesh. The material retained in the sieve should be transferred to plastic or glass storage containers and completely covered with a solution of 10% formalin (4% formaldehyde) buffered in seawater (i.e. 100 mL formalin to 900 mL seawater). Samples must remain in the 10% formalin solution for a minimum of 48 hours to ensure complete fixation.

After fixation, all samples must be rinsed thoroughly in fresh water and then transferred to shallow, clear trays for sorting under a stereo microscope. During sorting, all fauna present in the samples are removed and stored in labelled storage jars in a solution of 70% ethanol. All species present should then be identified to the lowest possible taxonomic level and each taxon counted. Appendix A provides illustrations of a number of key benthic indicator organisms for reference. Full identification of the faunal species requires highly skilled and well-trained biologists/taxonomists and therefore further detail is not provided within this guide.

It is important that all samples are processed separately and identically. To ensure that samples can be revisited in the future where confirmation of identification is necessary, they must be stored for a minimum of 4 years in storage jars in a solution of 70% ethanol and 1-2% glycerol. Storage jars should be labelled clearly both inside (using waterproof paper annotated in pencil) and outside the jar with information on the date of collection, site location, collection method and collectors' and identifiers' names.

4.1.2 Sediment core sampling

In monitoring for effects from finfish farms in the marine environment, sediment core samples are typically collected for analysis of redox potential, sulphide concentration, carbon and nitrogen (including stable carbon and nitrogen isotope signatures), particle size analysis, metals analysis and environmental DNA (eDNA) as needed.

The same cores can be used for analysis of redox, sulphide, carbon and nitrogen and stable isotopes and particle size analysis. It should be noted that for metals analysis and eDNA analysis separate cores should be taken as analyses require sampling of the top 3 cm and 5 cm of undisturbed sediment respectively. For metals analysis, it is important that on sediment core retrieval, the top 3 cm of sediment is immediately transferred to an acid-washed glass jar and stored at a constant, cool temperature, whilst any remaining sampling/transit is conducted for the survey.

There are a variety of sediment corers available on the market that can be deployed from boats. Corers that are routinely used in Tasmania include the Craib corer, multicorer and HAPs corer (Figure 5). In addition, cores can be collected by divers where necessary.

Figure 5:**a) Craib corer****b) multicorer****c) HAPs corer used to collect sediment samples**

To obtain enough sediment for the necessary analysis, cores should be taken using a clear Perspex corer with an internal diameter of at least 50 mm and a length of approximately 250 mm (Figure 6). Furthermore, for cores intended for measurement and analysis of redox and sulphide, cores need to contain side access sample ports (which can be sealed with duct tape during deployment).

Figure 6: Perspex corer with an internal diameter of at least 50 mm

In situations where cores are collected by divers, the core should be inserted into the sediment to a depth of approximately 15 cm. A bung is then inserted into the top of the core which produces a vacuum within the core to hold the sediment in position while it is then carefully removed from the sediment. A second bung is then inserted into the bottom of the core to contain the complete core. While this method has been found to be largely successful in fine sediments, it is less effective in sandy conditions and in these circumstances, it may be necessary for the diver to dig around the core to insert the second bung prior to removal from the sediment.

After core retrieval, a written description of each core should be made including the length of the core (in millimetres), sediment profile colour changes using a Munsell soil chart, presence of any visible animal or plant life, presence (size and position) of gas vesicles in the sediment, any sediment smell (i.e. indicating the presence/absence of hydrogen sulphide).

To ensure that cores are kept intact it is important to keep them upright during transit and storage. Use of a simple carrying box is recommended to protect the cores until sampling for analyses is complete. In situations where measurements/analyses are not being undertaken immediately, cores should be placed in a suitable container (e.g. an esky) and filled with water at the ambient seawater temperature to minimise alteration to the sediment prior to analyses.

4.1.3 Sediment chemistry analyses

Using sediment samples collected by sediment core, the following analyses can be carried out to provide information on the health and enrichment status of benthic sediment.

Redox

Measurement of redox potential should be taken from each sediment core (see Section 4.1.2 above) at a depth of 3 cm using a redox probe. There are a variety of redox probes available for use; combination electrodes are recommended as they are easier to use in soft sediment samples.

It is important to calibrate redox probes before each set of measurements is taken. It is possible to buy pre-packaged calibration solutions. Redox probe calibration is sensitive to temperature; therefore it is important to note the temperature of the calibration solution and the sample at the time that each core is measured. It is best if both temperatures are comparable.

To measure redox, the redox probe should be inserted into one of the side access sample ports on the core tube. Redox probes measure the oxidation/reduction potential of the sediments by determining the availability of free hydroxyl ions. The act of undertaking the measurement will affect this, resulting in the reading on the meter continuing to decline over time as the measurement is being undertaken. The point at which a redox reading should be taken from the probe is when the rate of decline has stabilised. This may happen quickly, or it may take a few minutes depending on the sediment conditions. An error margin of ± 10 -20 mV in redox readings is considered to be acceptable. Redox results then need to be corrected to a standard hydrogen reference electrode using the relevant correction factor specific to the redox probe. Corrected redox results and raw data for each core can then be reported in reported in millivolts (mV) for analysis.

Sulphide concentration

There are several different probes available for the measurement of sulphide concentration, but a silver/sulphide combination electrode is recommended. It is important to follow all manufacturer's guidelines for use, sensitivity and calibration as these may vary between manufacturers. Before carrying out each set of measurements, calibration must be undertaken by establishing standard calibration curves, following preparation of a Sulphide Antioxidant Buffer (SAOB).

SAOB can either be purchased or it can be prepared by adding 20.00 g of Sodium Hydroxide pellets (NaOH) and 17.90 g of Ethylenediaminetetra-acetic acid (or EDTA) in a volumetric flask, diluted with distilled water to a volume of 250 mL. SAOB solution must be refrigerated until required.

Immediately before use in calibration, 8.75 g of ascorbic acid must be added for every 250 mL of SAOB solution required. After the addition of ascorbic acid, the solution will only be viable for 3 hours.

Measurement of sulphide should be taken from each sediment core (see Section 4.1.2 above) at a depth of 3 cm using a sulphide probe. To do this, a 2 mL sediment sub-sample is extracted from one of the side access sample ports on the core tube using a 5 mL syringe and then placed in a glass vial. 2 mL of SAOB is then added to each vial. Sulphide concentration is then measured in millivolts (mV) by placing the probe into the vial and slowly stirring the sediment/SAOB solution mixture until the reading stabilises.

Using the calibration curve produced during calibration, sulphide measurements in mV can be converted to sulphide molar concentration (μM).

Organic content and stable isotope analysis

To undertake analyses for organic carbon, nitrogen and stable carbon and nitrogen isotopes the top 3 cm of each sediment core (see Section 4.1.2 above) should be retained. The sediment is then homogenised and can be split into two samples: one of at least 1 g (dry weight) for stable isotope and elemental analysis of organic C and N content (and C:N ratio) and one of at least 5 g (dry weight) for measuring percent organic matter using the loss on ignition (LOI) technique.

For the LOI technique, the sample should be dried at 60 °C for at least 48 hours or until all moisture is removed. It can then be placed in a muffle furnace for 4 hours at 450 °C where it is ignited. The percent weight loss that occurs due to ignition (loss on ignition) is equivalent to the total organic matter (TOM) present within the sample.

For the stable isotope analysis and C and N analysis the sample should be placed in a freeze drier to remove all moisture from the sample. The dried sample should then be ground and split into two separate vials, one each for carbon and nitrogen analyses. Sub-samples should weigh approximately 0.5 g each.

To undertake carbon analysis, one of the sub-samples must be acidified with dilute hydrogen chloride (HCl) solution to dissolve any solid carbonates present in the sample before analysis. Both carbon and nitrogen samples are then analysed using an elemental analyser interfaced to a continuous-flow isotope ratio mass spectrometer.

Stable isotope data must be expressed in delta notation ($\delta^{13}\text{C}$ and $\delta^{15}\text{N}$) relative to their respective standards. For carbon this is the stable isotopic ratio of Vienna Pee Dee Belemnite standard ($R(^{12}\text{C}/^{13}\text{C})_{\text{VPDB}} = 0.0111797$) and for nitrogen it is atmospheric N_2 ($R(^{14}\text{N}/^{15}\text{N})_{\text{Air-N}_2} = 0.0036765$).

Following analysis, all individual stable isotope samples should be retained in appropriate storage for at least 3 years in case samples need to be revisited.

Particle size analysis

To carry out particle size analysis of sediment, a subsample of sediment from the top 100 mm of each core (see Section 4.1.2 above) should be placed in a container of known volume and filled to the top to measure the sample volume.

Each sample can then be gently wet sieved through a sieve stack of 4 mm, 2 mm, 1 mm, 500 μm , 250 μm , 125 μm , 63 μm by hand or using a sieve shaker. The fraction of sediment <63 μm is not used in analysis and is allowed to drain away.

For each retained sieve fraction, the material should be carefully placed in a graduated cylinder. To measure the volume of sediment in each fraction, a known volume of water is added to the cylinder and the volume of sediment is measured as the displaced volume. To ensure consistency, the volume of water should remain constant throughout the analysis. Calculation of the <65 μm fraction is calculated by subtracting the sum of all sieve fractions from the initial sediment volume.

Particle size analysis data should be reported in an Excel spreadsheet and graphed as cumulative percentages.

Alternatively, samples can be provided to an external NATA accredited laboratory for analysis.

Metals

For metals analysis, samples are sent to a NATA accredited laboratory for analysis of As, Cd, Co, Cr, Cu, Mn, Ni, Pb and Zn.

4.1.4 Visual assessment

Soft Sediments

To undertake a visual assessment of soft sediment health, video footage collected using a Remotely Operated Vehicle (ROV) fitted with a minimum of two cameras and coupled with a precise geo-location system (e.g. ultra-short baseline) has been found to be the most suitable method (Figure 7).

Other visual methods for assessing sediment health have also been developed using benthic photographs (see Macleod & Forbes 2004). Further research has not been undertaken on these methods and they are therefore not addressed further in this field guide.

