

# Thatcher Bay Nearshore Restoration Monitoring 2015



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## Executive Summary

Wood waste contamination is a serious concern for many areas in the Pacific Northwest. Thatcher Bay on Blakely Island in the San Juan Archipelago underwent restoration work in December 2014 to remove sediment that had been contaminated from wood waste from a milling operation. In 2008, the Skagit Fisheries Enhancement Group (SFEG) collaborated with the University of Washington (UW) and Friday Harbor Labs to perform a restoration feasibility study and collect a variety of baseline data from the project site. In 2015, the SFEG again partnered with the UW in order to monitor the progress of the Thatcher Bay Restoration Project. Monitoring of the site is critical for assessing the project's ability to achieve predetermined objectives. The project's goals are to improve both ecosystem functions and nearshore habitat for flora and fauna. Monitoring conforms to current regulatory standards and guidelines and provides a template for future efforts to restore nearshore areas impacted by wood waste deposition. Individual elements of this monitoring plan include (1) assessing intertidal sediment characteristics, (2) evaluating benthic macroinvertebrates assemblages, (3) surveying for forage fish spawning activity, and (4) observing for the potential spread of native eelgrass (*Zostera marina*) into the restoration site.

Monitoring began after restoration work was completed in December 2014 and is currently ongoing. The project's parameters are being met through the coordinated efforts of state agencies, universities, local tribes, and both private and nonprofit organizations. Forage fish spawning surveys completed in June 2015 indicate that surf smelt (*Hypomesus pretiosus*) have utilized the restoration site for spawning habitat. Spawning, which had not been previously observed, is an indication that site conditions and natural processes are being restored and that project objectives are being met.

# 1. Introduction

Wood waste in Puget Sound is common due to the prevalence of lumber and paper industries along the shoreline (Kendall 1997). Types of wood waste include sawdust, wood chips, large logs, and other material that can become intermixed with nearshore sediment. Studies have demonstrated that large amounts of wood waste are slow to degrade in an aquatic environment and can persist for decades (Conlan 1977; Schultz and Berg 1976; Harmon et al. 1986). In December 2014, the Skagit Fisheries Enhancement Group (SFEG) completed a restoration project which involved the removal of wood waste from nearshore habitat in the San Juan Archipelago. The location of the project is in Thatcher Bay on Blakely Island (Figure 1).



**Figure 1.** Location of the Thatcher Bay Nearshore Restoration Project

Approximately 11,800 cubic yards of sediment were excavated from a 1.8 acre area (SFEG 2015). The wood waste had been deposited from a lumber mill that had operated on the island for decades. Nearshore processes and ecological functions were to be restored with the removal of contaminated sediments and the subsequent emplacing of non-contaminated fill into the project area. Monitoring of the restoration site was performed during the first year following the completion of work with the results presented in this report. The site will continue to be monitored through year two. The restoration and monitoring of Thatcher Bay has been a joint

effort between SFEG and the University of Washington which began in 2008 with the thesis work of Joel Breems.

### *1.1 Goals and Objectives*

The Skagit Fisheries Enhancement Group established that the goal of the Thatcher Bay Nearshore Restoration project was to improve the natural processes and habitat functions of the impacted nearshore area. The objectives included:

- Eliminate toxic sulfide contamination with the removal of wood waste.
- Restore the forage fish spawning habitat on the beach.
- Restore intertidal areas to improve benthic flora & fauna habitat.

The goals of monitoring restoration actions for the site follow those described by the Monitoring Framework established for the Puget Sound Nearshore Ecosystem Restoration Project (Brandon et al. 2013). They include:

- Assess the effectiveness of restoration actions in achieving defined objectives.
- Determine where corrective action is needed to improve the effectiveness of restoration actions, and inform decisions about how to take such corrective action.
- Reduce risks and uncertainties associated with future restoration actions by increasing understanding of the relationships between restoration actions and restored ecosystem processes, structures, and functions for Puget Sound nearshore ecosystems.

### *1.2 Project History*

Thatcher Bay is located on the western side of Blakely Island in Washington State's San Juan Islands. The island had been historically used by the Lummi and Samish tribes for fishing, hunting, and plant gathering (Roe 2005). In 1879, Thatcher Mill (later renamed Spencer Mill) began processing wood in Thatcher Bay up until 1942 (Figure 2). After decades of operation, portions of the bay had accrued large amounts of wood waste, so much so that local residents referred to the area as "sawdust beach" (Figure 3). Wood waste in large volumes, which natural systems and organisms are not adapted to, can overwhelm the assimilative capacity of sediment in aquatic environments and can potentially harm the environment (WA Dept. of Ecology 2013).





