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National Estuary Monitoring Protocol for Aotearoa New Zealand: Site-specific (Fine-scale) Intertidal Monitoring Methods

Prepared for

Ministry for the Environment

June 2025

Salt Ecology
Report 153

Cover and back photo: Rinsing macroinvertebrate cores, January 2025, Kokorua Inlet, Nelson.

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RECOMMENDED CITATION

Forrest BM, Stevens LM, Roberts KL. 2025. National Estuary Monitoring Protocol for Aotearoa New Zealand: Site-specific (Fine-scale) Intertidal Monitoring Methods. Salt Ecology Report 153, prepared for Ministry for the Environment, June 2025. 33p.

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for

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June 2025

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GLOSSARY

AMBI	AZTI Marine Biotic Index (macroinvertebrate index)
ANZG	Australian and New Zealand Guidelines for Fresh and Marine Water Quality (2018)
aRPD	Apparent Redox Potential Discontinuity (assessed visually)
As	Arsenic
BHM	Benthic Health Model (macroinvertebrate index)
Cd	Cadmium
Cr	Chromium
CSIG	Coastal Special Interest Group
Cu	Copper
DGV	Default Guideline Value (ANZG 2018)
GPS	Global Positioning System
Hg	Mercury
LiDAR	Light Detection And Ranging (remote sensing method for measuring elevation)
MfE	Ministry for the Environment
NEMP	National Estuary Monitoring Protocol
Ni	Nickel
Pb	Lead
pRPD	Probe Redox Potential Discontinuity (assessed with instrumentation)
QA/QC	Quality Assurance/Quality Control
RPD	Redox Potential Discontinuity
SACFOR	Super-abundant, Abundant, Common, Frequent, Occasional, Rare (epibiota categories))
SAR	Sediment accumulation rate, also referred to in this document as sedimentation.
SIDE	Shallow Intertidal Dominated Estuary
SOE	State of the Environment (monitoring)
SPI	Sediment Profile Imaging
SSRTRE	Shallow, Short Residence-time Tidal River Estuary
TBI	Traits Based Index (macroinvertebrate index)
TN	Total Nitrogen
TOC	Total Organic Carbon
TP	Total Phosphorus
WRC	Waikato Regional Council
Zn	Zinc

ACKNOWLEDGEMENTS

Many thanks to Hannah Jones (MfE) and members of the Technical Advisory Group (listed below) for reviewing the draft method/s. Thanks to James Rodgers and Anna Tarr (MfE) for contract management. Thanks to Don Morrisey (Salt Ecology) for insights on aRPD, metals and benthic microalgae.

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AC	Auckland Council
ECan	Environment Canterbury
ES	Environment Southland
HBRC	Hawkes Bay Regional Council
NCC	Nelson City Council
MDC	Marlborough District Council
MFE	Ministry for the Environment
NIWA	National Institute of Water and Atmospheric Research
WRC	Waikato Regional Council

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1. DOCUMENT PURPOSE

The National Estuary Monitoring Protocol (NEMP) for Aotearoa New Zealand (hereafter New Zealand), originally written in 2002 [1-3], describes an approach for monitoring intertidal estuary habitats using a suite of estuary-wide (broad-scale) and site-specific (fine-scale) methods. In this update to the NEMP, revisions to the broad-scale [4] and fine-scale methods are supported by an *Overarching Guidance* document [5], which outlines the scope of the update and provides guidance on estuary and indicator selection, as well as monitoring programme design and review. The relationship between these documents is described by Fig. 1.

This document describes methods for **site-specific (fine-scale)** monitoring, focusing on indicators for sedimentation, sediment quality (e.g., grain size, trace metals) and sediment biota (e.g., macroinvertebrates). The document is structured so that Sections 2 to 4 outline some general considerations for fine-scale monitoring, including the selection of sites and their layout for monitoring, an outline of a tiered monitoring framework, and considerations for sample replication and sampling frequency. The content of these sections is not intended to be prescriptive, and should be regarded as **supporting guidance** only.

Section 5 describes a sampling protocol **intended as a prescriptive set of methods** for sampling defined indicators at each fine-scale site. In this way, fine-scale sampling is standardised and repeatable, and results will be comparable among sampling occasions, estuaries and regions.

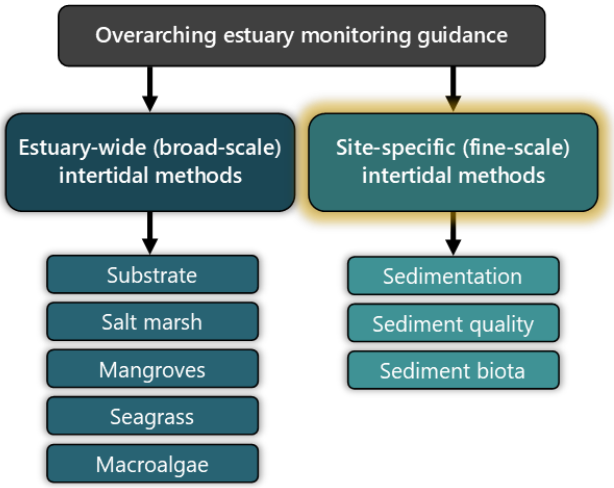


Fig. 1. Conceptual diagram of the NEMP update listing the estuary-wide (broad-scale) and site-specific (fine-scale) indicators being reviewed. This document addresses the fine-scale component highlighted in yellow.

Central, southern and eastern arms of Delaware/ Wakapuaka Inlet, Nelson.

2. FINE-SCALE INTERTIDAL MONITORING OVERVIEW

Fine-scale intertidal monitoring aims to characterise site-specific indicators of estuary state, and capture changes from one survey to the next. Indicators can be compared to thresholds of ecological quality [6], and temporal trends can be assessed where monitoring programmes are long-established [7]. The expectation is that long-term monitoring will enable changes in state at fine-scale sites to be linked to catchment stressors and other drivers.

The fine-scale method in the original NEMP was based on setting-up sites in the mid-low tide zone of the main sand/mud flats of estuaries. Implementation of the protocol has subsequently revealed that positioning sites on the main intertidal flats can disconnect them from immediate catchment influences. This has led to paradoxical situations in which broad-scale habitat mapping can indicate significant areas of degradation (e.g., muddy and eutrophic sediments in estuary side-arms), whereas fine-scale sampling on the main tidal flats of the same estuary can indicate relatively healthy conditions [8, 9]. Understanding the purpose of fine-scale monitoring is therefore integral to the overall sampling design and site selection.

The updated fine-scale method largely retains the overall approach of the original NEMP, with some additions and expansion on the original guidance. The key features are as follows:

- Fine-scale sites are selected in intertidal areas that are relatively uniform in terms of attributes such as substrate characteristics and tidal elevation.
- Each site is configured as a 3x4 grid of 12 sampling plots (Fig. 2). Site dimensions are not critical, and existing monitoring sites vary widely among estuaries and councils (e.g., from as small as ~10x30m up to ~1ha).
- Estuary condition at each site is monitored using the suite of indicators in Table 1. Sedimentation rate is included as new indicator.
- Flexibility is provided for by varying the indicators used across three sampling tiers.

Key method differences between the original NEMP and this revision are outlined in the *Overarching Guidance*, which also provides advice on 'supporting indicators' that assist with understanding estuary condition and drivers of change.

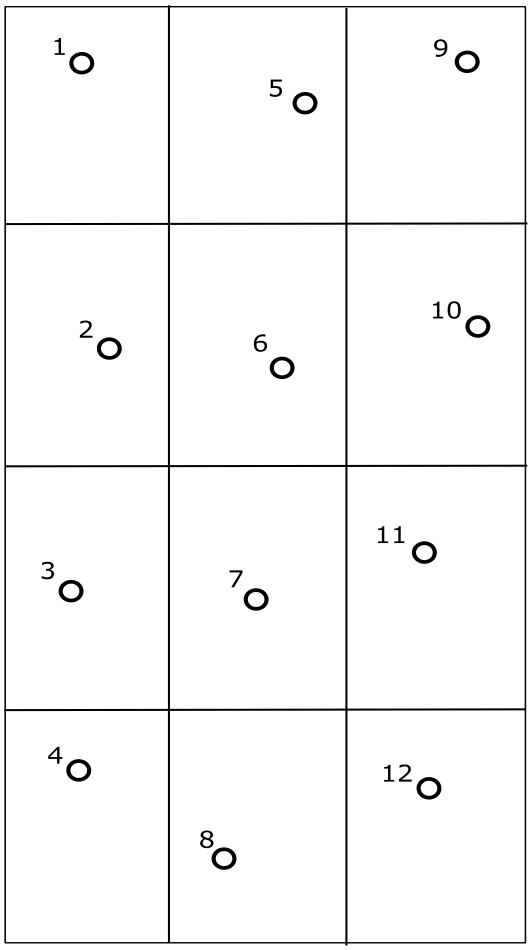


Fig. 2. Schematic of a fine-scale site divided into a 3x4 (columns x rows) grid of sampling plots. The original NEMP recommended discrete indicator sampling within 10 of these plots.

The NEMP fine-scale method document is intended to provide a robust and repeatable methodology for long-term SOE monitoring of intertidally-dominated estuaries that have extensive tidal flats.

Table 1. Description and rationale for site-specific sedimentation, sediment quality and sediment biota indicators proposed for the NEMP update. Further details are provided in the *Overarching Guidance*.

Generic indicator	Site-specific indicator	General rationale
Sedimentation		
Sediment accrual or erosion	Sedimentation rate	Measures the accrual (or erosion) of sediment above buried 'sediment plates'. Excess sedimentation can smother benthic biota and high-value habitats (e.g., seagrass) [10-12]. Sedimentation rate is a useful variable to measure at fine-scale sites, to help interpret changes in sediment quality and biota.
Sediment quality		
Sediment grain size classes	Grain size for %mud <63µm, sand <2mm to ≥63µm & gravel ≥2mm	Indicates the proportion of fine-grained sediments and other sediment types that have accumulated. Mainly used to track the percentage contribution of the mud size fraction. As well as mud itself being a stressor, the contaminant-holding capacity of sediments tends to increase with decreasing particle grain size [6 and references therein].
Trophic state and enrichment	Total Organic Carbon	Measures the organic enrichment of sediments, with high values leading to low sediment oxygenation, which can influence nutrient cycling and habitat suitability for infauna and vegetation (e.g., seagrass) [13-15]. This method is used by 13 of 16 councils in preference to organic content measured as ash-free dry weight (weight loss on ignition; Appendix 1 [16]).
	Nutrients (Total Nitrogen and Total Phosphorus)	These nutrients measure the eutrophication state of sediments. High sediment nutrients can increase the likelihood of algal blooms and other symptoms of enrichment (e.g., low sediment oxygen), and can also alter nutrient cycling and habitat suitability for infauna and vegetation [14, 15, 17, 18].
Oxygenation	apparent Redox Potential Discontinuity (aRPD) depth	Measures the enrichment response of sediments as the depth of transition between brown oxygenated surface sediments, and deeper grey or black oxygen-depleted sediments [15, 19]. The aRPD can occur closer to the sediment surface as organic matter loading or sediment mud content increase [19]. Although a commonly used field measure, aRPD has limitations that need to be considered during field sampling (see Section 5.4.1).
	Colour, odour & microbial growths	Qualitative indicators of eutrophic enrichment [20-23]. Eutrophic sediments can have a 'rotten egg' odour and have an intense black colour throughout the entire profile. In the presence of extreme eutrophication, growths of sulfur-oxidising bacteria and/or microalgae may be visible and conspicuous on the sediment surface.
Toxicants	Arsenic, cadmium, chromium, copper, lead, mercury, nickel, zinc	Toxicants are often associated with human activities such as urban development, but sometimes have natural sources [24-26]. The NEMP uses a suite of trace metals (often referred to as 'heavy metals') and the metalloid arsenic, as indicators. Their concentrations can be used to assess the potential for toxic effects, and may identify the need to investigate other anthropogenic contaminants (e.g., pesticides, hydrocarbons, plasticisers).
Sediment biota		
Macro-invertebrates	Macroinvertebrates	Macroinvertebrates (often called 'macrofauna') are defined as species retained on a 0.5mm mesh after sieving. They provide a food source for birds and fish. Their abundance, composition, and diversity are commonly used indicators of estuary health, especially due to their responsiveness to changes in sediment condition (e.g., enrichment) and sediment mud content [27-30].
Epibiota	Epifauna	Epifauna are surface dwelling organisms (e.g., mud snails) that provide a food source for birds and fish [31]. Their abundance and species composition are indicators of estuary condition.
	Macroalgae	The presence of macroalgae can influence the composition of infauna and epifauna. Macroalgae can also promote the settling of fine sediments, and the composition and prevalence of opportunistic macroalgae (especially species of *Gracilaria* and *Ulva*) can indicate nutrient enrichment [6, 14, 32, 33].
	Microalgae	Microalgae (microphytobenthos) can influence macroinvertebrate composition and indicate eutrophic enrichment [34, 35]. Under enriched conditions, microalgae can be highly conspicuous (e.g., bright yellow or green colour) on the sediment surface. Chlorophyll-*a* is a proxy indicator for microalgal biomass, and phaeopigments (e.g., phaeophytin) are a breakdown product that indicates microalgal senescence or degradation.