Video should be recorded in colour using a camera with high-definition imagery, 4K resolution and low light capabilities (operational at a minimum of 3 lux), so that footage can be adequately scored against all assessment criteria. A GoPro camera (GoPro Hero 5 or better) can also be used. For best results, an independent light source (preferably LEDs) that can adapt its intensity to site-specific conditions should be used, as lighting requirements may vary between locations. This ensures the necessary quality for identifying assessment features, such as sediment colour.

Figure 7: Photo showing 2 different ROV options



Left: Falcon ROV with two camera housings visible (no cameras present). **Right:** a Boxfish ROV with two cameras. Photo credit: Justin Hulls



To ensure an optimal view of the seafloor and consistency in video footage, the distance between the camera lens and the sediment surface should be maintained between 30 cm and 50 cm. This can be achieved by housing the camera within a frame. A minimum transect of 50 m (approximately 3.5 minutes at 0.5 knots) should be captured by video footage at each monitoring station. A record of the date and time (in coordinated universal time (UTC) format), location, depth and bearing at the start of each filming sequence must be logged.

For the assessment, the video is replayed and a numeric categorisation is assigned to each measured variable based on their presence/absence and density/abundance. A complete list of all the variables measured is provided in Table 9, along with their associated weightings. Variables whose presence is indicative of enriched sediments (e.g. gas bubbles, pest species, opportunistic fauna, pellets and faeces) are given negative weightings, while variables that are indicative of healthier sediment conditions (e.g. faunal tracks and burrows, presence of indicator species) are given positive weightings.

Photographic examples of different criteria used to assess enrichment are provided in Appendix B.

The score for each variable is calculated by multiplying its presence or abundance rating by its weighting. The products for each variable (either positive or negative) are summed to provide an overall site/video score, indicative of specific effect categories (see Table 9 and Table 10 for details). Higher scores reflect less impacted or unimpacted sediment, while lower scores indicate greater impact.

For finfish farm management purposes, calculating site video scores provides sufficient detail to determine the level of impact at each site, from which management practices can be adapted. Where more detailed interpretation is required, site video scores can also be analysed using univariate or multivariate techniques (e.g. using Primer software). Univariate analysis can be used to compare scores over time or space by providing a single score for each site and time. Where specific interest is in a particular assessment variable, multivariate analysis can be used to compare difference between sites with respect to the variable of interest.

Visual assessment scoring criteria

Video feature		Scoring levels				Weighting	Category
		0	1	2	3		
1	Gas bubbles	Absent	On disturbance	Spontaneous outgassing	NA	10	-ve
2	Sediment colour black/grey	Normal	Not normal (comp to ref)	NA	NA	1	-ve
3	Bacterial coverage (<i>Beggiatoa</i> spp.)	Absent	Thin patchy	Thick patchy or thin extensive	thick extensive or streaming	1.5	-ve
4	Pellets/ Faeces	Absent	Few	Many	Feed spill	1	-ve
5	Biofouling debris	Absent	Sparse	Moderate to dense	NA	1.5	-ve
6	Burrows/mounds	Absent	Few	Many	Dense	1.5	+ve
7	Worm tubes/casts	Absent	Present	NA	NA	1	+ve
8	Faunal tracks	Absent	Present	NA	NA	1	+ve
9	Algal cover	Absent	Sparse	Moderate	Dense	1.5	+ve
10	Brittle stars (Ophiuroids)	Absent	Present	NA	NA	1.5	+ve
11	New Zealand screw shell (<i>Moaricolpus roseus</i>)	Absent	Few (<10)	Many (≥10)	NA	1	+ve
12	Dog whelks (<i>Nassarius</i> spp.)	Absent	Few (<10)	Many (≥10)	NA	1	-ve
13	Side-gilled slugs (Pleurobranchidae)	Absent	Few (<3)	Many (≥3)	NA	1	-ve
14	Heart urchins (<i>Echinocardium cordatum</i>)	Absent	Few (<3)	Many (≥3)	NA	1	+ve
15	Squat lobsters (Galatheidae)	Absent	Few (<3)	Many (≥3)	NA	1	-ve
16	Swarming epibenthic crustaceans	Absent	Few (<3)	Many (≥3)	NA	1	+ve
17	Other worms (Annelids) including spoon worms (Echiurans)	Absent	Few (<3)	Many (≥3)	NA	1.5	+ve
18	Dorvilleid polychaete worm (<i>Schistomeringos loveni</i>)	Absent	Few (<30)	Many (≥30)	NA	1.5	-ve
19	Dorvilleid polychaete worm (<i>Ophryotrocha shieldsi</i>)	Absent	Few (<30)	Many (≥30)	NA	1.5	-ve
20	Sabellid polychaete worms	Absent	Few (<10)	Many (≥10)	NA	1	+ve
21	Capitellid polychaete worms (<i>Capitella</i> spp.)	Absent	Few (<10)	Many (≥10)	NA	1.5	-ve
22	Fish	Absent	Few (<3)	Many (≥3)	NA	1	+ve
23	Other crustaceans	Absent	Few (<3)	Many (≥3)	NA	1	+ve
24	Seastars others	Absent	Few (<3)	Many (≥3)	NA	1	+ve
25	Other fauna	Absent	Few (<3)	Many (≥3)	NA	1	+ve
26	Pie crust crab (<i>Metacarcinus novaezelandiae</i>)	Absent	Few (<3)	Many (≥3)	NA	1	-ve
27	Northern Pacific seastar (<i>Asterias amurensis</i>)	Absent	Few (<3)	Many (≥3)	NA	1	-ve



Table 9: Visual assessment scores and associated effect categories for low energy and high energy environments in Tasmania

Impact stage		Degrading				
		I	II	III	IV	
Effect category		No evidence of impact	Minor effects	Moderate effects	Major effects 1	
Video score	Low energy (incl. Macquarie Harbour)	>5	2.5 to 5	<2.5	<0	
	High energy	>2.5	2.5 to 5	<2.5	<0	

4.1.5 eDNA

To undertake eDNA metabarcoding analysis, approximately 5 g of sediment should be sampled from the top 3 cm of each core, using clean gloves and single-use sterile 5 mL syringes to limit contamination (see Section 4.1.2 above). Before sampling and between each sample collection, Perspex sediment core tubes should be soaked in 20% household bleach solution to avoid contamination. After collection, sediment samples should be stored in 10 mL cryovials and immediately placed within a dry shipper charged with liquid nitrogen for transport back to the laboratory. Upon returning to the laboratory, all samples should be stored at -80 °C until DNA extraction.

Before undertaking DNA extraction, samples should be weighed. Some samples may contain excess water which should be removed before weighing. If samples remain very wet after pouring off (e.g. fine sediment samples), they can be dried using centrifugation. This can be undertaken by removing the beads from a PowerBead tube, weighing the wet sample into it and centrifuging it for 30 seconds at 10000 g.

The excess water can then be carefully removed using a micropipette, after which the beads can be placed back in the PowerBead tube.

After weighing each sample, DNA extraction can be conducted using a commercial extraction kit (e.g. DNeasy PowerSoil Pro kit (QIAGEN), following the manufacturer's protocol.

To confirm that DNA extraction has been successful, two quality control procedures should be performed before samples are sent to a suitably accredited laboratory for Illumina paired end sequencing:

- 1 Assess DNA presence to confirm extraction success.** This can be done using a NanoDrop spectrophotometer. NanoDrop results can be used to guide the selection of a subset of samples (approximately 20%), which will undergo polymerase chain reaction (PCR) to check for amplification of the targeted genes (16S rRNA, 18S rRNA or both). Samples with very low (<5 ng/μL) or very high (>40 ng/μL) DNA concentration should be chosen for PCR, together with a random selection of other samples. PCR should be run using primers targeting the gene(s) to be sequenced (16S, 18S or both).
- 2 Check for amplification using gel electrophoresis.** PCR products should be run on a gel to verify the presence of a band at the expected amplicon size. If no band is present, troubleshooting such as sample dilution or DNA clean-up protocols should be performed, or the sample may need to be re-extracted and retested. It is important to keep PCR products away from the area where DNA extractions are being performed to avoid contamination.

			Recovering		
	V	VI	VII	VIII	IX
	Major effects 2	Severe effects	Major effects	Moderate effects	Minor effects
	<0	Conditions not observed; <-10	<0	<2.5	2.5 to 5 (stabilisation of cumulative scores within this range)
	<0	Conditions not observed; <-10	<0	<2.5	2.5 to 5 (stabilisation of cumulative scores within this range)

4.2 Inshore Reefs surveying and analyses techniques

Research into the most effective approaches for monitoring inshore rocky reefs for impacts from finfish farming is ongoing. However, several methods have been developed and scientifically validated through FRDC 2015/024 (Managing ecosystem interactions across differing environments: building flexibility and risk assurance into environmental management strategies), FRDC 2018/131 (Storm Bay Observing System: Assessing the Performance of Aquaculture Development) and subsequent work through the SMRCA. These include targeted diver-based assessments (RVA surveys) and methods using towed camera or ROV to survey a large spatial area. Combined with these, long-established biodiversity survey techniques exist to provide a complete census of marine life at any given site. Different techniques focus on different aspects of environmental assessment and are recommended to be used in association with each other to target different research questions as necessary.

Edgar-Barrett biodiversity surveys (also known as MPA surveys) are well suited for baseline surveys or when a detailed understanding of rocky reef ecosystems is needed. In contrast, the simpler, more targeted RVA surveys focus on specific functional aspects of reef ecosystems to provide information about nutrient enrichment. Techniques that rely on remote platforms for data collection (i.e. ROV or tow camera) are useful for determining the spatial extent of enrichment, particularly in situations where changes have been noted through targeted RVA surveys. Where change has been detected, Edgar-Barrett surveys may be necessary to better understand changes in the system (see Section 3.2 for more background on how these methods can be used to understand the effects of finfish aquaculture).