**Figures 2 and 3.** Spencer Mill (left), source Roe and “sawdust beach” (right) source: SFEG.

A 2009 study of the site done by the Skagit Fisheries Enhancement Group, the UW’s Friday Harbor Labs, and Joel Breems, of the University of Washington, assessed both environmental conditions and restoration feasibility options. That study, which estimated the amount and distribution of the wood waste, concluded that the elevated total organic composition was negatively impacting the nearshore habitat (Breems 2009). Sediment was analyzed at various locations within the project site and the results showed that the depth of wood waste averaged 1.8 feet below Mean Lower Low Water (MLLW). A large amount of organic material such as this is what likely facilitated an increase in anaerobic respiration and the production of sulfide. Breems showed that redox potential and sulfide measurements taken from sediment cores were elevated at Thatcher Bay when compared with data obtained from a nearby reference site. Sulfide can be toxic to marine flora and fauna (Wang and Chapman 1999) and has been shown to decrease benthic invertebrate species diversity and abundance (Hyland 2005).

The SFEG and UW study further concluded that the optimal method of improving the habitat was the removal of the contaminated sediment from a water-based platform. This option was preferred over alternative strategies like sediment capping or taking no action and allowing for autogenic repair to improve conditions. With a capping strategy, the long term effectiveness of a sediment cap is dubious in a dynamic intertidal environment like Thatcher Bay. Alternatively, the strategy of taking no action was also eliminated as natural processes were unlikely to restore the site to historical conditions before the mill existed.



In November of 2014, using funds from the Salmon Recovery Funding Board, the SFEG contracted with Pacific Pile & Marine to excavate 11,800 yd<sup>3</sup> of contaminated sediment from the project site (Figure 4). The contaminated sediment was then dispersed in open water at the Rosario Strait dispersive site in the eastern San Juan Islands (Figures 5). In December of 2014, a flat deck barge backfilled the project site with material obtained from Cowden Gravel & Ready Mix's Singer Pit in Whatcom County (SFEG 2015). The backfill material had chemical analysis preformed on it before placement to ensure compliance with construction specifications to maximize environmental protection. Upon completion of the restoration work, the SFEG began monitoring of the site by collaborating with the University of Washington, the Samish tribe, and both private and non-profit organizations.



**Figures 4 and 5.** Sediment excavation (left) and location of dispersal site (right). Source: SFEG.

### *1.3 Purpose of Monitoring*

Monitoring is a vital component of any comprehensive restoration project. The success of restoration work hinges on two stipulations; that ecosystems can be altered to recreate a desired condition, and whether it can be determined if the alterations have produced the desired condition (Keddy 2000). The scientific uncertainties involved in restoration outcomes are what monitoring efforts try to better understand so that similar endeavors in the future can be improved upon. The Thatcher Bay Restoration Project has benefited greatly from the variety of pre-project data observations obtained by the UW's Joel Breems. These data can be compared

with post-project monitoring results to help assess environmental changes. Another benefit of having pre-project data for a restoration project is that suitable and attainable objectives can be established that are based on the best current scientific understandings.

The Puget Sound Nearshore Restoration Project's Monitoring Framework defines effectiveness monitoring, which is what this project aims to accomplish, as evaluating whether or not restoration actions are achieving their stated goals. For this project, a set of parameters were established with the intention that monitoring would be accomplished according to the current regulatory standards and guidelines for each element. Those elements include:

- 1) Assessing intertidal sediment characteristics
- 2) Evaluating benthic macroinvertebrates assemblages
- 3) Surveying for forage fish spawning activity
- 4) Observing for the potential spread native eelgrass (*Zostera marina*) into the restoration site

One of the aims of this project is that it may offer as a model for future efforts to restore nearshore areas altered and impacted by wood waste deposition. While there are currently only a few sawmills in operation statewide, there were potentially hundreds of mills operating in Washington State at the height of the timber industry (WA Dept. of Ecology 2013). It is likely to assume that other sites in Puget Sound will undergo wood waste restoration work in the future. The monitoring criteria established in this report can provide a template for those remediation efforts that have goals and objectives similar to this project.

## **2. Sediment Characterization**

### *2.1 Overview*

The conventional analysis of sediments can provide a variety of uses when investigating the consequences of restoration practices. The Washington Department of Ecology's Toxics Cleanup Program's 2013 guidelines for wood waste investigations indicate, among other variables, that analysis of quantitative grain size, the percentage of solids, and the percentage of total volatile solids should be included as elements of a project's goals and objectives. The Toxics Cleanup Program's Sediment Sampling and Analysis Plan (2008) attests that sediment grain size can

provide insights into the interpretation of sediment toxicity test data and benthic macroinvertebrate abundance data as well as helping to evaluate sediment transport and deposition. Results for the percentage of total solids in a sediment sample allow for the expression of chemical concentrations on a dry-weight basis. Total volatile solids (TVS) represents nitrogen, oxygen, and sulfur containing compounds and their associated hydrogen atoms as well as the carbon content-associated sediment. TVS analysis may better correlate with biological results than total organic carbon and can be used to assess the overall volume of wood waste in a sample. TVS is the chemical indicator most often used to correlate with confirmatory bioassay results and is often used to develop site-specific cleanup standards. TVS also provides objective, reproducible measures of the overall organic content in sediment which can then be used to assess the percentage of wood waste present in sediment (WA Dept. of Ecology 2013). Sediment from the project site had previously been analyzed from samples collected in April 2009 as part of a suitability assessment for the Dredged Material Management Program (DMMP). The sediment collection methods in this report differed from those used for the 2009 results (hand tools vs vibracore). The 2009 results are included in this report for comparison.