3. FINE-SCALE MONITORING PROGRAMME DESIGN

3.1 OVERVIEW

To accommodate council requests for flexibility in assessing estuary condition, cater for varying budgets, and ensure that minimum requirements for systematic SOE monitoring are met, the *Overarching Guidance* outlines two programme components that can be adapted: (i) monitoring effort (tiered monitoring and sampling replication), considered here in Sections 3.2 and 3.3; and (ii) monitoring frequency, considered in Section 3.4.

Adjusting monitoring effort and frequency inevitably involves trade-offs, such as reduced ability to detect temporal trends, limited spatial or temporal resolution, and challenges comparing data across estuaries (Section 3.5). However, even limited monitoring can provide valuable insight into estuary condition, particularly where estuaries are at risk and councils need to implement management actions.

This document assumes that estuary and indicator selection, described in the *Overarching Guidance*, has been completed before choosing an appropriate monitoring tier or sampling frequency (Fig. 3). The *Overarching Guidance* also provides more detail on how to apply the tiered monitoring approach, and addresses programme design considerations that balance the monitoring tiers and sampling frequency with scientific rigour, e.g., spatial nesting, stratified, risk-based, sentinel, and temporal nesting (rotational sampling) designs.

3.2 DESCRIPTION OF MONITORING TIERS

The tiered monitoring approach (Table 2) balances varying levels of resourcing while ensuring monitoring remains robust and as nationally consistent as possible. The tiers are referred to as Basic, Intermediate, and Comprehensive, and differ mainly with respect to the indicators measured. Each tier requires progressively greater effort but offers increasing confidence and a more in-depth understanding of estuary condition.

3.2.1 Basic

The Basic approach draws on a small suite of important indicators that can be measured with minimal effort and cost. The approach involves quantifying sediment mud content, conducting a semi-quantitative, field-based assessment of key trophic state indicators (aRPD, macroalgae, microalgae), and recording qualitative enrichment indicators (e.g., sediment colour & odour). This approach is intended to enable tracking of site-specific trends in a small number of key indicators and attributes of habitat quality that can indicate impacts due to anthropogenic activities. Evidence of changes in state, or declining trends, may trigger escalation to the Intermediate or Comprehensive tiers, or more in-depth targeted investigations [5].

3.2.2 Intermediate

The Intermediate tier scales-up from Basic to include quantification of a wider suite of sediment quality indicators, and adds the sedimentation rate indicator (Table 2). The Intermediate tier also expands to include a semi-quantitative site-wide assessment of conspicuous epifauna.

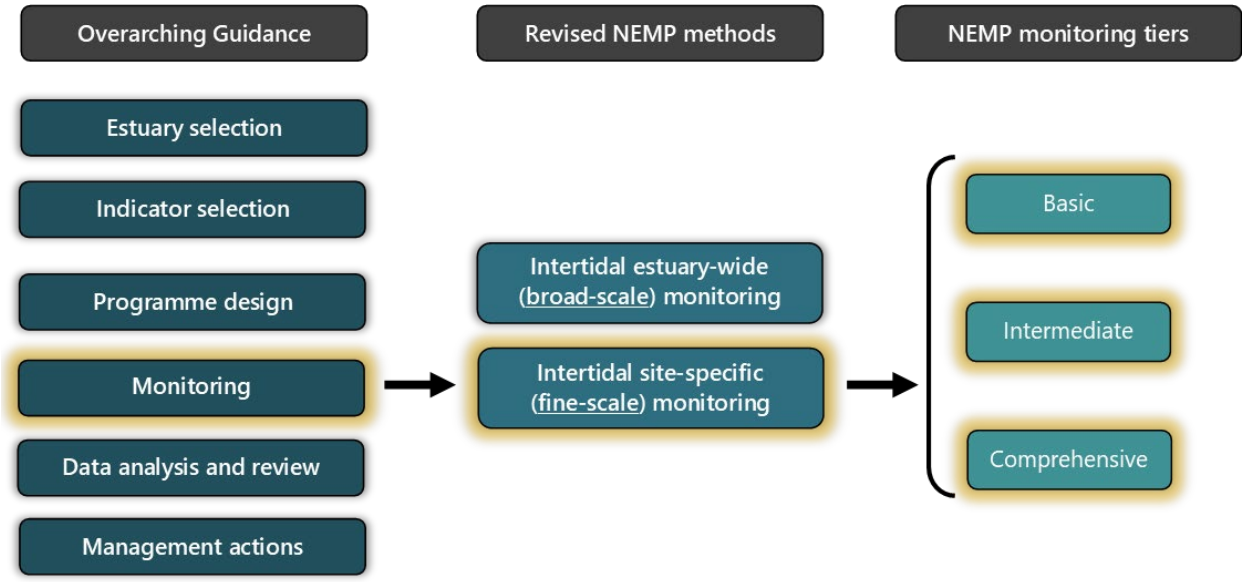


Fig. 3. Illustration of the links between the *Overarching Guidance* and this fine-scale method document.

Estuary condition, and the potential for adverse ecological effects, can be inferred through the comparison of sediment quality results to thresholds of ecological health such as described in a recent review for MfE [6]. As for the Basic tier, changes in state, or declining trends, may trigger Comprehensive monitoring or more detailed investigations.

3.2.3 Comprehensive

The Comprehensive tier includes the full suite of indicators (Table 2). Compared with Intermediate, it adds sampling of macroinvertebrates and microalgae as quantitative ecological indicators. It also provides the option to add quantitative quadrat sampling of epifauna and macroalgae.

The Comprehensive tier is intended to support robust sampling across a broad range of indicators that influence or directly measure ecological condition. The rationale is that a using the full suite of indicators enables a 'weight-of-evidence' approach to understanding estuary state and temporal trends.

Table 2. Example of a tiered fine-scale (FS) monitoring approach, illustrating the expansion of indicators from Basic to Comprehensive. Effort additive to a previous tier is indicated with a plus (+). Sample replication is discussed in Section 3.3.

Site-specific indicators		**Basic**	**Intermediate**	**Comprehensive**
Key elements of each tier		Grain size & qualitative FS site indicators	+ Quantitative FS sediment quality & sedimentation	+ Quantitative macroinvertebrate cores and other biota indicators
Sedimentation				
Sediment plates		*Not sampled*	✓	✓
Sediment quality				
Qualitative enrichment[1]		✓	✓	✓
aRPD		✓	✓	✓
Grain size (%mud, sand & gravel)		✓	✓	✓
TOC		*Not sampled*	✓	✓
TN & TP[2]		*Not sampled*	✓	✓
Toxicants (e.g., metals)[3]		*Not sampled*	✓	✓
Sediment biota				
Macroinvertebrates		*Not sampled*	*Not sampled*	✓
Epifauna	SACFOR[4]	*Not sampled*	✓	✓
	Quadrats (optional)[5]	*Not sampled*	*Not sampled*	✓
Macroalgae	SACFOR[4]	✓	✓	✓
	Quadrats (optional)[5]	*Not sampled*	*Not sampled*	✓
Microalgae	SACFOR[4]	✓	✓	✓
	Cores[6]	*Not sampled*	*Not sampled*	✓

[1] Qualitative enrichment status assessed based on sediment colour, odour & micro-organism growth, as per methods in Section 5.4.2.

[2] Total Phosphorus (TP) may be dropped from long-term monitoring if results show high collinearity with Total Nitrogen (TN) or Total Organic Carbon (TOC).

[3] Toxicants can be added as relevant to the estuary context (e.g., semivolatile organic compounds could be included in urban environments).

[4] SACFOR is a site-wide ordinal classification method for epibiota abundance or cover, as described in Section 5.5.2.

[5] Field-based quadrat assessment of epifauna abundance, and percent cover of macroalgae, is included as an optional method. For macroalgae, photoquadrats can also be taken for post-field percent cover quantification.

[6] Core sampling for microalgae is for analysis of chlorophyll-a and phaeopigments. Microalgae extent in eutrophic systems can also be assessed using qualitative enrichment criteria (as per footnote 1).

3.3 SAMPLE REPLICATION

3.3.1 Overview

The recommended level of sample replication for each indicator is summarised in Table 3, and the rationale detailed in sub-sections below.

Table 3. Recommended sample replication.

Indicator	Replication
Sediment plates	4 sediment plates[1]
Sediment grain size	3 composite samples[2]
Sediment quality (TOC, TN, TP, metals)	3 composite samples[2]
aRPD	Next to each sediment quality sub-sample[2]
Macroinvertebrates	10 cores
Epifauna & macroalgae SACFOR	Semi-quantitative measure across whole site (i.e., no replication)
Epifauna & macroalgae quadrats	10 x 0.25m^2 quadrats
Microalgae (chlorophyll-*a* & phaeopigments)	3 composite samples[2]

[1]The number of replicate measurements taken on each sediment plate varies depending on the method of measurement; see Section 5.

[2]Each sediment sample should be a composite of 4 sub-samples; see Section 5.

3.3.2 Sedimentation

Site-specific sedimentation, measured using the buried plate method, is a new indicator for the NEMP update, and there have been no national analyses of replication needs. Much of the method development and ground-work in this space has been conducted by Waikato Regional Council (WRC) [36].

In the WRC programme, various plate layout configurations have been used, but site-scale sedimentation rate is generally determined from 4 to 6 replicate plates. Other councils use varying plate configurations with generally 2-4 replicate plates per site [37-40].

Pending a national method review, it is suggested that 4 replicate plates be adopted as a minimum. As this is a new NEMP indicator, Section 5.3 details the sediment plate method and options for plate configurations at fine-scale sites.

3.3.3 Sediment quality

Monitoring results have shown that among-sample variance in laboratory-measured sediment quality analyte values is typically quite low within sites having uniform substrates and no obvious gradient in sediment type. Consequently, most councils and providers use a composite sampling approach for fine-scale sediment quality as a cost-effective alternative to the high-replication discrete sample collection (within each plot) that was recommended in the original NEMP (Appendix 1) [16, 29, 41, 42]. For homogeneous sites, a reliable measure of sediment quality can be achieved by collection of three composite samples within each of 3 columns of the 3x4 site layout schematic in Fig. 2. For a given column, a composite sample should consist of a sub-sample collected from within each plot (see Section 5). As aRPD can be relatively variable, measure aRPD depth next to each sediment quality sub-sample.

3.3.4 Sediment biota

Macroinvertebrates

Sample replication is a key consideration for macroinvertebrates. The risk in under-sampling is that low abundance species may not be detected, which can confound interpretation of spatial differences or temporal changes in metrics of species richness and community composition.

Hewitt (2021) [7] has proposed a guideline of 12 cores per site based on long-term data for Manukau Harbour, considering two factors: the number of samples needed to start approaching an asymptote of species detection, and the power to detect change. Similar methods (i.e., power analysis, species detection, and the coefficient of variation approach of the original NEMP) have been used to determine macroinvertebrate sampling sufficiency in other New Zealand estuaries where fine-scale sampling is undertaken. These analyses have shown that sampling effort in some estuaries/sites could be reduced to 9 cores without any substantive loss of ability to detect long-term changes, but identified that 10-12 cores was preferable in other instances [e.g., 41, 43-47].

For national comparisons there is a benefit in having consistent macroinvertebrate core sample replication, as species-richness and compositional analyses are linked to sampling effort (i.e., taking additional cores at most estuary sites would likely yield additional species).

A national analysis of sampling sufficiency would clearly be useful. In its absence, on balance it is suggested that sample replication for macroinvertebrates remains at a minimum of 10 cores per fine-scale site as

recommended by the original NEMP. This effort reflects the point where the species detection curve approaches the asymptote [e.g., 41, 45, 47], and aligns with what many councils already undertake (Appendix 1). That said, councils with established fine-scale datasets have the option of determining specific replication needs for their estuary sites [e.g., 41, 45]. There may be a case to reduce replication in situations where the focus is on characterising abundant indicator species that can be reliably sampled with less effort. However, it was beyond scope to consider this approach.

Epibiota

For the epibiota indicators, the recommended replication in Table 3 for discrete sampling (i.e., methods other than SACFOR) is provided as a rough guide only. Analysis of existing datasets may enable refinement of this guidance.

Quadrat methods (epifauna, macroalgae): For the optional component of quantifying epibiota in 0.25m^2 quadrats (as an addition to SACFOR) the recommendation of 10 quadrats in Table 3 is based on the original NEMP. However, due to the patchy or clumped nature of some epibiota, 10 quadrats may be insufficient, and may fail to detect species with very low abundances.