4.2.1 Biodiversity survey

The Edgar-Barrett biodiversity surveys involve two divers undertaking a comprehensive visual census of inshore rocky reef habitat along a 200 m transect line following the 5 m depth contour. Therefore, to be suitable for this assessment, a reef location must consist of 200 m of continuous reef. A waypoint is marked at the centre of each survey site to ensure consistency across surveys (i.e. the same patch of reef is assessed).

The 200 m transect is subdivided into 4 x 50 m transects as per the University of Tasmania Reef Ecosystem Monitoring Survey Procedures. Along each transect, all fish species are identified, counted and measured within 5 m of both sides of the transect line, with divers swimming parallel along each side of the transect line. Invertebrates and cryptic fish within 1 m of the transect are then identified and counted by a diver swimming next to the transect, while commercially or ecologically important invertebrate species are counted and measured. To assess algae, sessile invertebrates and benthic substrate, 0.25 m² quadrats are placed at 10 m intervals along the transect line (i.e. 5 quadrats per 50 m sub-transect = a total area of 1.25 m²). Percentage cover of the different species within the quadrats is quantified using the point-count method. This involves dividing the quadrat into a grid of 7 x 7 perpendicular wires to create 50 points (including one corner). To quantify cover, the diver undertaking the assessment counts and records the number of times each species occurs directly under the 50 points on the quadrat. Finally, the whole transect line is filmed by a diver using a hand-held camera to retain footage for archival purposes.

For consistency between surveys, it is important to consider the time of year that biodiversity surveys are conducted. It is suggested that biodiversity surveys are undertaken in February/March, as this tends to correspond to peak diversity of ephemeral algae and many fish species in inshore reef systems in south-east Tasmania.

Because of the natural seasonal variability in reef biodiversity, repeat surveys should be conducted at the same time of year as the original survey. The frequency of surveys is another important consideration. Surveying every five years is standard practice in areas where finfish aquaculture occurs. This is likely to be sufficient for assessing change, particularly when undertaken in conjunction with biannual RVA.

To analyse data from biodiversity surveys the abundance data for all species can be tallied per site for each of the three census techniques (fish abundance, invertebrates and cryptic fish and macroalgae and sessile invertebrates). Where more detailed analysis is required, data can also be assessed using univariate or multivariate techniques. When biodiversity surveys are undertaken as part of ongoing monitoring, changes in abundance of all species can be compared using multiple survey results.

4.2.2 Targeted assessment

Rapid visual assessments (RVA)

RVAs of inshore reefs are undertaken by two divers. At sites chosen for RVA, a 50 m permanent transect should be set up along the 5 m depth contour. This can be achieved by installing two permanent eyebolts, 50 m apart, onto the substrate, which can then be used by divers to tension a 50 m length of rope each time a survey is conducted. It is important to record the position of the start and finish of the chosen transect location using a GPS. Sites may need maintenance between surveys to ensure eyebolts and floats remain intact.

To undertake the survey, divers move along the transect line, attaching a 1 m² quadrat at random intervals and photographing it for archival purposes. The quadrat is then scored by the divers using a scorecard featuring 15 defined functional parameters (Figure 8, Table 11).

These include ten parameters to assess broad structural elements:

- » Total canopy cover as a percentage including percentage cover of each of the major canopy forming species. In southeast Tasmania, the two most common canopy forming species are *Phyllospora comosa* (crayweed) and *Ecklonia radiata* (common kelp).
- » Percentage cover of sub-canopy brown macroalgae
- » Percentage cover of sub-canopy green macroalgae
- » Percentage cover of sub-canopy red macroalgae
- » Percentage cover of encrusting pink coralline algae
- » Percentage cover of encrusting red attenuating algae
- » Percentage sponge cover and type
- » Turfing algae cover
- » Levels of encrusting fauna on macroalgae (recorded categorically as none, low, medium, high)
- » Numbers of dominant mobile invertebrates

As well as five parameters to assess enrichment:

- » Percentage cover of epiphytic algae
- » Percentage cover of filamentous algae
- » Percentage cover of nuisance/opportunistic green algae. In southeast Tasmania, observed opportunistic green algae includes the combined total cover of *Ulva/Enteromorpha*, *Chaetomorpha billardierii* and *Cladophora* spp.
- » Percentage cover of nuisance/opportunistic red algae. In southeast Tasmania, observed opportunistic red algae includes *Asparagopsis armata*.
- » Levels of “dust” covering algae (recorded categorically as none, low, medium, high)

Figure 8: Diver undertaking an RVA assessment on a rocky reef





To increase scoring accuracy, the 1 m² quadrat is sub-divided into 4 x 0.25 m² sections. All parameters are assessed in each subsection, except for the substrate parameters (encrusting pink coralline algae, encrusting red attenuating algae, sponge and turfing algae), which are only assessed in the 0.25 m² subsection closest to the transect line and used to determine dominant substrate cover.

To improve data quality assurance and quality control, a calibration is done at the beginning of each survey. Quadrats 1 and 2 at each site should be visually assessed by both divers and differences between scoring addressed in situ to ensure consistency in scoring the remaining quadrats. Following this in-water validation, either one or both divers can continue to score the remaining quadrats at the site. After scoring quadrats, the transect line should be filmed using a hand-held camera to retain footage for archival purposes.

RVA survey frequency is an important consideration and will depend on the research questions involved. We recommend undertaking two surveys per annum (biannually), one in late summer (February/March) and one in late winter (July/August).

If enrichment parameters persist across both late summer and late winter periods (i.e. sustained presence of epiphytic or filamentous algae), it is more likely to indicate a continuous nutrient source. Surveys at different times of year also help to better capture natural variability in the system.

Survey data can be analysed using both univariate and multivariate techniques. Examination of individual parameters can be useful in understanding variation in functional response to specific pressures (natural and anthropogenic), whereas multivariate analysis can be used to explore functional differences either between sites or across a system in response to an impact.

If RVA surveys indicate loss of canopy or extreme proliferation of enrichment indicator species, remote assessment techniques should be considered to better understand the spatial extent of impact.

More information on RVA surveys can be found in the “Illustrated scoring guide to Rapid Visual Assessment” (White & Hartog, 2025).



Circle Quadrat #	7		8		9		10		11		12	
Total % canopy												
Pcom / Sarg %												
Era / Lcor %												
% Sub- canopy brown + major spp.												
% Sub-canopy green + major spp.												
% Sub-canopy red												
% Epiphytic algae												
% Filamentous algae												
% Ulva / Chaeto / Cfe (adult only)												
% Asparagopsis												
Substrate characterisation												
% UALCC & type Pink vs. Red	P	R	P	R	P	R	P	R	P	R	P	R
% Sponge & type												
% Turfing algae (<2cm) (% R / % B / % G)												
Feather stars (N/L/M/H)												
MMI spp. and #												
Dust on algae (N/L/M/H)												
Enc. spp. on algae (N/L/M/H)												

Site comments:

4.2.3 Remote assessments

Where concern over the presence of filamentous or epiphytic algae is identified at RVA sites, or a widespread loss of canopy is suspected, remote surveys can be undertaken to monitor or better understand the spatial extent of any loss of ecosystem health.

To undertake a remote survey, suitable reef (or seagrass) sites within the area of concern should be identified, with site suitability dependent upon having substrate or canopy within the 3-5 m depth range. Video footage at 4K resolution or better can then be collected using either ROV or a towed video camera.

For simple but effective georeferencing, a towed camera system is recommended, with the survey area recorded via the GPS system of the vessel. Towed camera systems with both forward- and downward-facing cameras are recommended, as they provide capacity to score both qualitatively (forward-facing) and quantitatively (downward-facing).

Where footage is collected by ROV, the ROV should be deployed at the GPS site location and piloted directly down to the canopy or substrate.

Where possible, the ROV should then be piloted parallel to the coastline, ensuring that the substrate and depth remain consistent for the duration of the survey. The dominant substrate type should remain in the field of view throughout. To ensure that footage is suitable for analysis, the ROV should travel on a consistent heading at a speed of 0.1 m/s for one minute before returning to the surface. The heading, swim speed and end point should be recorded for each site. A USBL system is ideal for providing exact georeferencing for the ROV, but consistent swim speeds and soak times may be used as a substitute for qualitative analysis, if that is all that is required.

After video footage is collected, it should be post-processed and analysed in the laboratory. Initially, footage from each site should be cropped so that only the first continuous, usable 30 seconds of footage is retained. For each 30 second video, 5 second segments are then scored for epiphytic/nuisance algal growth using a qualitative scoring scale (Table 12). Canopy cover can also be scored in this manner.

Table 11: Qualitative score for epiphytic algae used in remote surveys

Score	Description	Percentage cover
0	No epiphytic growth present	0%
1	Very low epiphytic growth, virtually clean plant	≤5%
2	Low, minimal epiphytic growth	5% – 20%
3	Medium, obvious epiphytic growth	20% – 60%
4	High, most of the plant covered in epiphytic growth	60% – 90%
5	Very high, plants completely covered, blanket epiphyte	90% – 100%

The scores for each of the 5 second video segments are used to calculate a site average score. Site averages can then be mapped across the region of concern using points graded in size and colour to visualise results. To ensure consistency, each site should be scored independently by two people.

Where differences in site means exceeds 0.5, a third scorer should be involved to determine a final score.

The footage can also be analysed quantitatively using percentage cover techniques from benthic stills obtained from the downward-facing camera.

4.3 Deep reefs surveying and analyses techniques

Methods for monitoring the interactions between finfish aquaculture and deep reefs in Tasmania were trialled through FRDC 2018/131 (Storm Bay Observing System: Assessing the Performance of Aquaculture Development) (Strain et al. 2023). Quantitative surveys using techniques established through the NESP Hub were trialled, along with a more qualitative technique that was less resource intensive. Both methods focus on identifying the major taxonomic groups present and reporting on substrate characteristics. The quantitative biodiversity assessment involves a more complex analysis, where all biota are identified to the lowest taxonomic or morphological group possible and their abundance quantified. Conversely, the qualitative assessment focuses primarily on recording the presence of key taxonomic groups and is less powerful at detecting change. Further research is needed for these techniques to be fully fit for purpose, with limitations highlighted through FRDC 2018/131 (Storm Bay Observing System: Assessing the Performance of Aquaculture Development) (Strain et al. 2023). The development of a targeted assessment for deeper systems, similar to RVA surveys for inshore reefs, that assesses specific pathways of nutrient and organic enrichment from finfish aquaculture should be a priority for future research efforts.