<b>GRAIN SIZE</b>	<b>% Gravel</b>	<b>0.8</b>
	<b>% Sand</b>	<b>5.4</b>
	<b>% Silt</b>	<b>83.7</b>
	<b>% Clay</b>	<b>10.1</b>
	<b>% Fines (clay+silt)</b>	<b>93.8</b>
<b>Total Solids (%)</b>		<b>58.6</b>
<b>Volatile Solids (%)</b>		<b>12.9</b>

**Table 1.** 2009 sediment analysis results from DMMP study.

## *2.2 Methods*

Bulk sediment samples were collected on 9/28/2015 and frozen for preservation until analysis was performed at the Restoration Ecology laboratory at the University of Washington's Center for Urban Horticulture. All analysis was done according to the Puget Sound Estuary Program

(PSEP) 1986 protocols for sediment sampling and analysis (Appendix C). These methods are originally derived from Plumb (1981) and are the most up to date guidelines for performing sample collection and characterization for Puget Sound sediments. See figure 6 for the location of where sediment samples was obtained. Samples were homogenized both on site and before laboratory analysis as per PSEP guidelines.



**Figure 6.** Location of sediment grabs.

The percentage of total solids was obtained by homogenizing and then drying wet samples to constant weights in an oven at 105° C. The percentage was obtained by dividing the dry weight of a sample with the wet weight. Weights were measured to tenths of a gram.

The percentage of total volatile solids was obtained by initially following the same procedure as that of total solids. Once dried and then weighed, the samples were ignited in a muffle furnace to a constant weight at 550° C. The percentage of volatile solids was obtained by dividing the weight of the ignited residue by the dry weight. Weights were measured to tenths of a gram.

Grain size percentages were determined by wet sieving samples into size fractions greater than 62.5  $\mu\text{m}$  (i.e., sand and gravel) and less than 62.5  $\mu\text{m}$  (i.e., silt and clay). Sand and gravel samples were then dried to a constant weight at 90° C, weighed, and sieved through screens of different sizes. Each size fraction was weighed and divided into the total sample weight to obtain

a percentage for each size class. Due to the small amount of fine sediments, the total percentage of fines was presented and the classifying of fine sediments less than 62.5  $\mu\text{m}$  was ignored. A total of 50 samples with .25g of sediment for each was analyzed for both total solids and total volatile solids.

### 2.3 Results

Sediment Parameter	Percentage (%)
Total Solids	90.3
Total Volatile Solids	5.4
<b>Grain Size</b>	
Gravel (> 2.0 mm)	21.9
Very Coarse Sand (2.0-1.0 mm)	26.4
Coarse Sand (1.0-0.5 mm)	17.8
Medium Sand (0.5-0.25 mm)	19.2
Fine Sand (0.25-0.125 mm)	13.9
Very Fine Sand (0.125-0.0625 mm)	0.72
Total Sand and Gravel (>2.0-0.0625 mm)	99.2
Total Fines (Silt and Clay 0.0625-0.0039 mm)	0.08

**Table 2.** Results of sediment characterization analysis from 9/28/15 collection.

The grain size results show that sediment obtained from the project site, 10 months after completion of the work, are predominantly sand at 99.2% (Table 2). Previous results from 2009 sampling data indicated that the sediment that was to be dredged was predominantly silt at 83.7% overall (Table 1). Total volatile solids were also lowered from 12.9% in 2009 samples to 5.4%.

## 3. Forage Fish Spawning Surveys

### 3.1 Overview

Forage fishes are schooling fishes that are important prey items for large fish like salmon and other wildlife. Two of the most common forage fish species in Puget Sound are surf smelt (*Ammodytes hexapterus*) and sand lance (*Hypomesus pretiosus*) (Figure 8). There are many nearshore areas that have been documented as spawning habitat and that act as important nurseries and feeding grounds for these species (Penttila 2007). Surf smelt spawning has been previously documented in an area adjacent to the work site in Thatcher Bay (Friends of the San Juans, 2004) (Figure 9). Surveying the project site for evidence of spawning is an essential task towards assessing the restoration's efforts at improving habitat. One of the project's objectives is to improve forage fish habitat and evidence of spawning is a critical indicator toward assessing



the success of achieving this objective. Surf smelt in the San Juan Islands are known to spawn year round with the summer showing an increase in spawning activity (WDFW 2015). Sand lance spawn from November to late March or even early April.