Microalgae: The recommendation in Table 3 to collect 3 composite samples for analysis of chlorophyll-*a* and phaeopigments is aligned with the sediment quality sampling approach. The expectation is that compositing will average across some of the considerable spatial variability that is expected in these microalgal indicators, noting that expert judgement will be needed to ensure representative samples are collected.

3.4 GUIDANCE ON MONITORING FREQUENCY

Monitoring frequency needs to be linked to monitoring purpose and programme design, and is also dictated by natural variability in monitoring indicators, and the time-scales over which changes in pressures and responses are expected.

Sedimentation

Sedimentation is measured at least annually by most councils. For example, the analysis of long-term data by Hunt (2019) [36] suggested that sedimentation should be measured at regular annual intervals (to avoid seasonal confounding) which, over at least a 10-year time-scale, would provide reliable estimates of sedimentation and trends. Sedimentation patterns and drivers can also be informed by event-related sampling (e.g., flood sediment deposition) in addition to routine annual monitoring [48, 49].

Sediment quality and biota

The frequency of fine-scale monitoring of sediment quality and biota varies considerably within and among councils in New Zealand [16], largely due to differences in council management goals, monitoring objectives, and resource constraints.

For councils with limited resources, fine-scale monitoring has typically comprised establishing a contemporary 'baseline' of three consecutive annual surveys, followed by ongoing monitoring at intervals of ~5 years [16]. This interval is arbitrary, and usually reflects council resourcing. In this situation, assessment of estuary condition relies primarily on interpreting indicator values against ecological condition thresholds. However, it may require decadal time-scales for a reliable picture of temporal trends to emerge that can be linked to environmental drivers. Additionally, important determinants of estuary condition may change across shorter time-scales meaning more frequent monitoring is more appropriate. Examples include anthropogenic influences such as sediment release from forestry logging [50], and climatic phenomena such as marine heatwaves and the El Niño Southern Oscillation [7, 30, 51].

Accordingly, councils that have greater resources tend to undertake fine-scale assessments annually, or even more frequently [e.g., 7, 41]. The benefit of frequent monitoring is trend detection over short time-scales (i.e., ~10 years). Based on experience in Auckland, Hewitt [7] recommended monitoring at least twice per year (and up to six times per year) for 15 years to detect trends and tipping points with macroinvertebrate indicators. In the Waikato region, a time-series of ~10-years of annual monitoring was recommended for trend detection for macroinvertebrate indicators [36, 41]. Other councils have also shown the benefits of regular (annual or near-annual) sampling in terms of trend detection [52-54].

Note! The following minimum monitoring intervals are recommended:

Trend detection: monitoring at least annually around the same time of year, with a goal to link trends in indicators to changes in environmental drivers.

State assessment: monitoring no less frequently than every 5-years around the same time of year, with a goal to intermittently characterise estuary state relative to ecological condition thresholds.

3.5 ILLUSTRATION OF A MONITORING APPROACH

Fig. 4 illustrates how the various tiers in Table 2 might be integrated into a monitoring programme, according to estuary monitoring priorities (see *Overarching Guidance*) and purpose. The approach illustrated is based on progressive broadening of the indicator suite and increasing the monitoring frequency as the priority for monitoring increases.

High priority 'sentinel' estuaries could be monitored annually, using the full suite of monitoring indicators represented by the Comprehensive tier in Table 2. The purpose would be the timely detection of trends that showed declines in ecological state.

For **Medium** priority estuaries, a nested approach could be considered in which a few key indicators (e.g., from the Basic or Intermediate tier) were measured annually to facilitate trend detection, with the Comprehensive tier and associated biota indicators implemented periodically (e.g., every 5 years) to assess state. State assessment could be achieved through evaluation of results against condition thresholds for ecological health.

For **Low** priority estuaries, the Basic monitoring tier could be implemented periodically (e.g., every 5 years) to maintain a check on key indicators of concern.

In all scenarios, significant changes in state may trigger targeted investigations, and/or a shift to more in-depth monitoring. Additionally, frequent Comprehensive monitoring in sentinel estuaries would provide contextual information that may help interpret the results arising from less intensive monitoring of lower priority systems [e.g., 7].

Councils could also consider a hybrid approach whereby different monitoring tiers were applied within some estuaries. For example, monitoring in high priority sentinel estuaries could consist of:

- Comprehensive monitoring of fine-scale sites on the main tidal flats.
- Basic or Intermediate monitoring in estuary side arms vulnerable to catchment-related physical changes, but where the biota may be resilient to such stressors (i.e., potentially negating the benefits of monitoring the full suite of biotic indicators).
- 'Meso-scale' approaches which, in a fine-scale context, could consist of linking fine-scale sites with sampling transects that transition from 'healthy' to 'degraded' conditions, along which some key indicators could be measured (e.g., sediment mud content).

With many potential options and permutations available, it is recommended that councils think carefully about their monitoring purpose when designing their estuary programmes (see *Overarching Guidance*). Some relevant considerations are:

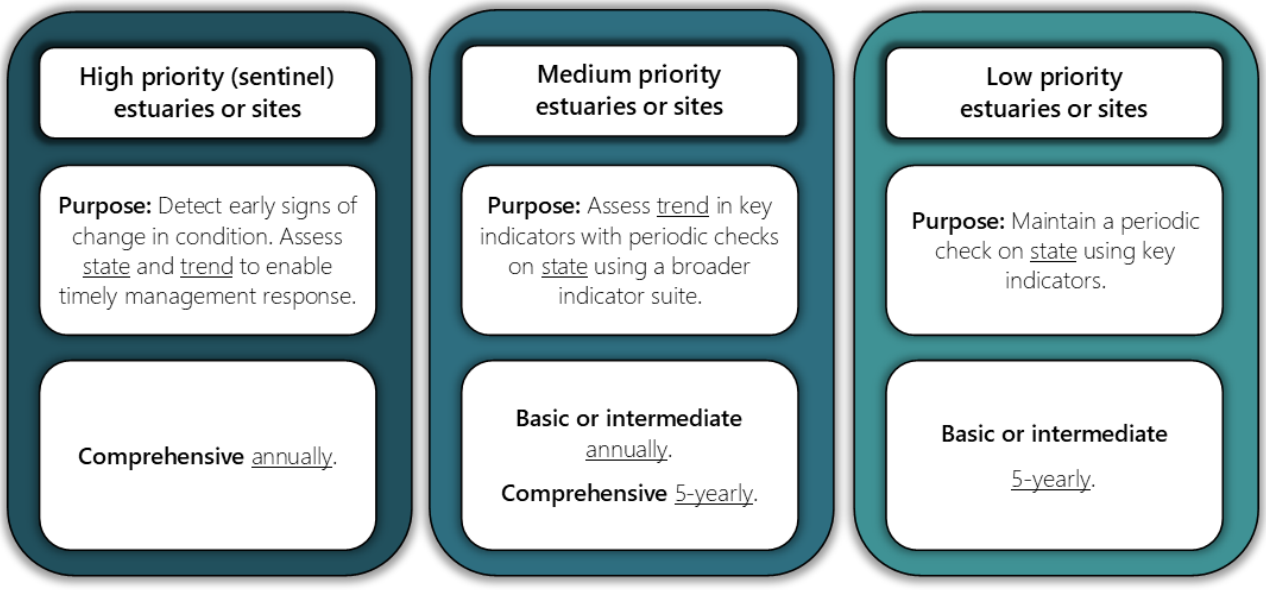


Fig. 4. Illustration of a regional sampling programme that combines the scaling of indicators (according to the monitoring tiers in Table 2), different monitoring frequencies, and different estuary priorities.

- If fine-scale sites are established only on the intertidal flats of the main estuary body (e.g., well-flushed mid-estuary areas), more degraded or susceptible areas may be missed, and immediate links to catchment pressures may not be evident.
- Targeted monitoring of susceptible areas would better inform councils about the more degraded parts of an estuary and, along with data on potential drivers of estuary condition (e.g., catchment activities), facilitate understanding of changes in state due to changes in pressures.
- Targeted monitoring of susceptible or degraded areas may also be used to inform councils about the long-term efficacy of management interventions (e.g., improvements in estuary condition due to source reductions of sediment and nutrients).
- If the goal was for fine-scale monitoring to be representative of overall estuary state, intensive sampling (i.e., reflecting the Comprehensive tier) would need to be implemented at numerous sites, which would likely be beyond the resources of many councils.

Accordingly, the hybrid approach described above, in which different tiers of monitoring are applied at different sites within priority estuaries, linked by 'meso-scale' monitoring along transects, may be a more realistic way of capturing estuary-wide condition.

Fig. 5 illustrates one of several examples where site-specific sampling of the full suite of NEMP fine-scale sediment quality and biota indicators was undertaken in representative estuary habitats that were identified by broad-scale surveys [e.g., 55, 56-58]. These were low-replication pilot surveys, based on collection of single composite sediment quality samples and triplicate macrofauna cores, from discrete sites. Such approaches provide a broad overview of estuary condition by combining the indicators used in both the broad-scale [4] and fine-scale protocols. Although macrofauna replication was low in the example depicted in Fig. 5, the dominant taxa were captured. A secondary purpose of that work was to identify potential fine-scale sites for ongoing Comprehensive monitoring, but the approach could similarly be used to inform the development of a within-estuary tiered monitoring design.

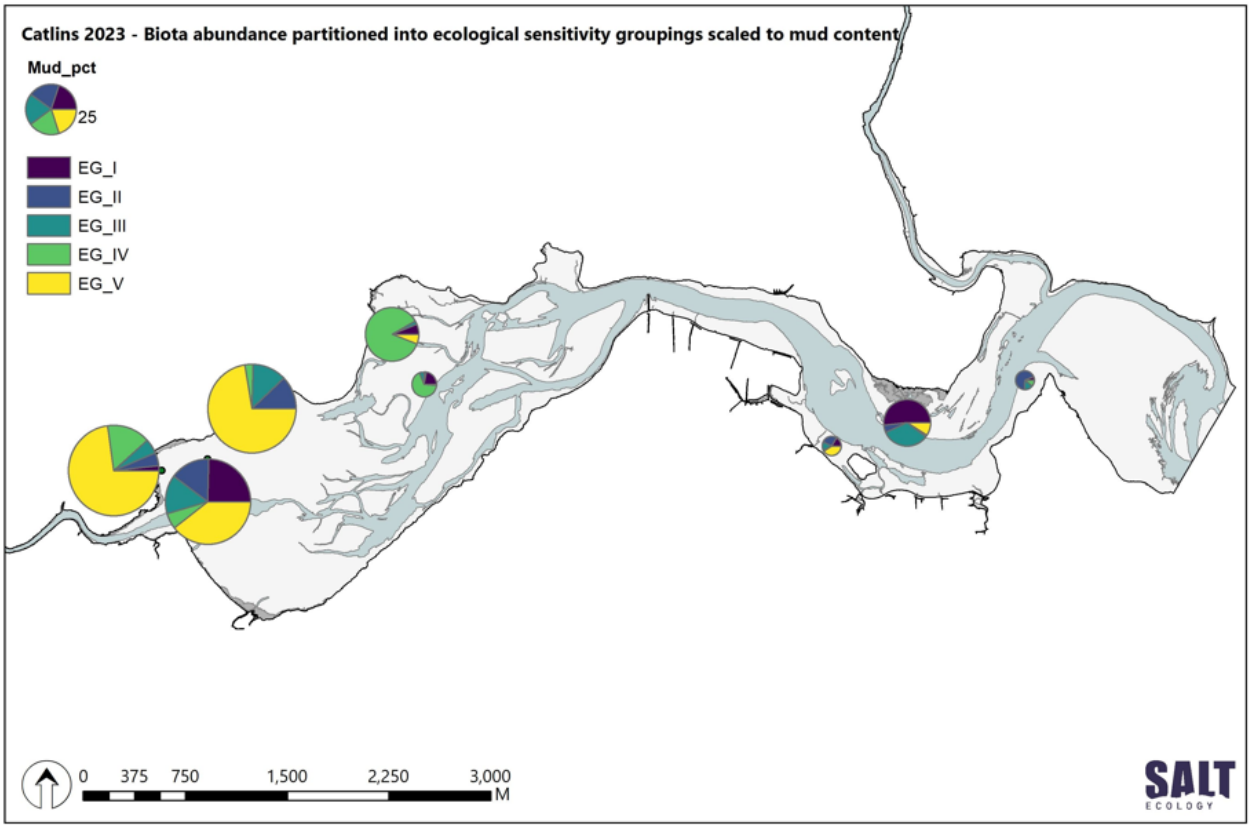


Fig. 5. Example of Catlins Estuary, Otago, where broad-scale mapping was supplemented by sampling of fine-scale indicators to better characterise estuary condition and inform the suitability of locations for establishing fine-scale sites. The upper estuary macroinvertebrates were characterised by 'hardy' species (eco-groups IV and V) resilient to a high sediment mud and other stressors.