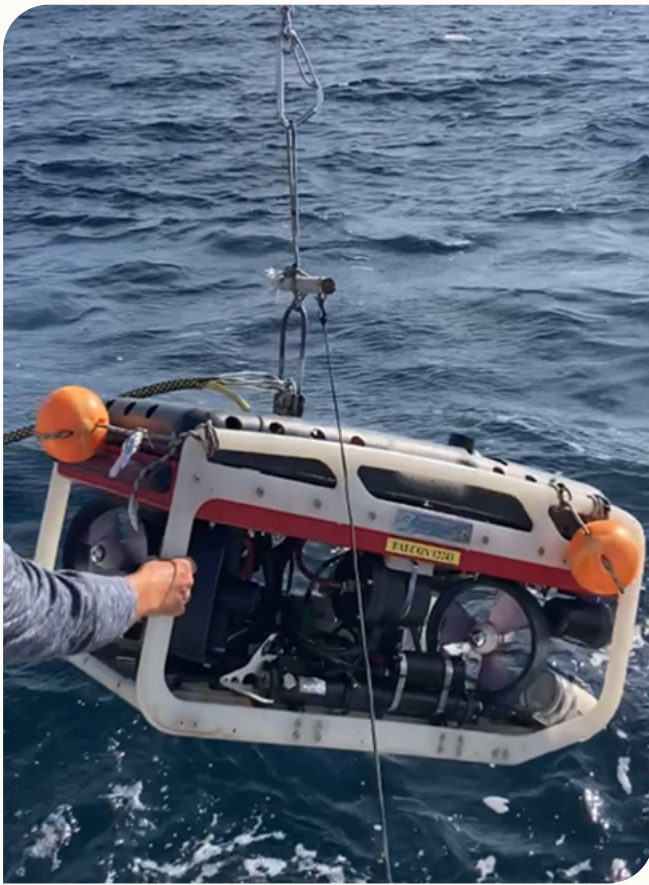
As SCUBA diving is restricted on deep reefs due to depth, monitoring relies on digital imaging technologies to collect video footage for assessment (Strain et al. 2023). While Automated Underwater Vehicles (or AUVs) and ROVs can both be used for collecting underwater visual footage of the benthic environment, towed camera or ROV can be used to capture imagery for assessment of fish and mobile invertebrates. Given ROVs can capture both benthic and forward-facing imagery, they are the more appropriate choice for undertaking deep reef monitoring in Tasmania (Strain et al. 2020).

Greater manoeuvrability of ROVs also makes capturing high-quality footage an easier task compared to both AUV or towed video methods.

Prior to undertaking deep reef surveys, it may be beneficial to carry out high-resolution seafloor mapping to identify the presence and spatial extent of deep reef habitat. This can be undertaken using multibeam mapping sonar (e.g. WASP S3) with differential GPS georeferencing (e.g. from a stand-alone Marinestar 9205 GNSS receiver). Further detail on high-resolution seafloor mapping is beyond the scope of this field guide.

4.3.1 Deep Reef Quantitative Biodiversity Assessment

An ROV (e.g. Saab Falcon ROV) with a dual forward- and downward-facing camera system is recommended for these surveys (Figure 9). Dual (stereo) camera systems provide the capability to measure distance within the field of view, thus providing the means to measure the length of fish or other species identified within a survey. The ROV should be fitted with lasers on the downward facing camera to provide a quantitative measurement of the area captured in images. An ultra-short baseline (USBL) system should also be used to georeference the collection of footage for a quantitative biodiversity assessment. This USBL system is linked to a differential GPS to ensure precise underwater positioning of the ROV throughout the survey. The USBL system consists of a pole mounted transceiver located below the hull of the vessel and a transponder mounted on the ROV allowing the absolute location and depth of the ROV to be recorded at 1-2 second intervals).

Figure 9:**a) Falcon ROV**

The biodiversity assessment is undertaken by piloting an ROV along a 200 m transect at each survey site. Previous work in Storm Bay indicates that 3-5 transects per site is adequate for capturing fish biodiversity, although for benthic biodiversity the number of transects required to capture the full biodiversity of a site is great than five, with further research needed in this space (Strain et al. 2023). However, as a starting point, we recommend a minimum of five transects to be collected at each site. To ensure that video footage collected is suitable for undertaking a visual census, the ROV should operate at no more than 1 m above the seafloor, with a speed no greater than 0.5 knots. Video should be recorded in 4K, using the forward-facing stereo cameras to capture footage with approximately 60% substrate and 40% water body present within the field of view.

**b) Falcon ROV control unit.**

The downward facing cameras should be set at a 90° angle and equipped with a strobe and/or LED lights. Video captured at 4K was trialled by IMAS to develop a method to best capture the benthic environment on deep reefs. Video footage can be used to assess larger mobile invertebrates that were unlikely to be captured in benthic quadrats, with image grabs taken from the footage to score benthic quadrats. All images from the downward-facing system need to be captured perpendicular to the substrate and contain the laser marks to ensure consistency of sampling effort (i.e. all benthic quadrats are roughly the same size).

In biodiversity assessments using ROV, the stereo forward-facing camera system is used to capture footage to assess all fish and mobile invertebrate species. Video files from the forward-facing cameras can be imported into a video transect analysis program (i.e. EventMeasure), where all fish within the 200 m transect can be identified, counted and measured. Following this, all mobile invertebrates can be identified and counted, although this may be better done from the downward-facing footage. This will provide data on the abundance, biomass and size class distribution of fish and mobile invertebrates at each site.

To ensure that fish length measurements are accurate, it is important to calibrate the left and right stereo cameras before and after data collection. Calibration of the forward-facing stereo cameras should be undertaken in the pool, both prior to and after the field surveys. Calibrations are conducted using a 3D calibration cube as a calibration target. Footage of the calibration target must be taken at a range of angles of orientation and aspect. Processing to synchronise images from both cameras and to produce a calibration factor can then occur. This factor is applied within the EventMeasure software to enable 3D measurement, including measurement of fish lengths.

Using the still images or video from the downward-facing camera, biota and substrate on the seafloor will be assessed. This is undertaken by analysing images along the transect using an image software analysis program (e.g. Squidle+). Further research is required to better understand how many images are needed in each 200 m transect, but one in every 10 m is a good starting point. Using the image analysis software, 30 points are generated on each image. At each point, the biota should be identified to the lowest taxonomic group possible. Where identification to species level is not possible, categorisation using the Collaborative and Automated Tools for Analysis of Marine Imagery (CATAMI) classification scheme should be used.

In addition, substrate should be categorised using CATAMI. The CATAMI scheme uses defined descriptors for specific biota to provide a consistent set of identifiers across a variety of marine environments (Althaus et al. 2015). Using the values obtained through point count analysis, estimates of percentage cover can be made for each species or category observed.

To present data in a consistent manner, abundance data for all species identified on transects should be tallied per site, with a site average across the five transects reported for each species. Summary tables for species abundance per site can then be produced for fish, mobile invertebrates, macroalgae and sessile invertebrates. Where quantitative biodiversity surveys are undertaken post-baseline, any site changes in abundance of species identified can be compared to the baseline survey results. Data can be further explored using both multivariate and univariate statistical techniques.

4.3.2 Deep reef Qualitative Assessment

To undertake a qualitative assessment of deep reefs, an ROV or towed camera with a forward-facing camera system should be used. In a qualitative assessment, the forward-facing camera system is used to capture footage from which an assessment can be made, such as identifying all biota to the lowest taxonomic group possible.

The survey can be undertaken by piloting an ROV or towing a video camera along 200 m transects at each survey site. Where an ROV is used, the location of each transect should be recorded (e.g. using a USBL system with a pole mounted transceiver located below the hull of the vessel and a transponder mounted on the ROV, allowing the absolute location and depth of the ROV to be recorded at 1-2 second intervals). To enable accurate assessment, it is important that footage is recorded at 4K quality, that the camera travels at a speed no greater than 0.5 knots and for the lens to be angled to capture a field of view that is 60% substrate and 40% water body. Where possible, 3-5 transects should be captured at each site.

To assess the video footage, all biota should be identified to the lowest taxonomic group possible. In some cases, identification to species level may not be possible for some algae and sessile invertebrates, in which case the CATAMI classification scheme should be used instead. For fish and mobile invertebrates identified on transects, an estimate of abundance can be made using the following abundance bins: 1, 5, 10, 50, 100, 500, 1000+. Information on visibility, depth and substrate should also be recorded within the assessment.

To present the data in a consistent manner, data should be tabulated for site, depth (m), visibility (m), substrate, algae, sessile invertebrates, mobile invertebrates and fish. In each of these categories, the dominant species (or CATAMI group) for each category should be listed for each site surveyed. For mobile invertebrates and fish, abundance estimates should also be presented in a separate table.