**Figure 7.** Surf smelt (above) and sand lance (below). Photo:WDFW.

The results of the grain size analysis, previously reported in this report, demonstrate that the sediment at the site went from being characterized as mostly silt to being predominantly a gravel and sand makeup. The Washington Department of Fish and Wildlife (WDFW) states that the preferred sediment attributes for surf smelt spawning are a sand and gravel mix where most of the sediment is between 1-7 mm in size (Penttila 2007). The WDFW maintains a database with historical records of the locations throughout Puget Sound that have had forage fish spawning observed from surveying. Thatcher Bay has had two locations, neither of which are in the project site, that have had surveys documenting the presence of surf smelt eggs (WDFW 2016). WDFW reports that those surveys occurred in September of 2003 and March of 1990.

#### *4.2 Methods*

All surveying was done according to the WDFW's Forage Fish Spawning Beach Survey Manual (Moulton and Penttila 2001). The complete guidelines for sample collection is included in Appendix B. Protocols include:

- Examine the beach for the most likely zone to contain eggs (+7 to +9 ft. MLLW).
- Identify a 100 ft. stretch of beach to sample and document with GPS equipment.

- Obtain and condense bulk beach sediment samples according to field manual guidelines.
- In lab, condense beach samples and examine under dissecting microscope for eggs.

The year one surveys took place in March, June, September, and December of 2015. Restoration site surveys were compared to surveys done at three other reference sites that were surveyed the same day as the restoration site (Figure 10). The reference sites are intended to represent relatively unimpacted or least impacted conditions. The reference beaches were located on Strawberry Bay on Cypress Island, the southwest exposed bay on James Bay Island State Park, and at Thatcher Bay on the beach found northeast of the reference site beach. All of the beaches were similar to Thatcher's in that they were shallow bays and had southwest exposures.



**Figure 8.** Map of forage fish survey reference site locations.

#### 4.3 Results

Analysis of the June 2015 forage fish spawning survey, for the Thatcher Bay restoration site, indicate that surf smelt (*Hypomesus pretiosus*) have utilized the upper beach as spawning habitat.



Eggs were found in the upper intertidal zone at a tidal elevation range of +7-9 feet (Figure 10). None of the surveys from reference sites yielded any evidence that spawning had occurred. There was also no evidence of spawning at the restoration site other than the June survey. This would corroborate that the summer is the more likely time to survey for surf smelt spawning.



**Figure 9.** Surf smelt eggs at 10x magnification from a June 2015 survey.

## **4. Macroinvertebrate Assemblage Evaluation**

### *4.1 Overview*

Benthic and epibenthic macroinvertebrate monitoring is important because they are indicators of ecosystem health. They play a crucial role in the ecology of the nearshore through a variety of activities. Studies show that sediment that is just 20 percent wood waste by volume could negatively impact the benthic community (Kathman et. al. 1984; Kirkpatrick et. al. 1998; SAIC 1999). Analyzing sediments for benthic and epibenthic macroinvertebrates assesses statistically significant alterations in the naturally occurring abundances of major taxa like Crustacea, Mollusca, and Polychaeta (DOE 2008). For this project, benthic and epibenthic macroinvertebrate assessments were performed consistent with the Puget Sound Ambient Monitoring Program's (PSAMP) procedures to allow for a direct comparison of data. Parameters that the PSAMP uses to assess macroinvertebrate abundance are: total abundance, major taxa abundance, taxa richness, Pielou's evenness, and the Swartz's Dominance Index (WA Dept. of Ecology 2008).

## *4.2 Methods*

On 30 June 2015, benthic samples were collected from three sites within the project site and at three sites from a reference location that is in Thatcher Bay but outside the restoration site. Sample collection was planned and overseen by the consulting firm of Anchor QEA, LLC. During the low tide, a hand coring device was pushed 10 cm into the sediment. As per Washington Dept. of Ecology guidelines (2008), the sediments retained in the corer were placed in large plastic bags, transported to a sieving station, and then sieved through a 500- $\mu$ m screen. Material retained on the screen was placed in jars and preserved with 10% phosphate-buffered formalin (WA Dept. of Ecology 2008).

Once the tide had risen to a depth of roughly 0.6 – 0.9 m, an epibenthic suction pump was used to collect replicate samples of the epibenthos at locations adjacent to the benthic sampling sites. The pump covered a 0.033 m<sup>2</sup> area of the bottom and had 0.130-mm screened ports that retained the macroinvertebrates but allowed water to pass through and flush the system. Samples were collected by running the pump for 60 seconds, running the outflow through a 160  $\mu$ m mesh. The retained materials were again placed in jars with 10% phosphate-buffered formalin. One week after collection, all samples were transferred to 70% ethanol and stained with rose Bengal. The samples were sorted and all invertebrates were identified to the lowest possible taxonomic level. Taxonomic analysis was performed by Brian Bingham at Western Washington University's Department of Environmental Sciences and his findings are presented below.