4. BROADER CONSIDERATIONS FOR FINE-SCALE MONITORING

Once estuaries have been prioritised, and the monitoring purpose determined, additional considerations for the fine-scale approach include:

- The number of fine-scale sites that are needed.
- Where to position sites so that they are effective for long-term monitoring.
- When to undertake monitoring.

4.1 HOW MANY SITES?

The number of fine-scale sites will depend on monitoring purpose, programme design, and budget. To date, many councils have followed the original NEMP guidance (or some variant of it) by setting up 2-4 sites in each estuary on the main tidal flats. Usually, a greater number of sites are placed in larger estuaries.

Application of a tiered approach, such as described above, may enable a greater number of estuaries to be monitored, a broader representation of habitat types within priority estuaries to be included, or sampling designs that target specific issues (e.g., stratification within estuaries to include sub-catchments with and without exotic forestry logging) to be incorporated.

4.2 SITE SELECTION AND LOCATION

A clear purpose for fine-scale monitoring, combined with knowledge of estuary habitats determined by broad-scale mapping, a pilot survey, and/or expert opinion, will highlight (in general terms) where fine-scale sites could be established. When deciding exactly where to position sites, some key matters need to be accounted for, which are outlined below.

Where existing monitoring sites are sub-optimal, councils will need to decide whether to relocate them (and lose the time-series that has been established) or add new/improved sites (if available) to their programmes. It is unfortunate to end established time-series, but also futile to continue monitoring sites that may be unsuited to fine-scale monitoring for reasons such as outlined below.

Site utility for specific indicators: A preliminary (pilot) survey of possible sites (such as illustrated by Fig. 5), will help identify their suitability for long-term monitoring. Pilot surveys could include measurement of field-based indicators, or sample collection for laboratory analyses, depending on purpose, budget and time-frames. For example, where macroinvertebrates are to be used as a monitoring indicator, it is important to choose sites that have a sufficiently diverse array of species present, ideally including those likely to be sensitive to catchment-related and other stressors. It may be sufficient to inspect sieved macroinvertebrate cores in the field, or samples may need to be sent to a taxonomic laboratory for identification.

Field inspection of macroinvertebrate samples can inform the suitability of sites for fine-scale monitoring.

Habitat stability: Unstable habitats such as mobile sands, or estuary areas subject to excessive physical disturbance (e.g., from tidal or river flows, or waves), should be avoided for several reasons, including: (i) their macroinvertebrate assemblages can be impoverished and/or dominated by opportunistic species that are resilient to stress; (ii) sediments can variably erode or build-up depending on hydrodynamic conditions; and (iii) grain size characteristics can be highly variable, affecting macroinvertebrates and other sediment quality indicators. Accordingly, the influence of catchment and other pressures can be masked by natural pressures.

Salinity influences: Habitats near river channels, or in inner estuary areas that are subject to regular or episodic low-salinity conditions, can have impoverished and hardy macroinvertebrate assemblages that are relatively insensitive to changes in anthropogenic pressures. Sampling in such locations is useful for characterising the range of estuary habitats and their condition, but may be of lower value for long-term monitoring.

Tidal elevation: The original NEMP advocated establishing fine-scale sites in the low-mid tide zone where the period of drying at low tide is minimised, flushing with seawater is high, and salinity variation from

freshwater is minimised. However, low-mid tidal flats can be physically unstable (e.g., mobile sands) and therefore unsuitable for monitoring, or logistically difficult to access in a narrow tidal exposure window. In addition, in some estuaries the main tidal flats are 'perched' above mid-tide. As such, it may be necessary – even though not ideal – to consider establishing sites relatively high in the tidal zone, where habitat conditions may be less suitable for macroinvertebrates [59]. In such situations, a pilot survey would be advised before setting up a fine-scale site, especially if the intent was to monitor using the Comprehensive tier. To help select areas to sample, tidal elevation can be estimated from LiDAR datasets [60, 61].

Estuary typology: The fine-scale method is mainly suited to monitoring shallow intertidal dominated estuaries (SIDE), which have extensive, stable tidal flats, enabling sites to be positioned away from flow channels.

In river-dominated estuaries (e.g., tidal river estuaries), it is reasonably common for intertidal sites to experience physical scouring by water flow, or large-scale sediment accretion/erosion due to the mobilisation and redistribution of bed sediments. Similarly, intertidal habitats in such systems can be small in area and subject to extended inundation by low-salinity water. Hence, anthropogenic effects can be difficult to discern as described above. Moreover, monitoring sites and equipment (e.g., sediment plates) can be lost during flood events, compromising long-term data collection.

The fine-scale indicators described in this document can be used to characterise these river-dominated systems, but establishing and monitoring long-term fine-scale sites may not be the best approach. Alternative or complementary monitoring approaches for tidal-river estuaries have been described in a recent MfE report [62].

Fine-scale sites positioned on channel margins are usually susceptible to physical disturbance from tidal or river flows which, along with marked variability in substrate composition and tidal elevation, can make anthropogenic effects difficult to discern.

Tip! When picking locations for fine-scale monitoring sites, some 'rules of thumb' are:

- At least 50m (further if possible) from main tidal channels.
- Away from areas where there is strong evidence of flood deposition (e.g., accumulated terrestrial debris).
- Across a uniform tidal elevation and homogeneous sediments within the mid-low tide zone, where possible.
- In stable sediments, avoiding areas of rippled mobile sand.
- Away from significant freshwater influences, to the extent feasible.

Ideally, undertake a pilot survey of key indicators to determine suitability for monitoring. For example, consider whether sites have a sufficient range and abundance of macroinvertebrate species that will be sensitive to degradation in habitat quality.

Site substrates: Substrate types may range from mud-dominated to sand-dominated. When sampling in sandy habitats, the stability of the substrate needs to be considered for reasons described above. When sampling in substrates that are already muddy, the sensitivity of the habitat to increased stressors (e.g., increased catchment sediment loads) may be difficult to discern in ecological terms. Hence, even where there is a measurable physical change (e.g., an increase in mud content in sediment that is already mud-dominated), the ecological consequences may be less obvious than in sandy sediments.

Site vegetation: The NEMP is designed for sampling in unvegetated habitats, but some estuary monitoring programmes already include sites in seagrass or sites that have become covered in opportunistic macroalgae such as *Gracilaria* spp. When applying the NEMP in these scenarios, it is important to consider whether to sample in bare patches between vegetation or within the vegetated beds. Where macroalgae is extensive, an additional consideration for macroinvertebrates is whether to collect whole samples of macroalgae and sediments for combined analysis, whether to split whole samples into macroalgae and sediment components for separate analysis, or to sample and analyse macroalgae and sediment from separate patches.

For example, eutrophic sites in New River Estuary (Invercargill) have very high abundances of certain amphipods in dense surface growths of opportunistic *Gracilaria* spp., despite the presence of near-surface

anoxia in sediments beneath [63]. Hence, the choice of sampling approach can have a significant bearing on results, whereby sediment quality values can indicate highly degraded conditions, but overlying macroalgae can support sensitive taxa that are indicative of relatively healthy conditions.

In New River Estuary (Invercargill), monitoring of eutrophic fine-scale sites that are covered in extensive mats of *Gracilaria* spp. requires a consistent approach to sampling, and decisions on whether to collect cores from dense macroalgae beds, spaces between beds (if they exist), or to split samples into overlying macroalgae and underlying sediment.

4.3 TIMING OF MONITORING

The original NEMP recommended that field surveys be carried out between January-March. In practice most councils and providers appear to have undertaken surveys throughout late spring to early autumn in conjunction with broad-scale monitoring, which commonly targets the period of peak macroalgae growth [4]. Guidance for calculation of National Benthic Health Model (BHM) scores is that macroinvertebrate data should be collected over October to March, if possible, to reduce the influence of seasonal changes on model results [64].

Hewitt et al. 2014 [65] and Hewitt 2021 [7] noted that it may be most cost-effective to conduct annual fine-scale surveys in spring (September to November) when macroinvertebrate recruitment tends to be relatively low. The benefit of conducting sampling during low recruitment periods is not only that analysis costs are lower, but that variability in recruitment (driven by natural cycles rather than anthropogenic factors) doesn't impact analyses of species abundances or community composition.

Councils implementing monitoring in new estuaries may therefore benefit from conducting surveys in low recruitment windows. However, for established programmes, a more important consideration is to conduct repeat sampling at roughly the same time of year as previously, to ensure consistency. Recruitment pulses can be addressed as part of the interpretation of results or, alternatively, unidentified juveniles can be excluded from analyses, as is standard procedure for the derivation of biotic indices such as AMBI [66] and the BHM [67].

Irrespective of the ideal timing, on the basis that different councils and providers already undertake sampling at different times of year, the possibility of seasonal differences should be accounted for when comparing results from different estuaries or regions.

Extensive intertidal flats of Freshwater Estuary, Rakiura.

5. FINE-SCALE METHODS

5.1 OVERVIEW

This section describes the sampling methods for implementing the fine-scale methodology, and covers the following:

- Fine-scale site layout.
- Options for sediment plate configurations, and plate installation methods.
- Methods for the sampling and analysis of the indicators summarised in Table 1 of Section 2.
- Metadata requirements, and key considerations for QA/QC, data handling and data analysis.

An example equipment list for a typical fine-scale survey is provided in Appendix 2.

5.2 SITE LAYOUT

A similar site layout to that described in the original NEMP should be adopted, as follows:

- Chose sites in uniform/homogeneous intertidal areas, adhering to the selection guidance described in Section 4.2.
- Aim for a minimum site area of 15x30m, to provide sufficient space for repeat sampling. If an area of this size is not available (e.g., on narrow intertidal margins of tidal river estuaries), it may mean that the location is unsuitable for fine-scale monitoring.
- As described in Section 2, divide each site into a 3x4 (column x row) grid of 12 sampling plots. Fig. 6 below illustrates an example in Otago. Retaining a 3x4 plot layout maintains consistency with the original NEMP, enables the Comprehensive monitoring tier to be implemented, and caters for councils or providers that take 12 macroinvertebrate cores per site.

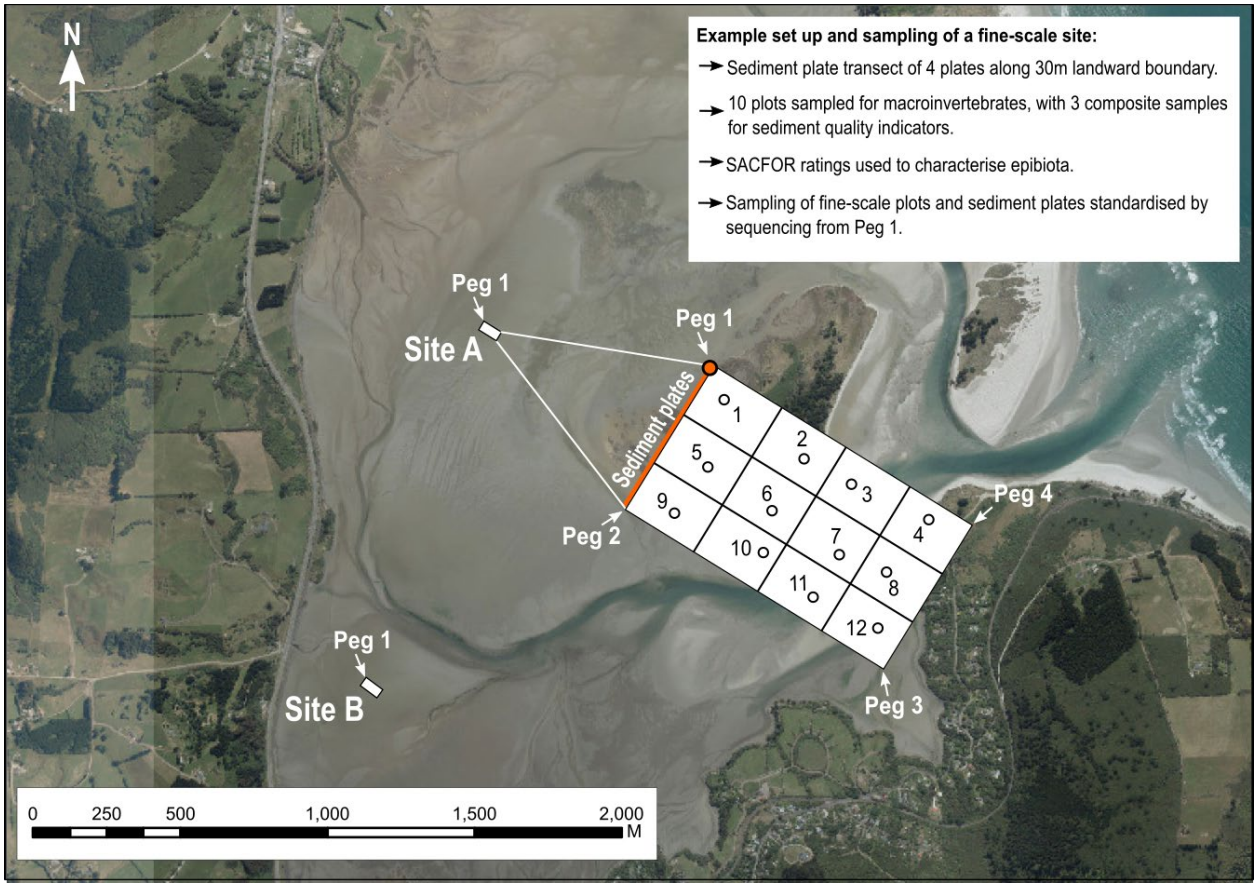


Fig. 6. Example of a fine-scale site set-up and sampling approach, based on two 30x60m sites (white rectangles) established in Blueskin Bay, Otago. The numbered rectangle represents an enlargement of Site A.