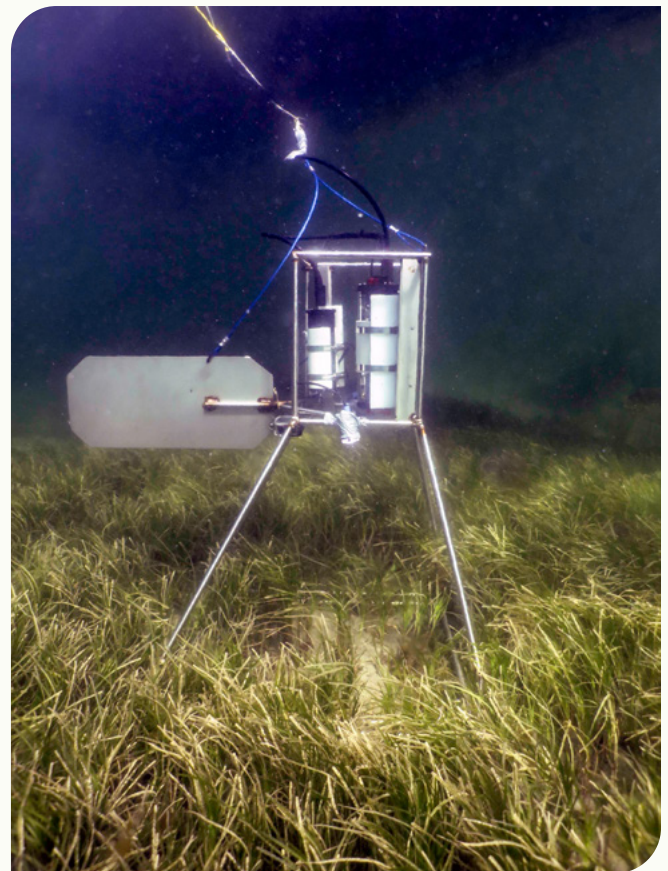
4.4 Seagrass surveying and analysis techniques

The two most important characteristics for assessing the robustness of seagrass are a) bed extent and b) seagrass health. Bed extent can be obtained by mapping the area of interest, and three methods for monitoring seagrass health have been developed in Tasmania to address different monitoring questions. They include 1) an ecological assessment of seagrass bed & associated biota, 2) a quantitative bed condition assessment and 3) a qualitative bed condition assessment. See Section 3.4 for a more detailed description of how these methods are used to determine effects of finfish farming on seagrass ecosystems.

4.4.1 Seagrass bed extent mapping

Bed extent surveys are best undertaken using a drop camera (e.g. internet protocol (IP) camera). If possible, the drop camera should be mounted in a frame with a built-in 0.5 x 0.5 m quadrat to ensure consistency of image collection (Figure 10). Either 4K video footage or still images should be taken, with images captured by lowering the drop camera to the substrate along the transect line.

Figure 10: Seagrass frame with drop camera and built-in 0.5 x 0.5 m quadrat



To establish bed extent, underwater footage or still images should be taken along parallel transects perpendicular to the shore. To ensure the entire bed is mapped, surveys should start outside the bed at the seaward side and work backwards towards the shore. Where the seaward edge of the bed cannot be located within a reasonable depth range, the commencement depth of the survey should be recorded. To effectively map the bed edges, transects should be no greater than 50 m apart, with this distance reduced as necessary (e.g. 10-20 m) in areas with significant undulation or irregularity in bed extent. When using a drop camera, georeferencing can be undertaken using the vessel's differential GPS system.

The camera tether must be kept vertical to ensure geolocation accuracy. Capturing images approximately every 10 m is recommended initially, with this revised as necessary depending on the variability, complexity and size of the seagrass bed.

After surveying, video footage should be reviewed to classify seagrass density every 1-2 m along transects. The results should be mapped in GIS, including the location of all transect lines. These descriptors are based on the SeaMap Tasmania classification system for marine substrate (Table 13).

Table 12: Seagrass bed classification categories for mapping bed extent

Seabed category	Definition
Sand	Totally bare substrate, no bed forming species present
Sparse seagrass	Low seagrass cover <33% (as defined at the SeaMAP convention).
Patchy seagrass	Dense seagrass cover (>33%) with bare substrate patches in image
Seagrass	Seagrass cover (>33%) evenly distributed throughout the quadrat
Seagrass/macroalgae	Two bed forming taxa present, e.g., seagrass and <i>Caulerpa</i> , neither appears to be dominant.
Macroalgae	Macroalgae is the dominant bed forming species (e.g., <i>Caulerpa</i>)

When carrying out seagrass bed extent mapping it is important to ensure survey timing is consistent between years. Regular surveys conducted at the same time ensures that changes in bed extent can be effectively tracked over time.

4.4.2 Seagrass health

Three approaches for monitoring seagrass health in relation to impacts from finfish farming have recently been trialled by IMAS (Strain et al. 2023). These three methods range from a full ecological assessment to a highly qualitative rapid assessment, providing a hierarchy in terms of the quantitative understanding of seagrass bed ecology and biodiversity.

The ecological assessment is a detailed and comprehensive assessment of the health of the seagrass bed and all associated biota. The two bed condition assessments focus on the seagrass bed and associated epiphytic communities, with the quantitative method providing a higher level of detail than the qualitative method.

An ecological assessment of a seagrass bed and associated biota is generally used in baseline surveys or in situations where a full understanding of seagrass ecosystems is critical (e.g. in beds with high commercial or biological significance). Quantitative bed condition assessments are simpler to carry out and focus on providing measurable and repeatable information on spatial and temporal change at specific sites.

In contrast, the qualitative condition assessment methodology was designed as a broadscale tool for assessing seagrass health where changes have been identified from a quantitative bed condition assessment or ecological surveys. See Section 3.4.2 for background information on how these methods are used to determine interactions between finfish aquaculture and seagrass beds.

Ecological assessment of seagrass bed and associated biota

To provide a comprehensive survey of seagrass health, an ecological status assessment can be carried out. These surveys are designed to provide detailed information on the ecological status of seagrass bed ecosystems including fish, macroinvertebrates, infauna and epibiota associated with the bed. Ecological assessments of seagrass beds can be undertaken as part of baseline assessments or as necessary.

To undertake the assessment, 4 x 50 m transects are laid across the main seagrass bed of interest. Depth of transects will depend on the bed of interest, but should be recorded and kept consistent across surveys. Start and end points of each transect should be georeferenced using differential GPS. Once established, video footage of the transect lines should be collected by diver with handheld camera.

Using the transect, an ecological census of the area is then performed for the following:

Fish abundance and size:

a diver swims parallel to the transect line and counts all fish observed within 5 m on either side, identifying species where possible and recording size. Complementary fish survey techniques may be required (e.g. BRUV or seine netting) to fully capture fish biodiversity at each site.

Mobile invertebrates:

all invertebrates >3 cm are recorded in 1 m blocks by a diver swimming parallel to the transect line. All observed invertebrates should be identified to species level where possible and counted.

While the invertebrates are being assessed by one diver, a second diver can obtain video footage of the transect line for qualitative assessment and archival purposes.

Seagrass bed composition:

0.25 m² quadrats should be placed randomly along the transect line at 10 m intervals (i.e. 5 quadrats per 50 m transect). After placing each quadrat, a still photograph should be taken for quantification of percentage cover of all seagrass and algae in the laboratory.

Seagrass condition & benthic infauna and epifauna:

150 mm diameter sediment cores are taken along the transect line every 10 m (i.e. 5 cores per 50 m transect). To take the cores, a diver should randomly toss a core tube to the side of the transect line (within 1 m) at 10 m intervals, and then gently push the core tube into the sediment to 100 mm depth.

The contents of the core should then be placed in a mesh bag with 1 mm mesh size by the diver. After closing the bag, the contents of the bag should be gently agitated underwater to remove sediment. Alternatively, the diver can return the whole intact core to the surface and gently pass the sediment through a 1 mm mesh sieve. The sample should be preserved in a 10% buffered formalin solution.

When processing the core in the laboratory, each species of seagrass should be separated, and then for each species, the seagrass partitioned into blades, stems and rhizomes. Any epiphytic algae should then be stripped from the seagrass leaves and sorted into different species. Any non-epiphytic macroalgae present in the retained core sample (e.g. *Caulerpa* sp. etc) should be identified to species level where possible.

Once this has been undertaken, all epifauna, epiphyte and macroalgae retained from the core sample should be washed thoroughly and dried by placing in an oven at 60 °C or freeze drier until a constant weight is achieved. This final weight is recorded in g dry mass/m².

Once the seagrass and macroalgae have been removed from the core sample, the remaining material should be collected to assess benthic infauna and epifauna.

Using the preserved benthic infauna and epifauna sample, all specimens should be identified to the lowest taxonomic level possible and species abundance recorded.

Sediment chemistry:

In the same location that a core was taken for assessment of seagrass condition, benthic infauna and epifauna, a duplicate 150 mm diameter core should also be taken for sediment chemistry. Analysis for sediment chemistry should be undertaken following the same procedures outlined in Section 4.1.3 of this field guide.

Quantitative bed condition assessment

Quantitative bed condition assessments provide key information for assessing spatial and temporal change associated with seagrass beds. They are undertaken using photoquadrats from which values of percentage cover of seagrass, green macroalgae and associated epiphytic and wrack algae can be determined. The frequency of sampling may vary depending on the research questions involved. Where seasonal variability is of interest, quarterly or bimonthly sampling is recommended.

Quantitative bed condition assessments are carried out by capturing benthic images at 10 m intervals along transect lines using a drop camera set up with a downward facing camera capable of capturing in 4K resolution. A 0.5 x 0.5 m quadrat frame should be attached to the drop camera and centred below the camera lens to ensure that the quadrat frame accounts for most or all of the field of view of the camera. To take consistent benthic images, the drop camera should be lowered to the seafloor so that the quadrat sits on the substrate and is able to take an image perpendicular to the seafloor (Figure 10). Using a differential GPS, the location of photoquadrats can be recorded.

After the field survey is complete, photoquadrats can be used to undertake a condition assessment. Any photoquadrats containing sand only are excluded from the analysis.

To carry out the assessment, parameters (including percentage cover of seagrass cover, epiphytic algae on seagrass, *Caulerpa* spp., wrack algae (brown, green and red) and other algae cover (brown, green and red), and information on substrate and fauna) are quantified through standardised image analysis using Coral Point Count with Excel extensions (or CPCe) software (Kohler & Gill 2006), or a similar image capture program. Using this software, 50 random points are distributed across each image. The user can then identify the feature (parameter) underneath each point, assess and record.

While the number of transects and the number of photoquadrats within transects needed to adequately and accurately assess the bed will vary from site to site, 5 x 100 m transects with 10 photoquadrats in each is recommended as a good starting point.

Assessment should be undertaken using a standardised scorecard to enable consistency between surveys (Table 14). Percentage cover for each parameter can then be calculated out of 50 for each quadrat by doubling each point count. Data can then be presented in summary tables with visualisation using bar-graphs.