## *4.3 Results*

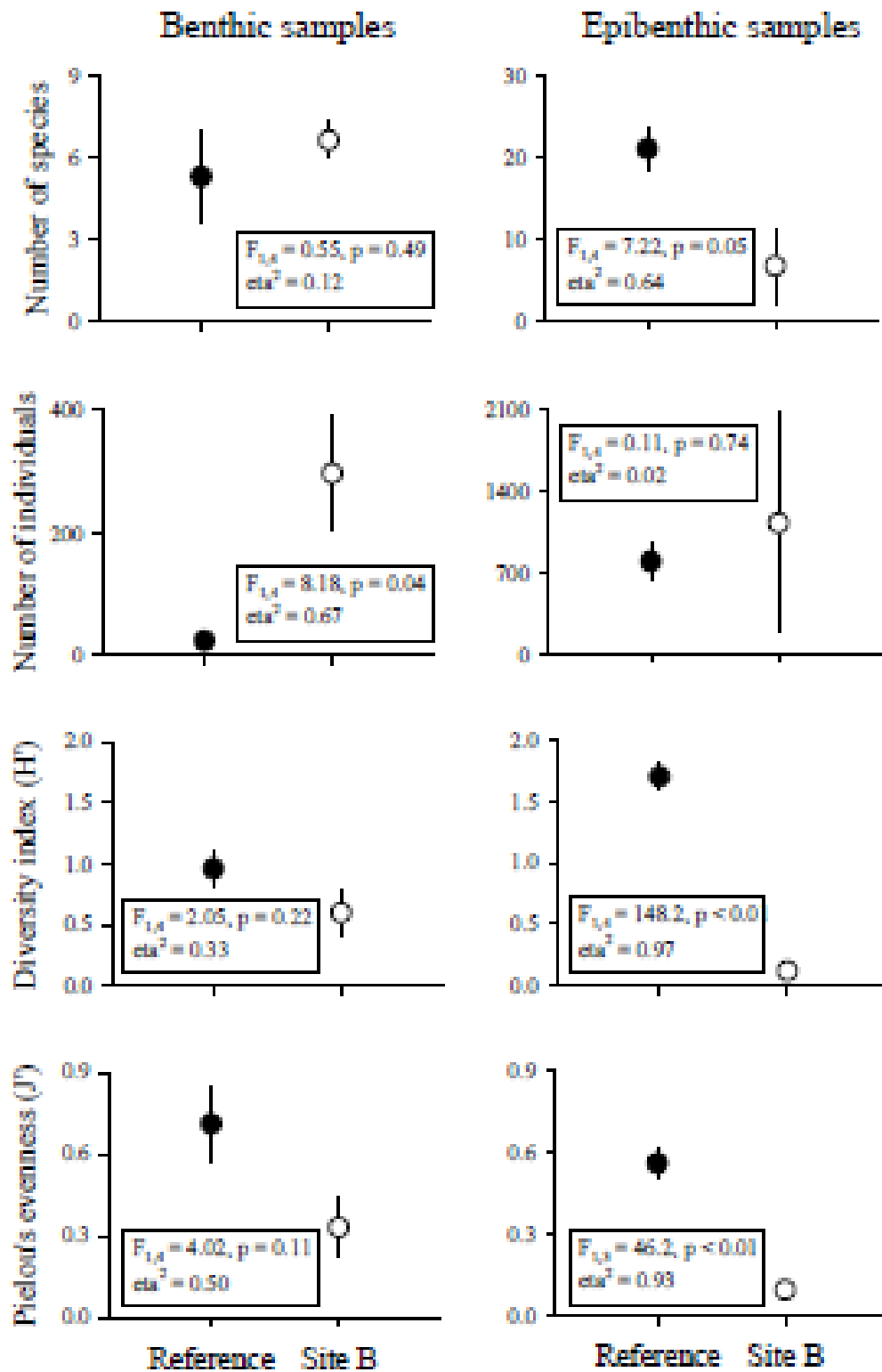
Taxonomic analysis indicated that few species were present in benthic samples from either the restoration site or the reference site (Table 3) and there was no statistically significant difference in number of invertebrate species, diversity or evenness at the two sites (Figure 10). The epibenthic samples from both the restoration site and the reference site held many more species and many more individuals than were present in the benthic samples (Table 3). Analysis also showed significant differences in number of species, diversity and evenness; all were higher in the reference site (Figure 10).