Ten of the 12 plots are sampled for macroinvertebrates, but sediments (n=3) are composited across each of the three columns in the grid (i.e., across 1-4, 5-8, 9-12). Plot numbering and sediment plate sampling starts in the corner labelled Peg 1. Note that each site is in a homogeneous sandy habitat with no appreciable change in tidal elevation, hence the sediment plates (depicted by the orange line) are arranged along the landward 30m boundary. The pros and cons of different sediment plate configurations are described in Section 5.3.2.

- Configure and sample plots using a standard numbering sequence, such as shown in Fig. 6, to help with consistent implementation (e.g., to ensure sediment plates are sampled in the correct order).
- Mark site corners with 400mm long H4 treated wooden pegs driven 300mm into the sediment (leaving 100mm visible), and log with a GPS waypoint. During field sampling, bamboo canes with flagging tape can be used to temporarily mark plot corners as needed.

5.3 SEDIMENTATION METHODS

5.3.1 Overview

Fine-scale sedimentation monitoring involves burying 'sediment plates' as a means of monitoring sedimentation rate (sometimes called sediment accumulation rate; SAR) by measuring the change in sediment depth on top of the plate. This method, developed by Waikato Regional Council [36], is now widely used nationally. Concrete pavers (e.g., 19x23cm) are an inexpensive and practical option to use as sediment plates. As per Section 3.3.2, at least 4 sediment plates should be installed at each site.

5.3.2 Plate layout

Plate layout configurations at fine-scale sites vary among regions. The most commonly-used plate arrays are as follows:

- Plate(s) at opposite ends of fine-scale sites (sometimes outside the site boundary). This type of configuration is useful where there is a tidal elevation gradient across the site, as water depth can influence hydrodynamics and rates of sedimentation [36, 40].
- Plates arranged along a transect on one boundary of a fine-scale site [38]. This method is best suited to homogeneous sites with reasonably uniform substrates and no appreciable gradient in tidal elevation.

The main consideration for plate layout is that sedimentation measurements are representative of the depositional environment that the site biota is exposed to. Suggested configurations that reflect the above two approaches are shown schematically in Fig. 7. Layouts similar to Configuration 1 are preferable as a default, in that the wide distribution of plates across a site will, in theory, capture spatial variation in sedimentation in most scenarios. However, Configuration 2 is acceptable where site features are sufficiently uniform that a gradient in sedimentation is not expected, or the gradient is captured across the alignment of the transect (e.g., where a site slopes towards a channel).

The main reasons for not positioning plates inside the fine-scale sites are: (i) they could easily be trodden on and disturbed during fine-scale sampling unless their exact location was marked and avoided during field work; (ii) if frequent sediment plate sampling is undertaken it would lead to considerable physical disturbance within the fine-scale site; and (iii) plate installation around the site perimeter avoids the need for additional marker pegs (unless differential GPS is used for plate relocation).

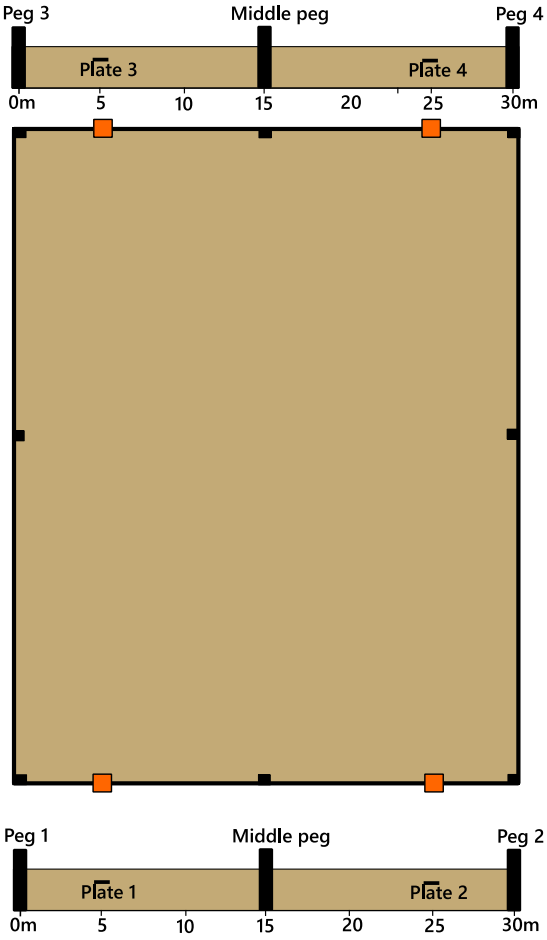
Note that a different plate installation configuration may be used when measuring sedimentation in locations other than at fine-scale sites – see example on p. 16.

5.3.3 Installing plates

Installing plates 2m or more from marker pegs should ensure that hydrodynamic changes due to the pegs do not influence the buried plates. The method below involves installing each sediment plate on top of a short (400mm) steel waratah to help with stability. If marker pegs were lost, the waratah would also enable relocation using a metal detector. The basic installation sequence is as follows:

1. Measure each plate installation point (e.g., relative to a peg), and mark with a 400mm waratah.
2. Hammer (or push) each waratah vertically until flush with the sediment surface.
3. Dig a 100-150mm deep hole around each waratah wide enough to fit a paver horizontally. In stable sediment, a target plate depth of 100-150mm is usually sufficient, but some studies recommend installing plates deeper [36].
4. Hammer the waratah until it is flush with the bottom of the hole, then centre the paver on top of the waratah. Remove or repack sediment beneath the paver, tapping it down with a mallet, until it is stable (i.e., not wobbling on top of the waratah).
5. Using a spirit level, ensure the plate is level, repacking sediment and tapping down as required.
6. Backfill the hole and level the excavated diggings with the surrounding sediment.
7. Record plate and peg locations using GPS (differential GPS can provide ±1cm accuracy).

Configuration 1. Suited to all fine-scale sites, but especially where gradients in substrate or tidal elevation mean that sedimentation may be highly variable across the site. Option to redistribute the 4 plates with one along each side of the site, or increase replicates and add two plates along each of the long boundaries.



Configuration 2. Best suited to sites with a homogeneous substrate and tidal elevation, such that sedimentation is expected to be relatively uniform. This transect alignment would capture left-to-right gradients, for example if the left side of site sloped towards a channel. Option to increase replicates or restribute plates, as described for Configuration 1.

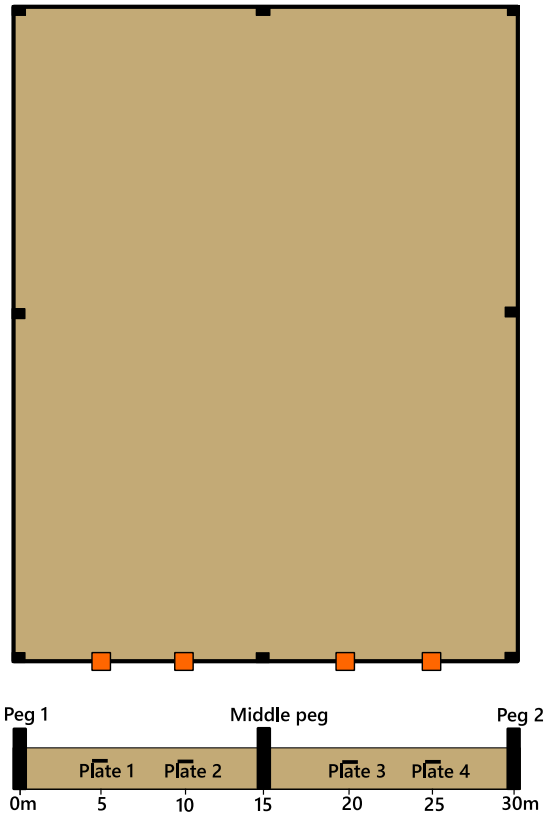


Fig. 7. Schematic of possible sediment plate (orange squares) configurations at fine-scale sites.

Both configurations assume 4 replicate plates are installed as a minimum. Configuration 1 is preferred as it will likely cater for most scenarios, with the plates widely distributed so that they capture site-scale variation in sedimentation. Configuration 2 provides an acceptable alternative where sites are located in areas of uniform substrate and tidal elevation, although the transect alignment can nonetheless be configured to capture a directional gradient (e.g., towards a channel).

Setting up a site for a transect sediment plate installation using Configuration 2 above.

Final leveling of a plate using a spirit level.

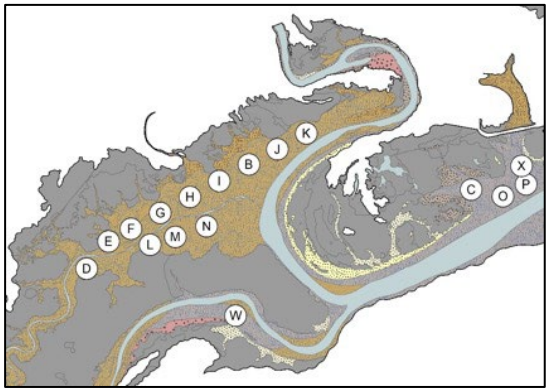
Other sediment plate configurations

In addition to its use in fine-scale monitoring, sedimentation monitoring can be used to target specific areas of interest in either a site-based configuration (as described in the text), as a transect across transitional zones, or as spot measurements across a larger spatial area.

The principles of installation and measurement are the same as described in the text. However, some additional considerations are as follows:

- Where possible, plates should be installed in a transect line or an organised configuration with common distances between pegs and/or plates (e.g., each peg is 50m apart and each plate is ≥2m from the peg).
- The tidal elevation of each plate should be estimated (e.g., based on LiDAR) to account for sedimentation differences due to water inundation depth and associated factors (e.g., disturbance due to waves).
- Composite grain size samples should be collected next to each plate, to assist with understanding changes in deposited sediments across the monitored area.

Below is an example from Akatore Estuary, Otago where the sediment plate configuration was designed to assess sedimentation and grain size changes expected from forestry harvest.



5.3.4 Measuring plate depth

There are two methods that can be used for measuring paver depth, as illustrated in Fig. 8 and photos on p. 17.

Method 1: 'Straight edge'

Use a 'probe' such as a fibreglass stock rod to locate each buried paver. Measure the depth from the paver to the bottom of a straight edge (e.g., 2.5m length of T-section aluminium). Ensure the straight edge level is representative of the surrounding sediment, e.g., not inadvertently placed on raised shell or on drift algae.

At a minimum, triplicate measures should be made at each plate so that average plate depth and variance can be calculated. Reposition both the probe and the alignment of the straight edge for each replicate measure. The use of a straight edge smooths out depth variance caused by small undulations in the sediment surface. Where plates are positioned at fixed distances from a marker peg, a straight edge can substitute for a tape measure when relocating plates. For example, a 5m distance from a peg to a plate can be measured at two lengths of a 2.5m straight edge.

Method 2: 'Knitting needles' (or small rods)

At each plate location, use knitting needles or similarly small rods to locate the buried paver. Measure the rod depth from the plate to the sediment surface. This method was developed by Waikato Regional Council [36]. The approach measures plate-scale sediment heterogeneity that is smoothed over when using a straight edge (Method 1). For example, small scale depressions or mounds caused by biota (e.g., burrowing crabs) will affect the estimate of paver depth. As such, a minimum of 10 replicate knitting needle measurements should be made per plate [36, 40].

Note! A paired comparison of the straight edge (Method 1) and knitting needle (Method 2) approaches indicates differences of up to ~4mm to a measured plate depth. As such, the method chosen at the start of monitoring at a given site should be used consistently at that site in subsequent surveys.

For both methods, an atypical depth measurement across a plate may indicate that the probe has hit hard debris (e.g., shell), or the measurement has been incorrectly recorded. High variance across plate replicates may also indicate that the paver has become tilted (e.g., it may have moved after initial levelling).