To ensure consistency in analysis it is recommended that data from photoquadrats is averaged across each transect for each year class. Site averaged data from each transect can then be reported in summary tables for percentage cover of all parameters scored. To help visualise the data, bar charts can be produced for each of the parameters quantified in Table 14. To explore temporal trends in data, site averaged data for every year class can be used for each of the above parameters and incorporated into bar charts, with a separate chart for each site and parameter.

It should be noted that these methods have been developed and tested by IMAS on *Zostera tasmanica* complex beds in the south-east of Tasmania. While methods should be transferrable to *Ruppia* sp. or *Amphibolis* sp. elsewhere in the state, a conservative approach regarding replication and choice of method should be applied in the first instance.

Table 13: Scorecard for assessing vegetation and other features of interest in photoquadrats as part of the seagrass quantitative bed condition assessment in southeast Tasmania

Site:

Date:

Transect no:

Quadrat no:

Parameters		Points counted	x 2
Seagrass	<i>Zostera</i> spp.		
	Other seagrass spp. (+ ID)		
Epiphytic algae	Filamentous brown		
	Filamentous green		
	Filamentous red		
	Epiphytic brown algae		
	Epiphytic green algae		
	Epiphytic red algae		
	Total epiphytic		
	Total filamentous		
Caulerpa spp.	<i>Caulerpa brownii</i>		
	<i>Caulerpa flexilis</i>		
	<i>Caulerpa geminata</i>		
	<i>Caulerpa longifolia</i>		
	<i>Caulerpa scalpelliformis</i>		
	<i>Caulerpa simpliciuscula</i>		
	<i>Caulerpa trifaria</i>		
	<i>Caulerpa</i> spp.		
	Total <i>Caulerpa</i>		
Wrack algae	Brown wrack algae		
	Green wrack algae		
	Red wrack algae		
	Total wrack		

Parameters		Points counted	x 2
Other macroalgae	Brown algae		
	Green algae		
	Red algae		
	Dead algae		
	<i>Chaetomorpha billardierii</i>		
	<i>Chaetomorpha coliformis</i>		
	<i>Ulva</i>		
	<i>Enteromorpha</i>		
Substrate	Sand		
	Shell grit		
	Cobble		
Fauna	Other fauna spp. (+ ID)		

Qualitative bed condition assessment

Qualitative bed condition assessments are undertaken to provide an assessment of overall seagrass health over a wider spatial extent using estimates of epiphytic cover as a proxy. They have been designed to assess the spatial extent of impacts (observed in quantitative bed condition assessments). Assessments are undertaken using video footage from which a categorical score for overall cover of a) seagrass b) green algae and c) the epiphytic growth on seagrass is determined.

To collect video footage, video transects should be taken along transect lines across seagrass beds. This involves laying down a lead-core rope in the desired transect location, marked at 10 m intervals. To ensure that the transect can be effectively georeferenced, the start and end of the transect should have surface floats attached.

Video footage can be collected by diver with handheld video, towed video or ROV with video recording capability. To ensure that footage is of suitable quality for assessment purposes it is important that the video camera used can capture footage at 4K quality. In addition, the video camera should be moved along the transect line at a speed no greater than 0.5 knots, and no higher than 1.5 m above seabed cover (i.e. seagrass or epiphyte) and should be angled appropriately so that the field of view includes about 60% substrate and 40% water body.

After collection, the video footage can be reviewed and broken into 10 m increments (using the markers on the transect line as a guide). Each 10 m section is then scored for seagrass cover, green algae cover and epiphytic cover using a categorical score scale (Table 15). This data can be averaged across the transect or mapped against each 10 m increment within the transect to examine bed dynamics.

Table 14: Categorical score scale for assessing seagrass bed condition

Categorical score		0	1	2	3	4	5
Cover description	Seagrass and green algae	None (0%)	Very low (1-5%)	Low, minimal (5-20%)	Medium, obvious (20-60%)	High (60-95%)	Very high (95-100%)
	Epiphytic cover	None (0%)	Very low, virtually clean seagrass plant (1-5%)	Low, minimal (5-20%)	Medium, obvious (30-60%)	High, most of seagrass plant covered (60-95%)	Very high, seagrass plant completely covered (95-100%)

References

- Althaus F, Hill N, Ferrari R, Edwards L, Przeslawski R, Schonberg CH, Stuart-Smith R, Barrett N, Edgar G, Colquhoun J, Tran M, Jordan A, Rees T, Gowlett-Holmes K (2015) A Standardised Vocabulary for Identifying Benthic Biota and Substrata from Underwater Imagery: The CATAMI Classification Scheme. *PLoS One* 10:e0141039
- Apostolaki ET, Tsagaraki T, Tsapaki M, Karakassis I (2007) Fish farming impact on sediments and macrofauna associated with seagrass meadows in the Mediterranean. *Estuar Coast Shelf Sci* 75:408-416
- Aquenal Pty Ltd (2019) Trumpeter Bay MF281 - East of Yellow Bluff: Baseline environmental assessment. Aquenal Pty Ltd, Tasmania
- Aylagas E, Atalah J, Sánchez-Jerez P, Pearman JK, Casado N, Asensi J, Toledo-Guedes K, Carvalho S (2021) A step towards the validation of bacteria biotic indices using DNA metabarcoding for benthic monitoring. *Mol Ecol Resour* 21:1889-1903
- Aylagas E, Borja A, Rodríguez-Ezpeleta N (2014) Environmental Status Assessment Using DNA Metabarcoding: Towards a Genetics Based Marine Biotic Index (gAMBI). *PLoS One* 9:e90529
- Aylagas E, Borja Á, Tangherlini M, Dell'Anno A, Corinaldesi C, Michell CT, Irigoien X, Danovaro R, Rodríguez-Ezpeleta N (2017) A bacterial community-based index to assess the ecological status of estuarine and coastal environments. *Mar Pollut Bull* 114:679-688
- Ball D, Soto-Berelov M, Young P (2014) Historical seagrass mapping in Port Phillip Bay, Australia. *J Coast Conserv* 18:257-272
- Bannister J, Sievers M, Bush F, Bloecher N (2019) Biofouling in marine aquaculture: a review of recent research and developments. *Biofouling* 35:631-648
- Barrett N, Monk J (2021) Progress towards a nationally integrated benthic biodiversity monitoring program for Australia's marine realm. Report to the National Environmental Science Programme, Marine Biodiversity Hub. Institute for Marine and Antarctic Studies, University of Tasmania, Hobart
- Barrett N, Sanderson JC, Lawler M, Halley V, Jordan A (2001) Mapping of inshore marine habitats in South-Eastern Tasmania for marine protected area planning and marine management. Tasmanian Aquaculture and Fisheries Institute, University of Tasmania, Hobart
- Bastiaansen A (2020) Quantifying epibenthic assemblages across a depth gradient for monitoring in Storm Bay, Tasmania. Honours Thesis, University of Tasmania, Hobart
- Burkholder JM, Tomasko DA, Touchette BW (2007) Seagrasses and eutrophication. *J Exp Mar Biol Ecol* 350:46-72
- Connell SD, Ghedini G (2015) Resisting regime-shifts: the stabilising effect of compensatory processes. *Trends Ecol Evol* 30:513-515
- Connell SD, Russell BD, Turner DJ, Shepherd SA, Kildea T, Miller D, Airolidi L, Cheshire A (2008) Recovering a lost baseline: Missing kelp forests from a metropolitan coast. *Mar Ecol Prog Ser* 360:63-72
- Coutts A, Zimmermann D, Davey A, Bowman JP, Ross DJ, Strain EMA (2024) A comparison of visual and molecular methods for inferring biological communities in aquaculture enriched sediments - Impact assessment and cost-benefit analysis. *Mar Pollut Bull* 209:e117172
- Diaz-Almela E, Marba N, Alvarez E, Santiago R, Holmer M, Grau A, Mirto S, Danovaro R, Petrou A, Argyrou M, Karakassis I, Duarte CM (2008) Benthic input rates predict seagrass (*Posidonia oceanica*) fish farm-induced decline. *Mar Pollut Bull* 56:1332-1342
- Edgar GJ, Barrett NS (1997) Short term monitoring of biotic change in Tasmanian marine reserves. *J Exp Mar Biol Ecol* 213:261-279