	Number of species	Number of Individuals	Shannon Diversity Index	Pielou's Evenness
<b>Benthic samples</b>				
Restoration site	6 – 8 (x = 6.6)	198 – 486 (x = 297.3)	0.20 – 0.84 (x = 0.60)	0.09 – 0.47 (x = 0.32)
Reference site	2 – 7 (x = 5.3)	2 – 50 (x = 24.3)	0.69 – 1.21 (x = 0.96)	0.50 – 1.00 (x = 0.71)
<b>Epibenthic samples</b>				
Restoration site	1-16 (x = 6.6)	159 - 3001 (x = 1123.3)	0 – 0.23 (x = 0.11)	undef – 0.09 (x = 0.09)
Reference site	18 - 26 (x = 21.0)	508 - 1067 (x = 794.6)	1.48 – 1.81 (x = 1.70)	0.45 – 0.62 (x = 0.56)

**Table 3.** Benthic and epibenthic invertebrate community metrics. Ranges and mean values are shown.

<b>Benthic samples (average dissimilarity = 86.3)</b>			
	Average abundance at Reference Site	Average abundance at Restoration Site	% contribution to site differences
Unidentified nematodes	0.3	15.5	48.1
<i>Pseudopolydora bassargensis</i>	0	4.1	13.7
<i>Mediomastus</i> sp.	3.3	2.8	7.6
<i>Ectinosoma</i> sp.	0	1.6	5.2
<b>Epibenthic samples (average dissimilarity = 67.5)</b>			
<i>Harpacticus</i> sp.	16.4	3.4	17.8
Unidentified nematodes	15.2	26.8	16.2
<i>Tisbe</i> sp.	9.1	0	11.9
<i>Mediomastus</i> sp.	7.3	0.7	8.6

**Table 4.** Analysis indicating the top 4 species contributing to differences in the benthic communities between each pair of study sites. Average abundances are given in individuals per sample.



**Figure 10.** Invertebrate indices for benthic and epibenthic samples ( $\pm$ SE). Source: Bingham 2015.

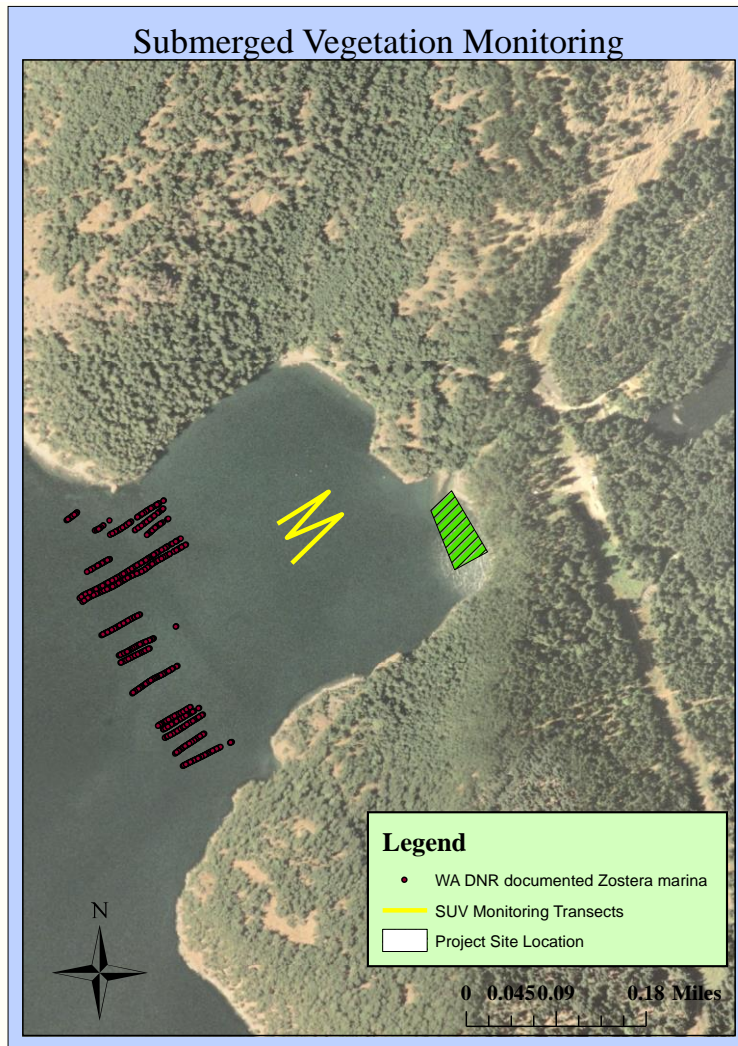
## 5. Eelgrass (*Zostera marina*) Observations