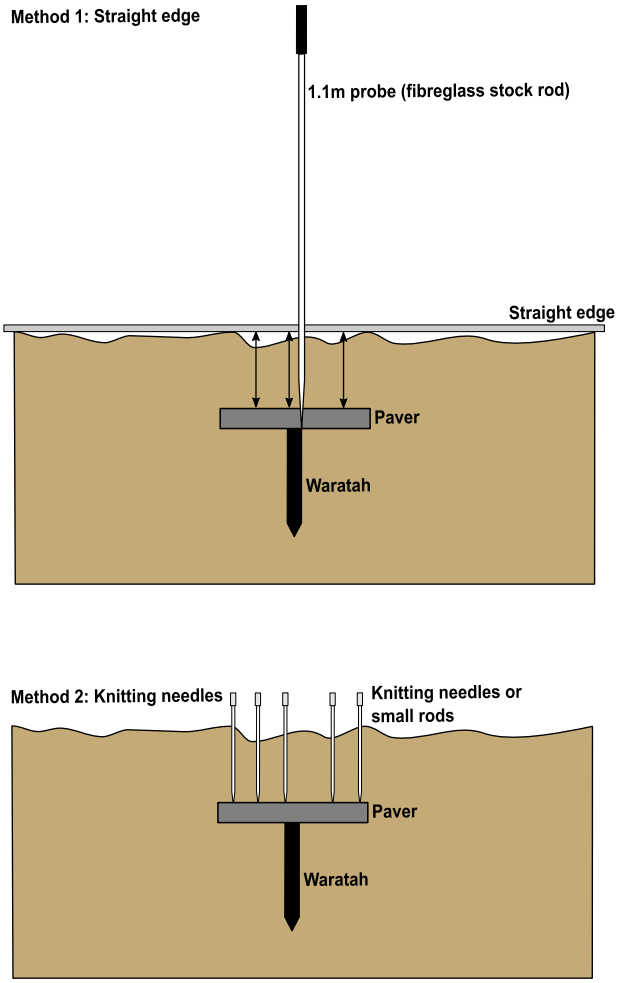


Fig. 8. Schematics of sediment plate methods.

Top: The 'straight edge' method smooths across surface undulations, meaning the main variance across a paver will be due to realignment of the straight edge for each subsequent measure, or by tilt if the paver was not installed level or has been disturbed.

Bottom: The 'knitting needle' method measures plate-scale surface undulations, meaning there may be considerable variance across a plate (depending on the features of the sediment surface), in addition to variance introduced by factors such as paver tilt or disturbance.

Measuring a buried sediment plate using knitting needles.

When using a straight edge where there is surface shell, make sure it contacts the sediment surface and is not elevated on top of shell material.

Measuring a buried sediment plate using a fibreglass stock rod (1.1m long) and straight edge.

Care is needed in shelly sediments to ensure that the measurement probe penetrates to the plate, and an erroneous depth is not recorded due to the probe hitting shell.

5.4 SEDIMENT QUALITY METHODS

This section describes 'per sample' methods for each sediment quality indicator in Table 1. Sample replication and compositing were described in Section 3 and illustrated in Fig. 6.

5.4.1 Sediment oxygenation – aRPD method

The Redox Potential Discontinuity (RPD) is the boundary between oxic near-surface sediment and underlying oxygen-depleted sediment. The aRPD depth occurs where there is a visible change in sediment colour from typically brown surface sediment to, dark grey or black sediment beneath (Fig. 9). To assess aRPD in the field:

1. Extract a sediment core up to 150mm in depth, carefully remove it from the corer, and place horizontally onto a tray.
2. Split the core vertically (with hands or a trowel), trying not to smear mud and obscure the aRPD. In very muddy sediment, or where the aRPD is shallow, it can be easier to lift a plug of sediment with a trowel rather than using a core.
3. Photograph the core, using a ruler for scale (with 0mm at the top of the core).
4. Record the aRPD depth to the nearest mm. Where no defined transition is observed (Fig 9, bottom) the aRPD should be recorded as 'indeterminate'.
5. To improve reliability and repeatability, it can be helpful if the aRPD is assessed in the field by more than one person.
6. As necessary, photographs (with ruler for scale) can be later evaluated by the field team. If the observers cannot reach a consensus on the aRPD depth, it should be recorded as 'indeterminate'.

Assessing a split aRPD core.

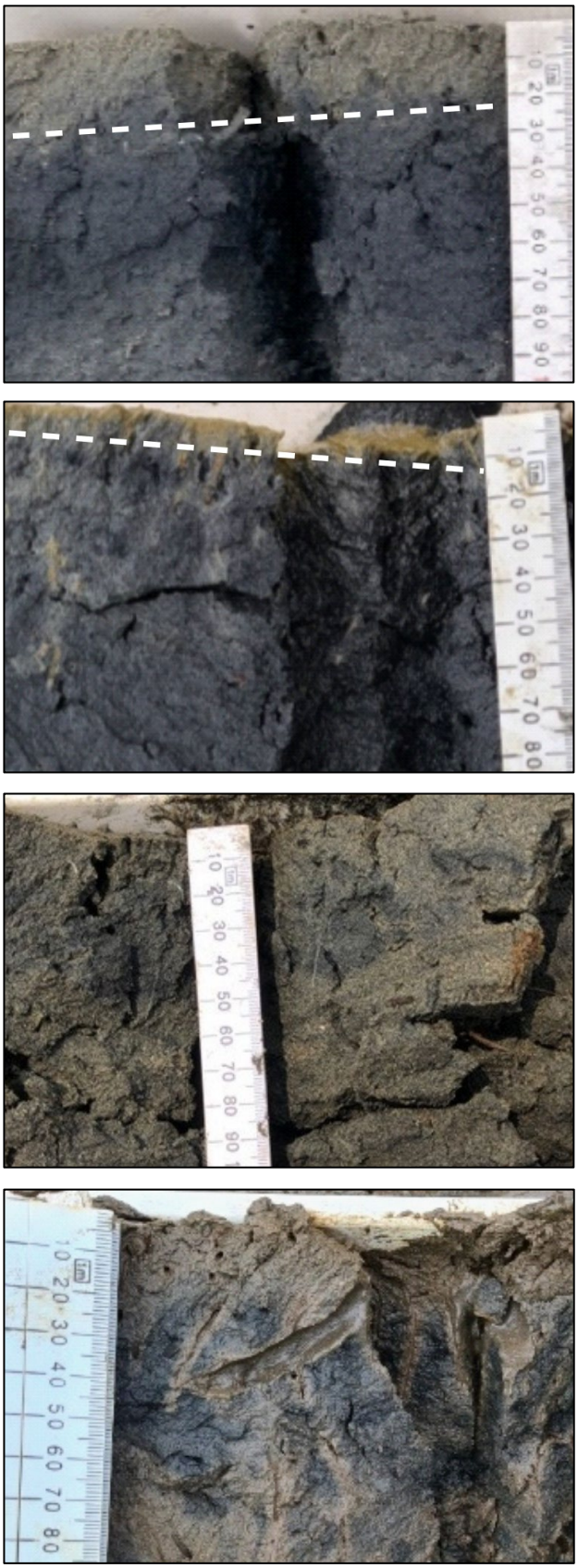


Fig. 9. Sediment aRPD depth, illustrated in the two top photos by a dashed white line. The bottom two photos illustrate where aRPD is classified as indeterminate due to the absence of a clear transition between browner oxic sediment and grey/black oxygen-depleted sediment.

5.4.2 Sediment oxygenation - odour, colour, and micro-organism growths

Qualitative oxygenation indicators based on odour, colour and microbial or microalgal growths can help to:

- Identify obvious situations when enrichment is severe.
- Complement aRPD measurements, and help discriminate situations where a shallow aRPD depth is a result of enrichment versus other factors.
- Provide a record of sediment enrichment status in situations when the aRPD has been classified as 'indeterminate'.

Qualitative rating criteria are outlined in Table 4. These are intended as a rough guide to help ensure that situations of severe enrichment and eutrophication are characterised in terms of their conspicuous features.

The Table 4 criteria build on methods used for salmon farm monitoring [23], and draw on experience in two highly eutrophic New Zealand estuaries [68, 69]. In these locations, patches of sulfur-oxidising bacteria typically occur among beds of opportunistic macroalgae (*Gracilaria* spp.), whereas green/yellow mats of microalgae tend to be most evident after the macroalgae has died and decayed, leaving otherwise bare anoxic sediment [69].

Table 4. Qualitative rating criteria with descriptors for field-based enrichment indicators that complement aRPD measurements, or provide an alternative when the aRPD is 'indeterminate'.

Indicator	Enrichment status				
	Very low	**Low**	**Moderate**	**High**	**Very high/extreme**
Sulfide odour	None Clean, well-flushed sand with no sulfide smell	Mild Typical smell of estuary sand/mud in absence of obvious enrichment.	Moderate Odour detectable when sediment is disturbed, but not unpleasant to most people.	Strong Odour pronounced and unpleasant to most people when sediment disturbed.	Very strong Odour pronounced and unpleasant, and evident even when sediment is not disturbed.
Colour (0-150mm sediment depth)	Brown/clean sediment throughout most of profile	Brown/clean surface sediment and mainly grey beneath.	Brown/clean surface sediment with grey and black beneath	Very shallow (~1-2mm) brown surface sediment overlying black sediment	Intense black throughout sediment profile, with black sediment in patches on surface.
Sulfur-oxidising bacteria (on sediment surface)	Absent	Absent	Absent	Absent or trace	Present and conspicuous
Benthic microalgae (on sediment surface)	Absent or trace	Absent or trace	Absent or trace	Absent or trace	Present and conspicuous

Examples of severe sediment enrichment across broad eutrophic areas. Left: Microbial mats resulting from decay of macroalgae blooms, Middle: Surface anoxia and growths of sulfur-oxidising bacteria (such sediments emit a strong sulfide odour). Right: intense yellow/green colour of microalgae on the surface of anoxic muds, in this example manifesting following the post-bloom decay of *Gracilaria* spp.

5.4.3 Sediment grain size, nutrients, organic matter and toxicants

Sediment quality indicators measured by laboratory analyses are summarised in Table 5. The method for sediment sampling during monitoring is as follows:

1. Request sample containers from the analytical laboratory. If the site IDs are known in advance, the laboratory can usually pre-label the containers.
2. At the fine-scale site, collect sediment to a depth of 20mm using a trowel or core. Each sample should be a composite of four sub-samples (see Fig. 6), to provide a total sample of roughly 500g wet weight. Record site metadata (i.e., location, estuary name, site name, replicate, date, time, etc.)
3. Avoid collecting large material that is unrepresentative of the surface (0-20mm deep) sediment matrix (e.g., stones, sticks, whole shells, cockles, seagrass fronds, macroalgae).
4. Ensure sample containers are sealed to prevent leakage, and are correctly labelled. Labels need to be robust enough that they don't rub off when the sample containers get wet and/or sandy or muddy.
5. When moving between sites, rinse sampling gear to avoid cross contamination.
6. Chill samples after collection and for dispatch to the analytical laboratory. Samples can be refrigerated (≤4ºC) for up to 14 days, or frozen if necessary.
7. Submit samples to the laboratory using standard Chain of Custody (COC) procedures, and include relevant metadata such as site name, sample ID, and date and time of collection.

5.4.4 Laboratory analytical methods and detection limits

Recommended sediment analysis methods and routine detection limits are summarised in Table 5. For trace metals, analytical detection limits of <25% of the Default Guideline Value (DGV) for sediment quality [70] will enable comparison against thresholds for ecological health [6].

Following ANZG (2018) guidance [70], analysing toxicants in the <2mm sediment size fraction is advised for evaluating results against sediment quality guideline values. Additional analysis of the mud-only fraction (<63µm) should be undertaken when more detailed investigations of toxicant bioavailability are required.

Analytical laboratories will have their own QA/QC procedures to ensure the accuracy of sample results. These procedures are not described here. Councils already using their own, or alternative methods, do not necessarily need to change approaches as this could affect long-term datasets. However, the laboratory methods should be recorded as metadata for use in any national or regional data comparisons.

Note! Additional contaminants, such as PAHs, pesticides and plasticisers, could be included where they are of potential concern (e.g., in estuaries with urban or industrial catchments). For these types of analytes, concentrations should be normalised to 1% TOC following ANZG (2018), to enable evaluation against sediment quality guidelines.

Table 5. Sediment quality analytical methods and default detection limits for the analytes that require a specialist laboratory.

Indicator	Method	Recommended analytical detection limit
Grain size classes[1]	Mud (<63µm), sand (≥63µm to <2mm) and gravel (≥2mm) by wet sieving, and calculation of percent fractions by dry weight difference.	0.1 g/100g (0.1%)
Total Organic Carbon	Acid pre-treatment and catalytic combustion (900ºC and O_2) on Elemental Analyser.	0.05 g/100g dry wt (0.05%)
Total Nitrogen	Catalytic combustion (900ºC and O_2), Elemental Analyser, trace level.	200 mg/kg dry wt
Total Phosphorus	Nitric/hydrochloric acid digestion, then analysed via ICP-MS (Inductively coupled plasma mass spectrometry).	40 mg/kg dry wt
Toxicants	Trace metals and arsenic measured on <2mm fraction after nitric/ hydrochloric acid digestion, then analysed via ICP-MS at the trace level.	0.01 - 0.8mg/kg dry wt

[1] Some Councils undertake grain size analysis in 7 size classes, to obtain finer-resolution data on the sand fraction.