- Edgar GJ, Barrett NS (1999) Effects of the declaration of marine reserves on Tasmanian reef fishes, invertebrates and plants. *J Exp Mar Biol Ecol* 242:107-144
- Edgar GJ, Davey A, Shepherd C (2010) Application of biotic and abiotic indicators for detecting benthic impacts of marine salmonid farming among coastal regions of Tasmania. *Aquaculture* 307:212-218
- Edgar GJ, Stuart-Smith RD (2014) Systematic global assessment of reef fish communities by the Reef Life Survey program. *Sci Data* 1:140007
- EPA Tasmania (2020) Draft Review of Tasmanian and International Regulatory Requirements for Salmonid Aquaculture. Environment Protection Authority, Hobart
- Fernandez-Jover D, Martinez-Rubio L, Sanchez-Jerez P, Bayle-Sempere JT, Jimenez JAL, Lopez FJM, Bjorn PA, Uglem I, Dempster T (2011) Waste feed from coastal fish farms: A trophic subsidy with compositional side-effects for wild gadoids. *Estuar Coast Shelf Sci* 91:559-568
- Frederiksen MS, Holmer M, Díaz-Almela E, Marba N, Duarte CM (2007) Sulfide invasion in the seagrass *Posidonia oceanica* at Mediterranean fish farms: assessment using stable sulfur isotopes. *Mar Ecol Prog Ser* 345:93-104
- Guinda X, Gracia A, Puente A, Juanes JA, Rzhhanov Y, Mayer L (2014) Application of landscape mosaics for the assessment of subtidal macroalgae communities using the CFR index. *Deep Sea Res II* 106:207-215
- Holmer M, Hansen PK, Karakassis I, Borg JA, Schembri PJ (2008) Chapter 2: Monitoring of Environmental Impacts of Marine Aquaculture. In: Holmer M, Black K, Duarte CM, Marbà N, Karakassis I (eds) *Aquaculture in the Ecosystem*. Springer, Dordrecht, p 47-85
- IMAS (2020) Annual BEMP Report 2019/20. Environmental Licence 10180/1 Marine Farming Lease No 281 at Yellow Bluff. Institute for Marine and Antarctic Studies, Hobart
- Keeley NB, Cromey CJ, Goodwin EO, Gibbs MT, Macleod CM (2013a) Predictive depositional modelling (DEPOMOD) of the interactive effect of current flow and resuspension on ecological impacts beneath salmon farms. *Aquac Environ Interact* 3:275-291
- Keeley NB, Forrest BM, Macleod CK (2013b) Novel observations of benthic enrichment in contrasting flow regimes with implications for marine farm monitoring and management. *Mar Pollut Bull* 66:105-116
- Keeley NB, Forrest BM, Macleod CK, (2015) Benthic recovery and re-impact responses from salmon farm enrichment: Implications for farm management. *Aquaculture* 435:412-423
- Keeley N, Taylor D (2015) A Review of Total Free Sulfide Concentrations in Relation to Salmon Farm Monitoring in the Marlborough Sounds. Cawthron Institute
- Keeley N, Valdemarsen T, Woodcock S, Holmer M, Husa V, Bannister R (2019) Resilience of dynamic coastal benthic ecosystems in response to large-scale finfish farming. *Aquac Environ Interact* 11:161-179
- Keeley N, Wood SA, Pochon X (2018) Development and preliminary validation of a multi-trophic metabarcoding biotic index for monitoring benthic organic enrichment. *Ecol Indic* 85:1044-1057
- Kohler KE, Gill SM (2006) Coral Point Count with Excel extensions (CPCe): A Visual Basic program for the determination of coral and substrate coverage using random point count methodology. *Comput Geosci* 32:1259-1269
- Lapointe BE, Herren LW, Brewton RA, Alderman PK (2020) Nutrient over-enrichment and light limitation of seagrass communities in the Indian River Lagoon, an urbanized subtropical estuary. *Sci Total Environ* 699:134068



Macleod C, Bissett A, Burke C, Forbes S, Holdsworth D, Nichols P, Revill A, Volkman J (2004) Development of Novel Methods for the Assessment of Sediment Condition and determination of management protocols for sustainable finfish cage aquaculture operations. Aquafin CRC Project 4.1/ FRDC Project No. 2000/164. Tasmanian Aquaculture and Fisheries Institute, Hobart

Macleod C, Eriksen R, Simpson S, Davey A, Ross DJ (2014) Assessment of the environmental impacts and sediment remediation potential associated with copper contamination from antifouling paint (and associated recommendations for management). FRDC Project No. 2011/041. Fisheries Research & Development Corporation, Hobart

Macleod C, Forbes S (2004) Guide to the assessment of sediment condition at marine finfish farms in Tasmania. Aquafin CRC Project 4.1 (Extension). Tasmanian Aquaculture and Fisheries Institute, Hobart

Middelburg JJ, Nieuwenhuize J (1998) Carbon and nitrogen stable isotopes in suspended matter and sediments from the Schelde Estuary. *Mar Chem* 60:217-225

Monk J, Barrett N, Bond T, Fowler A, McLean D, Partridge J, Perkins N, Przeslawski R, Thomson PG, Williams J (2020) 10. Field Manual for Imagery Based Surveys using Remotely Operated Vehicles (ROVs). In: Przeslawski R, Foster S (eds) *Field Manuals for Marine Sampling to Monitor Australian Waters*. National Environmental Science Programme (NESP)

Monk J, Barrett N, Hulls J, James L, Hosack G, Oh E, Martin T, Edwards S, Nau A, Heaney B (2016) Seafloor biota, rock lobster and demersal fish assemblages of the Tasman Fracture Commonwealth Marine Reserve Region: Determining the influence of the shelf sanctuary zone on population demographics. National Environmental Research Program, Hobart

MPI (2023) Siting and management guidelines for open ocean aquaculture: Ecological considerations for benthic environments and water quality. Ministry for Primary Industries, Wellington

O'Brien KR, Adams MP, Ferguson AJ, Samper-Billarreal J, Maxwell PS, Baird ME, Collier C (2018) Seagrass Resistance to Light Deprivation: Implications for Resilience. In: Larkum A, Kendrick G, Ralph P (eds) *Seagrasses of Australia*. Springer, Cham, Switzerland

Oh ES, Edgar GJ, Kirkpatrick JB, Stuart-Smith RD, Barrett NS (2015) Broad-scale impacts of salmon farms on temperate macroalgal assemblages on rocky reefs. *Mar Pollut Bull* 98:201-209

Parravicini V, Morri C, Ciribilli G, Montefalcone M, Albertelli G, Bianchi CN (2009) Size matters more than method: Visual quadrats vs photography in measuring human impact on Mediterranean rocky reef communities. *Estuar Coast Shelf Sci* 81:359-367

Pearson TH, Rosenberg R (1978) Macrobenthic succession in relation to organic enrichment and pollution of the marine environment. *Oceanogr Mar Biol Annu Rev* 16:229-311

Pérez M, García T, Invers O, Ruiz JM (2008) Physiological responses of the seagrass *Posidonia oceanica* as indicators of fish farm impact. *Mar Pollut Bull* 56:869-879

Perkins N, Monk J, Barrett N (2021) Analysis of a time-series of benthic imagery from the South-east Marine Parks Network. Institute for Marine and Antarctic Studies and IMOS, University of Tasmania, Hobart

Perkins N, Zhang Z, Monk J, Barrett N (2022) The annotation approach used for marine imagery impacts the detection of temporal trends in seafloor biota. *Ecol Indic* 140:109029

Pochon X, Atalah J, Fletcher L, Laroche O, Elvines D, Zaiko A, Wood S, Keeley N (2021) A molecular index refined for benthic monitoring of salmon farms in the Marlborough Sounds. Prepared for the Benthic Standards Working Group (Marlborough). Cawthron Report No. 3602. 26 p. plus appendices.

Rector ME, Weitzman J, Filgueira R, Grant J (2021) Environmental indicators in salmon aquaculture research: A systematic review. *Rev Aquac* 14:156-177

Ross J, Macleod C, White C, Hadley S, Moreno D, Bush F, Barrett N (2021) Managing ecosystem interactions across differing environments: building flexibility and risk assurance into environmental management strategies. FRDC Project No. 2015/024. Institute for Marine and Antarctic Studies, University of Tasmania, Hobart

Rountos KJ, Peterson BJ, Karakassis I (2012) Indirect effects of fish cage aquaculture on shallow *Posidonia oceanica* seagrass patches in coastal Greek waters. *Aquac Environ Interact* 2:105-115

Sanz-Lazaro C, Belando MD, Navarrete-Mier F, Marin A (2011) Effects of wild fish and motile epibenthic invertebrates on the benthos below an open water fish farm. *Estuar Coast Shelf Sci* 91:216-223

Sherman CD, Smith TM, York PH, Jarvis JC, Ruiz-Montoya L, Kendrick GA (2018) Reproductive, dispersal and recruitment strategies in Australian seagrasses. *Seagrasses of Australia*. Springer

Strain E, White C, Ross J (2020) The Storm Bay Observing System - preliminary review of the sampling parameters and design for assessing the performance of salmon aquaculture. FRDC Project No. 2018/131. Institute for Marine and Antarctic Studies, Hobart

Strain E, White C, Hartog M, Hortle J, Coutts A, Hadley S, Ross J (2023) The Storm Bay Observing System: An evaluation of the sampling parameters and design for assessing the performance of salmon aquaculture. FRDC Project No. 2018/131. Institute for Marine and Antarctic Studies, Hobart

Sward D, Monk J, Barrett N (2019) A systematic review of remotely operated vehicle surveys for visually assessing fish assemblages. *Front Mar Sci* 6:134

Sward D, Monk J, Barrett NS (2022) Regional estimates of a range-extending ecosystem engineer using stereo-imagery from ROV transects collected with an efficient, spatially balanced design. *Remote Sens Ecol Conserv* 8:105-118

Taberlet P, Coissac E, Hajibabaei M, Rieseberg LH (2012) Environmental DNA. *Mol Ecol* 21:1789-1793

Teagle H, Hawkins SJ, Moore PJ, Smale DA (2017) The role of kelp species as biogenic habitat formers in coastal marine ecosystems. *J Exp Mar Biol Ecol* 492:81-98

Tett P, Verspoor E, Hunter DC, Coulson M, Hicks N, Davidson K, Fernandes T, Nickell T, Tocher D, Benjamins S, Risch D, Wilson B, Wittich A, Fox C (2018) Review of the environmental impacts of salmon farming in Scotland. Book Issue 01. SAMS Research Services Ltd, Oban, Scotland

Thompson P, Wild-Allen K, Macleod C, Swadling K, Blackburn S, Skerratt J, Volkman J (2008) Monitoring the Huon Estuary and D'Entrecasteaux Channel for the Environmental Effects of Finfish Aquaculture (Aquafin CRC Project 4.2[2]). CSIRO Marine and Atmospheric Research, Hobart

Valentine JP, Jensen M, Ross DJ, Riley S, Ibbott S (2016) Understanding broad scale impacts of salmonid farming on rocky reef communities. FRDC Project No. 2014/042. Fisheries Research & Development Corporation, Canberra

Volkman JK, Thompson P, Herzfeld M, Wild-Allen K, Blackburn S, Macleod C, Swadling K, Foster S, Bonham P, Holdsworth D, Clementson L, Skerratt U, Rosebrock U, Andrewartha J, Revill A (2009) A whole-of-ecosystem assessment of environmental issues for salmonid aquaculture. CSIRO Marine and Atmospheric Research, Hobart

Waycott M, McMahon K, Lavery P (2014) A guide to southern temperate seagrasses. CSIRO Publishing

White CA, Bannister RJ, Dworjanyn SA, Husa V, Nichols PD, Kutti T, Dempster T (2017) Consumption of aquaculture waste affects the fatty acid metabolism of a benthic invertebrate. *Sci Total Environ* 586:1170-1181