### 5.1 Overview

*Zostera marina* (eelgrass) is a marine flowering plant that is acknowledged as an indicator of ecosystem health and stability in the Pacific Northwest (Phillips 1984; Wyllie-Echeverria and Ackerman 2003). Eelgrass provides important habitat functions in the nearshore region (Mumford 2007, Eissinger 2007), reduces current flow and stabilizes sediment (Gambi et al. 1990, Fonseca et al. 1982) and can act as an indicator to assess water quality (Dennison et al. 1993). Monitoring for the potential colonization of the emplaced sediment and other areas of the bay will be integral towards understanding the ecology of the both restoration site and the bay as a whole. Previous sampling done by the Washington Department of Natural Resources in 2010 show that there is an existing bed of native eelgrass (*Zostera marina*) at the mouth of Thatcher Bay (Figure 11). Eelgrass beds though can be ephemeral and move over time. The Puget Sound Partnership uses eelgrass as one of its “vital signs” to assess the health of Puget Sound (PSP 2016). Both the restoration site and other areas of Thatcher Bay were visually assessed for the presence of underwater vegetation and included as a criteria of this monitoring report.

### 5.2 Methods

In September, 2015 a GoPro™ camera was mounted to 10 feet of PVC pipping and submerged beneath a boat to observe for the presence of vegetation. The setup was held by hand and underwater video was taken on randomly chosen transect lines established beforehand. The boat travelled at a speed of 1 knot and observations were taken midday in order to maximize the amount of available light. A GPS device was used while taking underwater video to document the position of the boat. The sampling design was intended to adhere to guidelines for estimating the basal area for underwater vegetation established by Norris et. al (1997). The depth was obtained using the boats’ sonar. During low tide the project site was monitored for vegetation by visual observations.



**Figure 11.** Map of documented *Zostera marina* and SUV transects.

### 5.3 Results

The results of underwater video analysis did not indicate the presence of any submerged underwater vegetation in the transect area covered. No vegetation was visually observed in the project site during monitoring at low tide either.

## 6. Discussion

### *Sediment Characterization*

These data are important in the context of the project’s second objective, restoring forage fish habitat. The reduction in TVS, in what can be considered the sediment’s overall organic content,

is crucial to potentially restoring the site's historical natural processes. By reducing the organic content, anaerobic conditions are less likely to persist which should decrease the amount of toxic sulfides. As the intertidal zone is a dynamic environment, it is important that the monitoring of sediment characteristics continue in year two to assess whether changes to the site require any further analysis or management actions.

#### *Macroinvertebrate Assemblage Evaluation*

Macroinvertebrate sampling and analysis are scheduled to be performed at roughly the same time in year two of monitoring. A comparison of the results from years one and two will provide further insight into the ecology invertebrate communities.

#### *Forage Fish Spawning Surveys*

Previous observations of surf smelt spawning in unimpacted sections of the bay meant that improvements to the restoration site had the opportunity to establish more spawning habitat. Changes in sediment grain size appear to have been an important alteration towards facilitating surf smelt egg deposition at the site. Forage fish are known to reuse the spawning grounds that they have utilized in the past (Penttila 2007). It is likely to assume then that the restoration site may continue to provide suitable habitat for future spawning events. Forage fish spawning surveys will be continued in year two of monitoring for both sand lance and surf smelt to document any biological use of the project site.

#### *Observe for Eelgrass*

Observing for submerged underwater vegetation should be continued in future assessments to determine whether native eelgrass (*Zostera marina*) or other species have colonized the project site or other areas of Thatcher Bay. The Washington Department of Natural Resources' Nearshore Habitat Program is the agency tasked with documenting underwater vegetation in the Puget Sound (DNR 2016). Conversations with personnel at the WA DNR nearshore habitat program however, indicate that the likelihood of Thatcher Bay entering the DNR site sampling pool in the near future is minimal. It is not evident how likely it is that any existing eelgrass will move inward from the mouth of the bay. On multiple visits to the project site it was noted that water visibility was not much more than a few feet. As native eelgrass can be negatively



impacted by an insufficient amount of available light, it is recommended that measurements of available light also be taken throughout different areas of Thatcher Bay.

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## Appendix A: Macroinvertebrate results



**Figure 12.** MDS ordination showing patterns in the benthic and epibenthic invertebrate communities in the two study areas. Individual points represent single samples. Points that are closer together on the plot had more similar invertebrate assemblages. The stress value indicates that the MDS provided a good 2-dimensional representation of the community. Circles around points indicate 60% similarity as indicated by a group average cluster analysis.

Species checklist for invertebrates collected at the Thatcher Bay study site (Bingham 2015).

Phylum Cnidaria

Class Hydrozoa

Family Campanulariidae

*Obelia* sp.

Phylum Nematoda

Unidentified nematodes

Phylum Nemertea

Unidentified nemertean

Phylum Annelida

Class Polychaeta

Order Capitellida

Family Capitellidae

*Mediomastus* sp.