5.5 SEDIMENT BIOTA METHODS

5.5.1 Macroinvertebrates

Macroinvertebrates are defined here as sediment-dwelling invertebrates retained on a 0.5mm mesh after sieving, and comprise both infauna and any epifauna species that are sampled by coring. A 0.5mm mesh size is widely used internationally, and reflects the size used by the majority of councils in New Zealand [Appendix 1; 16]. A 0.5mm mesh captures the majority of the macroinvertebrate community and is preferred for temporal sampling and national comparisons [65].

Sampling methods below are based on the original NEMP. Sediment cores should be 130mm diameter and collected to 150mm depth. These dimensions provide a core volume of close to 2L. Collect and process each sediment core as follows:

1. Record site metadata (i.e., location, estuary name, site name, replicate, date, time, etc.)
2. Push the corer to a 150mm sampling depth. In firm or shelly sediment, a handle can be used to rotate the corer in alternating directions to penetrate the sediment matrix. Alternatively, some councils use a mallet to drive the corer into the sediment.
3. Extract the core for sieving. A trowel or similar may be needed to help dig out the core. When removing the core from the sediment, place a hand underneath the corer to avoid sample loss from the bottom.
4. Sieve the samples to remove fine sediment. Key method considerations are:
   i. Sieving needs to be undertaken gently to avoid macerating soft-bodied organisms.
   ii. When sieving in the field, care should be taken so that no water is introduced over the top of the sieve to avoid sample 'contamination' with non-benthic organisms.
   iii. The two most commonly used sieve types are stacked metal sieves and pliable mesh bags. Mesh bags (see bottom photo opposite) are useful as a rapid field method, and a draw-cord facilitates carrying. Metal sieves can also be used in the field, with samples temporarily placed in bags before being sieved in bulk.
   iv. Whole samples can also returned the laboratory, and passed through stacked sieves to facilitate subsequent processing [65].
5. Following sieving, transfer contents to pre-labelled pottles/bags (e.g., using a wide funnel), taking care to wash all retained material from the sieve and the sides of the funnel. A squirt bottle containing seawater or preservative is helpful for this, followed by careful inspection and picking (using tweezers) of any remaining organisms.
6. Preserve the sieved sample in a mix of at least 70% ethanol or isopropyl alcohol (IPA) in water/seawater, or according to laboratory instructions. Be generous with the preservative, using roughly double the sieved volume to prevent degradation of the specimens. This is especially important for samples with high organic matter content (e.g., detritus, numerous cockles).

Tip! As well as labelling the sample pottle/bag, it's a good idea to write the sample name in pencil on waterproof paper, and put it inside the pottle/bag. Note: the preservative will remove permanent marker.

Collecting a macroinvertebrate core. A handle inserted through holes drilled in the top of the corer can be used to rotate it in alternating directions to penetrate the matrix of firm or shelly sediments. A trowel or similar may be needed to dig around the edges to enable corer extraction.

Macroinvertebrate core collected and field-sieved using a mesh bag. Some councils use metals sieves for this purpose. Optionally, whole samples can bagged and returned to the laboratory for sieving.

5.5.2 Epibiota

The epibiota considered in this section are conspicuous (≥5mm body size) organisms that live on the sediment surface, consisting of epifauna and macroalgae. Benthic microalgae are covered in the Section 5.5.3. Two approaches for epibiota monitoring are described below. These methods may also be used to characterise seagrass at fine-scale sites set-up in seagrass habitat, or where seagrass establishes over time. The methods are:

- A semi-quantitative site-wide 'SACFOR' approach based on a method modified from MNCR 1990 [71]. This method is recommended as a **minimum**. SACFOR is useful for a general characterisation of site epibiota, but captures only high-level changes over time or differences among locations. However, it is well suited to characterising clumped and/or patchy epibiota (e.g., aggregations of whelks), where high within-site variability can undermine the utility of quadrat data for assessing temporal change.
- Quadrat sampling, as per the original NEMP. This method is retained as **optional**. Counts of epifauna and percent cover estimates for macroalgae (and seagrass if relevant) are made in 10 replicate 0.25m^2 quadrats (0.5x0.5m) at each site. Quadrats are preferred if the goal is to quantitatively determine temporal changes or differences among locations, provided the limitations of sampling clumped or patchy species are recognised.

It is recommended that the assessment of epifauna excludes the following:

- Infaunal species that may be variably visible on the sediment surface, but whose abundance cannot be reliably determined from surface observation (e.g., cockles or crabs).
- Very small organisms such as the estuarine snail *Potamopyrgus* spp., which are <5mm in size and therefore not 'conspicuous' as defined above, and which are more reliably sampled using cores.

SACFOR estimates

SACFOR is a 6-point semi-quantitative ordinal scale, with thresholds in Table 6 corresponding to categories of: Super-abundant (S), Abundant (A), Common (C), Frequent (F), Occasional (O), and Rare (R). It provides a standardised approach that is easy to use in field surveys without the need for precise measurements. Applying the SACFOR method involves a site walkover to identify and rate the abundance/cover of epibiota. The visual guidance shown in Fig. 10 can be used to help ensure consistency among percent cover observations.

Table 6. Recommended SACFOR categories for semi-quantitative characterisation of epifauna density (per m^2) and macroalgae cover (estimated %), and also seagrass cover if relevant.

SACFOR category	Code	Density per m^2	Percent cover
Super abundant	S	> 1000	> 50
Abundant	A	100 - 999	20 - 50
Common	C	10 - 99	10 - 19
Frequent	F	2 - 9	5 - 9
Occasional	O	0.1 - 1	1 - 4
Rare	R	< 0.1	< 1

SACFOR categories are: Super-abundant (S), Abundant (A), Common (C), Frequent (F), Occasional (O), Rare (R).

Quadrats

Some councils may wish to undertake or continue quantitative quadrat sampling, especially if time-series data have already been established and found to be useful. In that case the following can be considered:

- As per the original NEMP, haphazardly place 0.25m^2 quadrats next to each macroinvertebrate core and/or sediment sub-sample location.
- Identify and count each species of conspicuous epifauna. Check empty shells for fauna that are living on or in them. Some councils prefer to remove all epifauna from the quadrat into a white sorting tray to facilitate identification and counting.
- Estimate the % cover of macroalgae (and seagrass if relevant) as per Fig. 10.

Additional considerations for % cover estimation are:

a) Make separate % cover estimates for opportunistic macroalgal species (e.g., *Gracilaria, Ulva*) versus others.

b) Supplement qualitative percent cover estimates, with field-based 'point counts' in gridded quadrats.

c) Consider taking high-resolution photoquadrats for later quantitative percent cover analysis, for example, using Coral Point Count software [72].

d) Where seagrass is being measured, further method guidance for quadrat sampling, including for percent cover, has been developed by the CSIG [73].

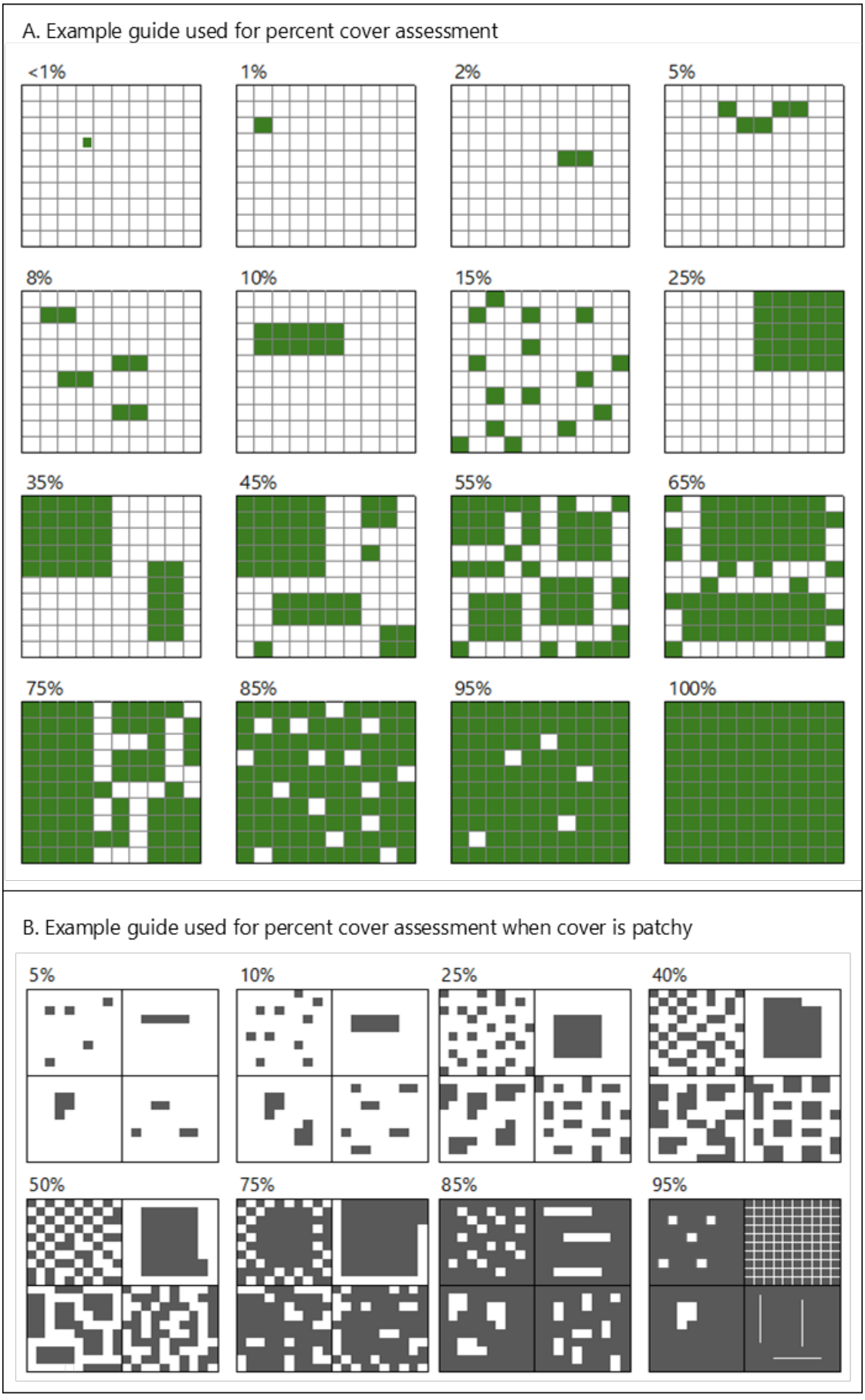


Fig. 10. Visual guidance for estimating percent cover of macroalgae (and seagrass if relevant). As noted in the broad-scale method document, there is scope to improve on this visual guidance using validated photos.

5.5.3 Benthic microalgae

Surface sediment chlorophyll-*a* and phaeopigments can be measured to characterise benthic microalgae (microphytobenthos), and may be useful when analysing drivers of macroinvertebrate community structure. Taxonomic composition analysis of microalgae is not undertaken by most councils due to a lack of national expertise for routine assessment. The method for chlorophyll-*a* and phaeopigments below is modified from the original NEMP, and based on collection of composite samples.

1. Sample the top 5mm of surface sediment (e.g., insert a cut-off 10cc syringe barrel into the sediment, remove barrel containing sediment, insert plunger into barrel and push sediment out, retain top 5mm).
2. Collect a sub-sample in each plot next to each macroinvertebrate core and/or sediment sub-sample. As for sediment sampling, pool sub-samples within each of the 3 columns of the 3x4 site layout grid (see Fig. 6), to create three composite samples per site.
3. Store samples on ice (in the dark) while in the field and freeze (-20^{o}C) until ready for analysis.
4. Once thawed and homogenised, a sub-sample is freeze-dried, extracted with 90% buffered acetone and measured using a fluorometer before and after the addition of hydrochloric acid, which removes phaeopigments.

In addition to quantitative analysis of chlorophyll-*a* and phaeopigments, benthic microalgae can be characterised in the field according to its visible extent using:

- The qualitative enrichment criteria in Section 5.4.2.
- The SACFOR % cover ratings in Table 6 (see preceding section), to provide a site-wide estimate.

These approaches are most appropriate for situations where microalgal mats are extensive and/or conspicuous (e.g., due to eutrophication, see photo).

Extensive conspicuous growth of microalgae on the sediment surface under eutrophic conditions, New River Estuary (Southland).