White CA, Bannister RJ, Dworjanyn SA, Husa V, Nichols PD, Dempster T (2018) Aquaculture-derived trophic subsidy boosts populations of an ecosystem engineer. *Aquac Environ Interact* 10:279-289

White C, Brasier M (2021) Rapid visual assessment surveys on rocky reefs in the Derwent Estuary. Institute for Marine and Antarctic Studies, Hobart

White C, McAllister J, Johnson O, Mundy C, Ross J (2021) Assessment of reef condition in the southern D'Entrecasteaux Channel. Institute for Marine and Antarctic Studies, Hobart

White C, Hartog M, Bush F, Elvines DM, Ross DJ (2025) Ecological effects of finfish aquaculture on shallow temperate reef ecosystems. Institute for Marine and Antarctic Studies, Hobart

White C, Hartog M, Brasier M, Ross J (2022) Rapid visual assessment of rocky reef assemblages in Port Arthur, Tasmania. Institute for Marine and Antarctic Studies, Hobart

White C, Hartog M (2025) Illustrated Scoring Guide: Rapid visual assessment of reef ecosystems in Tasmania for nutrient enrichment. Institute for Marine and Antarctic Studies, Hobart

Woodcock SH, Strohmeier T, Strand O, Olsen SA, Bannister RJ (2018) Mobile epibenthic fauna consume organic waste from coastal fin-fish aquaculture. *Mar Environ Res* 137:16-23

Appendix A

Photo guide to key indicator organisms associated with organic enrichment in soft sediment environments

Figure 11: Polychaeta and cnidaria

Annelida: Polychaeta



Capitellidae
Capitella sp.



Capitellidae
Mediomastus sp.



Pectinariidae
Lagis portus

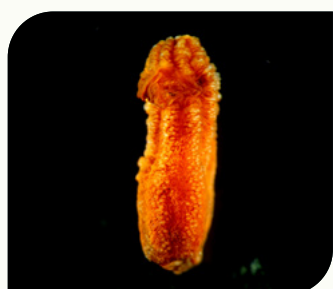


Spionidae
Malacoceros tripartitus



Spionidae
Paraprionospio coora

Cnidaria: Anthozoa



Actiniaria
Edwardsia sp.

Figure 12: Crustaceans**Crustacea: Malacostraca**

Amphipoda
Ampelisca australis



Amphipoda
Parawaldeckia yamba



Amphipoda
Tipimegus cf. thalerus



Amphipoda
Jassa sp.



Amphipoda
Eusiridae (Fm.)



Amphipoda
Australoecetes sp.



Tanaidacea
Apseudes cf. poorei



Leptostraca
Nebalia sp.

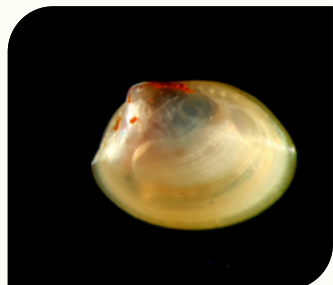
Crustacea: Ostracoda

Myodocopida
Euphilomedes sp.

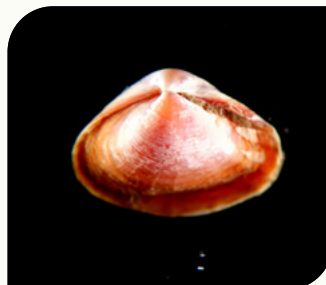


Figure 13: Mollusca

Mollusca: Bivalvia



Nuculidae
Ennucula obliqua



Corbulidae
Varicorbula gibba



Semelidae
Theora lubrica



Cardiidae
Pratulum thetidis

Mollusca: Gastropoda



Nassariidae
Nassarius nigellus sp.

Appendix B

Photographic guide to visual assessment criteria used to assess enrichment in southeast Tasmania and Macquarie Harbour

When scoring groups of fauna, ‘few’ usually refers to one to five individuals, whilst ‘many’ refers to more than five individuals.

1. Gas bubbles

Scoring:

0 = absent

1 = on disturbance (pictured)

2 = spontaneous outgassing



2. Sediment colour (black/grey)

Scoring:

0 = normal

1 = not normal compared to reference site conditions (pictured)



3. Bacterial coverage (*Beggiatoa* spp.)

Scoring:

0 = absent

1 = thin patchy (pictured top)

2 = thick patchy (pictured middle) or thin extensive

3 = thick extensive (pictured bottom) or streaming



4. Pellets/faeces

Scoring:

0 = absent (pictured top — faeces)

1 = few

2 = many

3 = feed spill (pictured bottom)



5. Farm-derived debris

Scoring:

0 = absent

1 = sparse (pictured)

2 = moderate to dense

**6. Burrows/mounds**

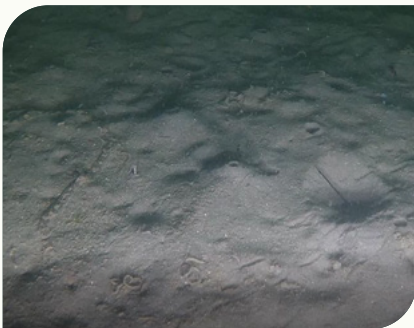
Scoring:

0 = absent

1 = few

2 = many (pictured)

3 = dense

**7. Worm tubes/casts**

Scoring:

0 = absent

1 = present (pictured)

**8. Faunal tracks**

Scoring:

0 = absent

1 = present (pictured)

**9. Algal cover**

Scoring:

0 = absent

1 = sparse

2 = moderate (pictured)

3 = dense

**10. Brittle stars (Ophiuroids)**

Scoring:

0 = absent

1 = present (pictured)

**11. New Zealand screw shell (*Moaricolpus roseus*)**

Scoring:

0 = absent

1 = few (<10)

2 = many (≥10) (pictured)

**12. Dog whelks (*Nassarius* sp.)**

Scoring:

0 = absent

1 = few (<10)

2 = many (≥10) (pictured)

**13. Side-gilled slugs (Pleurobranchidae)**

Scoring:

0 = absent

1 = few (<3) (pictured)

2 = many (≥3)



14. Heart urchins (*Echinocardium cordatum*)

Scoring:

- 0 = absent
- 1 = few (<3) (pictured)
- 2 = many (≥3)



15. Squat lobsters (*Galatheididae*)

Scoring:

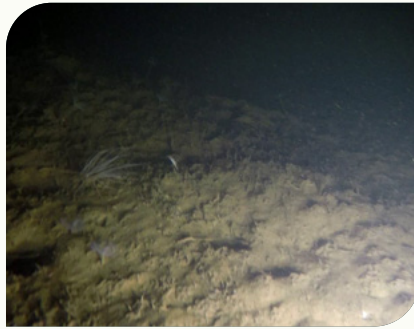
- 0 = absent
- 1 = few (<3) (pictured)
- 2 = many (≥3)



16. Swarming epibenthic crustaceans

Scoring:

- 0 = absent
- 1 = few (<3) (pictured)
- 2 = many (≥3)



17. Other worms (Annelids) including spoon worms (Echiurans)

Scoring:

- 0 = absent
- 1 = few (<3) (pictured)
- 2 = many (≥3)



18. Dorvilleid polychaete worm (*Schistomeringos loveni*)

Scoring:

- 0 = absent
- 1 = few (<30)
- 2 = many (≥30) (pictured)



19. Dorvilleid polychaete worm (*Ophryotrocha shieldsi*)

Scoring:

- 0 = absent
- 1 = few (<30)
- 2 = many (≥30) (pictured)



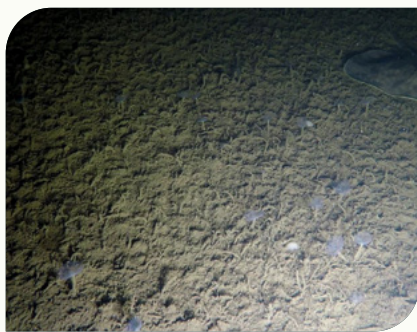
20. Sabellid polychaete worms

Scoring:

0 = absent

1 = few (<10)

2 = many (≥ 10) (pictured)



21. Capitellid polychaete worms (*Capitella* spp.)

Scoring:

0 = absent

1 = few (<10) (pictured)

2 = many (≥ 10)



22. Fish

Scoring:

0 = absent

1 = few (<3) (pictured)

2 = many (≥ 3)



23. Other crustaceans

Scoring:

0 = absent

1 = few (<3) (pictured)

2 = many (≥ 3)



24. Seastars others

Scoring:

0 = absent

1 = few (<3) (pictured)

2 = many (≥ 3)



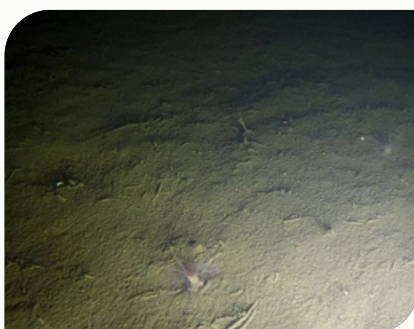
25. Other fauna

Scoring:

0 = absent

1 = few (<3) (pictured)

2 = many (≥ 3)



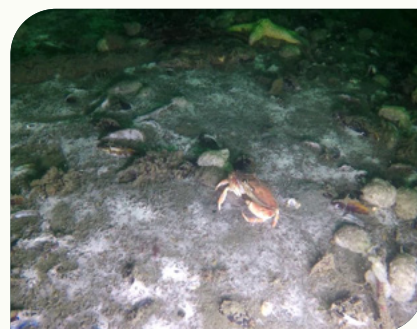
26. Pie crust crab (*Metacarcinus novaezelandiae*)

Scoring:

0 = absent

1 = few (<3) (pictured)

2 = many (≥ 3)



27. Northern Pacific seastar (*Asterias amurensis*)

Scoring:

0 = absent

1 = few (<3) (pictured)

2 = many (≥ 3)





UNIVERSITY of TASMANIA

IMAS 

Institute for Marine and Antarctic Studies