5.6 QA/QC AND ANALYSIS CONSIDERATIONS

5.6.1 General QA/QC

Some high-level guidance on QA/QC and data handling is as follows:

- For aRPD depth and field-based indicators that involve qualitative or semi-quantitative estimation (Table 4 enrichment indicators, Table 6 SACFOR ratings) it is preferable that the assessment is made by at least two experienced people, with the reported values representing their consensus.
- Where feasible, record field measurements (aRPD, sediment plate depth, etc.) and site metadata electronically in custom templates (e.g., Survey 123, Fulcrumapp). Specifying data entry constraints (e.g., data type, minimum/maximum values for each data type) can minimise the risk of erroneous data entry. Making fields 'compulsory' ensures all critical data are collected.
- Ensure checks are undertaken of the accuracy of any manual data entry. Use electronic laboratory results directly, to avoid the potential for transcription errors.
- When comparing datasets, ensure that the units of measurement are consistent. Check for anomalies or aberrant data points. Box plots or scatterplots may help to identify outliers.
- When calculating averages (e.g., site), it is important to distinguish between missing data and true zeros. For sediment quality, where no measurement was taken, a missing value should be excluded from the site mean. For macroinvertebrates, if a specific species is present in at least one core during a sampling event, its absence in any other core/s should be recorded as zero and those core/s included in the average for abundance.
- For data wrangling and data manipulation, software such as R/RStudio [74] has merit over maintaining large spreadsheets in Excel, as the coding process enables transparent tracking of the data collation and manipulation processes, leaving the raw data unchanged.
- Assigning unique sample identification codes that follow a consistent format can help when merging different data types (i.e., sedimentation rate, sediment quality, sediment biota). An example format is Year-Month-Estuary-Site-Rep (e.g., 2025-11-Waim-A-03; where 11 refers to November and Waim is short for Waimea Inlet in Nelson).

5.6.2 Data handling and analysis

Guidance on data analysis is outlined below, although it is recognised that each council will have its preferred reporting metrics:

Sedimentation

Because sediment plates are generally buried at different depths within a site, the absolute depth of plates should not be used in reporting, as it is unrelated to the sediment accrual measures of interest. Instead, sediment plate data can be compiled to display:

- Cumulative change (at each site) in sediment depth since baseline plate installation. For example, adjust the baseline depth to 0mm, and report changes from the baseline at each plate in mm. Plate measurements can be averaged over the site, with estimates of variance calculated, e.g., mean ± standard error.
- Annual sedimentation rate, preferably calculated from a minimum 5-year record of annual measurements, but ideally from a 10-year record [36].

Note that annual sedimentation rate can be calculated simply as the change in sediment depth (since baseline) divided by n-years. A more sophisticated approach recommended by Hunt (2019) is to fit a linear slope to the data with a confidence interval, thereby determining the upper and lower bounds of sedimentation [36].

Sediment quality

Where sediment quality data include values less than analytical detection limits, half of the detection limit value should be used for data averaging, according to standard convention. When undertaking this process (i.e., for calculating mean values) it is essential to maintain a record of the raw data with 'less than' values unchanged.

Analytical detection limits can change over time, differ with method used, and differ among analytical laboratories (e.g., as has been the case for sediment analysis of TN). Hence it is important when making spatial and temporal comparisons to identify situations where apparent differences in analyte values are merely a reflection of a change in analytical detection limits.

If storage space allows, consider retaining sediment sub-samples (e.g., 100-200g) in case future re-analysis is required, for example, for checking anomalous results or for undertaking analysis of historical samples for new contaminants.

Note, normalising sediment quality values to the % mud fraction, recommended in Part C of the original NEMP [3], is not recommended as it is likely to be misleading in terms of potential for adverse ecological effects.

Macroinvertebrates

Macroinvertebrate processing and taxonomy should be subject to QA/QC processes specified by the provider to ensure the efficacy of sample sorting, accuracy of species identifications, and counts of individuals. It is beyond scope to detail these components as they are usually a service provided by a specialist taxonomy laboratory. However, it is critical that a robust QA/QC process is in place to ensure consistency in the identification and naming of taxa.

Key matters for sample processing and taxonomy were described in a report by Hewitt et al. (2014), with a recommended protocol provided [65]. Since the Hewitt et al. (2014) report was produced, online guidance and mechanisms to foster consistency have been produced as a CSIG Coastal Species Resource Tool, available (with login) at: https://specieskey.atlasmd.com/.

However, even with the best practical macroinvertebrate QA/QC, there are almost invariably differences among providers, or from the same provider at different times, which may need to be resolved prior to analysis of datasets from different surveys or estuaries. Steps to achieve consistency include:

- Update species names in older datasets to match any new names in the latest data. The World Register of Marine Species has accepted species names that can be used for this purpose. See: https://www.marinespecies.org/index.php
- Determine whether there are subtle differences in naming for the same species that need to be resolved (e.g., use of sp. vs sp.1 vs sp.#1 vs spp.).
- Where taxonomic differences remain between datasets, but are suspected to be artefacts (usually due to a change in provider), it may be necessary to aggregate species or taxa to higher taxonomic levels before analysis. This process usually requires case-by-case expert judgement.

No guidance is provided here on the most appropriate analyses for macroinvertebrate data, as these are likely to be situation-specific. At least three macroinvertebrate indices are potentially useful for regional or national use [6]. These are the international biotic health index AMBI [27, 75]; the National Benthic Health Model or BHM [67]; and Traits-Based Index or TBI [76], noting that the TBI has, to date, not been rigorously tested outside of Auckland and Waikato.

5.7 METADATA RECORDS

The following general information should be recorded:

General site information

- The presence of visible gradients (e.g., in terms of sediment type, slope, current exposure) across each sampling site.
- Tidal elevation of each site relative to mean sea level, and any elevation differences. LiDAR data can be used to estimate this as part of a desktop assessment.

Survey-specific information

- Date/time and GPS position for each sample. Note that electronic platforms such as Survey 123 and Fulcrumapp create these fields automatically, and also enable linked photographs to be taken.
- Site and/or sample sediment type and texture as per guidance in the broad-scale methods document [4].
- Sediment surface features and evidence of disturbance, such as flood scouring or deposition events (e.g., mud, detritus, terrestrial debris).
- Sample features and sampling issues (e.g., extent and removal of stones, sticks, etc., from sediment samples).
- Low tide time and tidal range on the day of sampling, and lags in estuary tides compared with local predictions or nearest tidal stations. Among other things, this information can help with planning of future surveys (e.g., by providing understanding of when sampling sites are likely to have uncovered).
- Synoptic measurements of water salinity and temperature in tidal channels can help with ecological data interpretation.
- The people in the field team, especially those making assessments that are subject to observer differences (e.g., aRPD).
- The provider(s) and methods for laboratory analyses, macroinvertebrate sample processing, and taxonomic identification.

Additional macroinvertebrate information

- The size of the corer and sieve mesh used (most commonly the recommended 130mm diameter corer and 0.5mm mesh).
- The nature of the sieved sample (amount of shell, gravel, terrestrial woody debris, marine-derived organic material, etc.) and an estimate of the sieved volume. The latter is useful for planning pottle sizes and preservative volumes for future surveys.
- The method of sieving (core bags, metal sieves), and whether sieving was undertaken in the field or laboratory.
- When targeting vegetated habitats it is important to keep records of where cores were taken (e.g., on top of the vegetation feature or in bare sediment between patches). As already noted, macroinvertebrate results are likely to differ depending on what is sampled [e.g., 63], so it is important that sampling is representative, and consistent over time.

Fine-scale monitoring in Pleasant River, Otago.

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APPENDIX 1. FINE-SCALE MONITORING UNDERTAKEN BY COUNCILS

This information has been extracted verbatim from the 'Scoping review to update the National Estuary Monitoring Protocol' [16].

Question	YES	NO	na
Do you do fine-scale monitoring using the NEMP?	12	2	2
In-house	3	-	-
Contracted out	4	-	-
A combination of in-house and contracted out	7	-	-
Do you deviate from the NEMP fine-scale methods?	14	0	2
Where are your fine-scale sites located?	YES	NO	na
Representative areas of the dominant intertidal habitat type	11	3	2
Deposition zones where eutrophic impacts are likely to be first expressed	3	11	2
In a range of intertidal habitats	5	9	2
Subtidal areas	1	13	2
What fine-scale parameters do you analyse?	YES	NO	na
Carbon as Ash Free Dry Weight (AFDW)	3	11	2
Carbon as Total Organic Carbon (TOC)	13	1	2
Sediment aRPD (apparent Redox Potential Discontinuity) depth	10	4	2
Sediment ORP (Oxidation-Reduction Potential)	1	13	2
Particle grain size (PGS) by wet sieving	13	1	2
Particle grain size (PGS) by laser diffraction	2	12	2
TN (Total Nitrogen)	13	1	2
TP (Total Phosphorus)	13	1	2
TS (Total Sulfur)	2	12	2
Basic metals suite (Cu, Cd, Cr, Ni, Pb, Zn)	14	0	2
Extended metals (e.g. As, Hg)	12	2	2
Polycyclic aromatic hydrocarbons (PAHs)	4	9	2
Semi-volatile organic compounds (SVOCs)	4	10	2
Emerging contaminants	2	11	2
Sediment Chlorophyll-a	7	7	2
What type of samples are collected? (D = discrete, C = composite)	C	D	na
Metals	11	3	2
Nutrients (TN, TP)	10	3	3
Carbon (AFDW/TOC)	11	3	2
Particle Grain Size (PGS)	11	3	2
PAHs, SVOCs	5	0	11
Emerging contaminants	2	0	13
Chl-a	3	3	10
aRPD/ORP	1	8	7
For chemical analyses, what depth of sediment is collected?	YES	NO	na
Surface 20mm	14	0	2
Core depth (150mm)	0	14	2
For chemical analyses, what fraction of the sediment sample is analysed?	YES	NO	na
Whole sample	11	3	2
<2mm (mud and sand fractions)	2	12	2
<0.5mm	1	13	2
<63µm	1	13	2

Question			
What is the average number of sediment chemistry samples collected per site?	≤3	4-9	≥10
Metals	8	4	2
Nutrients	8	3	2
AFDW/TOC	7	3	2
PGS	7	2	1
PAHs	1	2	1
SVOCs	1	1	1
Emerging contaminants	0	1	1
Chl-a	1	3	1
aRPD/ORP	2	1	4
What macroinvertebrate sampling parameters do you use?	130mm	150mm	Other
Core diameter	13	1	0
Core depth	0	14	0
What macroinvertebrate sampling parameters do you use?	0.5mm	1.0mm	Other
Mesh size	13	1	0
What is the average number of macrofauna samples collected per site?	≤9	10	>10
	2	8	4
How do you assess epibiota?			na
Not assessed	2		2
Quadrat counts	7		2
Site SACFOR rating	6		2
Derived from macroinvertebrate cores	2		2
Are epibiota excluded from macrofauna analyses? (Unkn = unknown)	YES	NO	Unkn
	9	2	3

NA=Not Answered/Not Applicable

APPENDIX 2. EXAMPLE OF AN EQUIPMENT LIST

FINE-SCALE MONITORING (WITHOUT SEDIMENT PLATES)

In the field:

- Site information such as a map, GPS coordinates, sampling plan and tide predictions.
- Straight edge, sediment plate probe and/or knitting needles (depending on method).
- Stainless or plastic trowel for collecting samples.
- Ruler (foldable 1m ruler).
- Camera, phone and/or tablet (GPS enabled).
- GPS (if needed).
- Backup waterproof notebook/paper.
- Pre-labelled sediment sampling containers supplied by analytical laboratory.
- Bamboo markers with flagging tape to mark fine-scale plot boundaries (as necessary).
- Corer (diameter 130mm, ~200-300mm long) with sharpened base, and handle to enable rotating action for digging into the sediment). The corer should be marked at the sampling depth of 150mm.
- 0.5mm mesh sieve bags, or containers to transport macroinvertebrate samples to the laboratory for sieving.

At the vehicle:

- Chilly bin with ice packs for sediment samples.
- Pre-labelled macroinvertebrate sample pottles (potentially supplied by taxonomic laboratory), with internal labels resistant to alcohol (e.g., waterproof paper tags labelled with pencil).
- Wide mouth funnel for transfer of sieved residue to pottles.
- Squirt bottle for washing sieved residue adhering to side of funnel.
- Preservative (e.g., isopropyl alcohol) at vehicle (>70% mix in water/seawater).
- Courier tickets and address labels.
- Packing tape.

SEDIMENT PLATE MONITORING

Installation of plates:

- Concrete pavers (e.g., 19cm x 23cm, 4 per site).
- Steel waratahs (400mm, 4 per site).
- H4 treated wooden pegs (400mm).
- 50m measuring tape.
- Club hammer/mallet.
- Spade.
- Level.

Measuring plates:

- Straight edge, sediment plate probe and/or knitting needles or metal rods (10) (depending on method).
- Ruler (foldable 1m ruler).
- Pre-labelled sediment sampling containers supplied by analytical laboratory.
- Stainless or plastic trowel for collecting samples.
- Camera, phone and/or tablet (GPS enabled).
- Backup waterproof notebook/paper.

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