- Order Phyllodocida
  - Family Glyceridae
    - Glycera tenuis* Hartman, 1944
  - Family Goniadidae
    - Goniada* sp.
  - Family Nephtyidae
    - Nephtys* sp.
  - Family Nereidae
    - Nereis vexillosa* Grube, 1851
    - Nereis* sp.
  - Family Phyllodocidae
    - Eteone spetsbergensis* (Malmgren, 1865)
  - Family Polynoidae
    - Harmothoe* sp.
  - Family Syllidae
    - Exogone lourei* Berkeley & Berkeley, 1938
- Order Opheliida
  - Family Opheliidae
    - Armandia brevis* (Moore, 1906)
- Order Spionida
  - Family Spionidae
    - Pseudopolydora bassargensis* Zachs 1933
- Phylum Mollusca
  - Class Gastropoda
    - Family Littorinidae
      - Littorina* sp. egg capsules
    - Unidentified veliger larvae
  - Class Bivalvia
    - Order Mytiloida
      - Family Mytilidae
        - Modiolus modiolus* (Linnaeus, 1758)
      - Family Tellinidae
        - Macoma* sp.
- Phylum Arthropoda
  - Subphylum Chelicerata
    - Class Arachnida
      - Order Acari
        - Unidentified mites
  - Subphylum Crustacea
    - Class Branchiopoda
      - Family Podonidae
        - Evadne* sp.
        - Podon* sp.
    - Class Copepoda
      - Order Callanoida
        - Unidentified species
      - Order Harpacticoida

*Ectinosoma* sp.  
*Harpacticus* sp.  
*Nannopus* sp.  
*Orthopsyllus illgi* (Chappuis, 1958)  
*Tisbe* sp.

Class Cirripedia

- Unidentified barnacles
- Unidentified barnacle cyprids

Class Malacostraca

- Subclass Peracarida
  - Order Cumacea
    - Family Nannastacidae
      - Cumella vulgaris* (Hart, 1930)
  - Order Tanaidacea
    - Family Paratanaidae
      - Leptochelia* sp.
    - Family Tanaidae
      - Sinelobus stanfordi* (Richardson, 1901)
  - Order Amphipoda
    - Superfamily Corophioidea
      - Family Caprellidae
        - Caprella* sp.
    - Superfamily Gammaroidea
      - Family Anisogammaridae
        - Eogammarus* sp.
  - Order Isopoda
    - Suborder Flabellifera
      - Gnorimosphaeroma oregonense* Dana, 1854-55
  - Order Decapoda
    - Infraorder Brachyura
      - Family Cancridae
        - Cancer oregonensis* (Dana 1852)

Class Ostracoda

- Suborder Podocopida
  - Unidentified ostracode

Subphylum Hexapoda

- Class Entognatha
  - Subclass Collembola
    - Unidentified springtail species

## Appendix B: Forage Fish Survey Protocols

### WDFW Intertidal Forage Fish Spawning Habitat Survey Protocols

#### Procedures for obtaining bulk beach substrate samples

##### Field materials needed:

Measuring tape (100+ feet)

16-ounce plastic jar or large scoop

8 inch x 24 inch polyethylene bag (or large, sturdy ziplock)

Handheld GPS device

Tide table

Digital camera (optional)

Hypsometer (if available)

Data sheet (preprint on Write-in-the-Rain paper if possible)

Note: Sampling should occur on the lowest tide practicable. Prior to sampling any site consult tide tables to ensure you will be able to access the +7-9 (surf smelt) and +5-8 (sandlance) tidal height. It may also be necessary to obtain permission to access the beach from private or corporate landowners.

##### Procedure:

1. Upon arriving on the beach, fill out the header information on the attached data sheet. Do not fill in "Reviewed by." Before conducting the first sample, describe the character of the upland and beach environment using the codes provided on the back of the data sheet. For additional details on sample codes see Moulton and Penttila (2001)\*.
2. Identify a landmark from which you will measure the distance to the bulk substrate sample tidal elevation. Typical landmarks include the upland toe of the beach, the last high tide mark or wrack line, and the edge of the water.
3. Measure the distance from the landmark to the tidal elevation to be surveyed. Note that linear measurements along the beach face serve as an index of tidal height but do not directly quantify vertical tidal height. If available, a hypsometer can be used to measure vertical sampling height.
4. Stretch a measuring tape at least 100 feet along the selected tidal height. Note that beach contours may cause the landmark to be „wavy“ and that the tape should remain a consistent distance from the landmark.
5. Standing at one end of the measuring tape, record a GPS fix on the data sheet.
6. Using a 16-ounce sample jar or large scoop remove the top 5-10 cm (2-4 in) of sediment from the location recorded in Step 6 above. Place the sediment in an 8 inch x 24 inch polyethylene bag or large, sturdy ziplock. You may need to take two scoops to get sufficient sediment, depending on the coarseness of the beach.
7. Walk ten paces (single steps) along the measuring tape, repeat the sediment scooping action, and place the sediment in the bag. Move an additional ten paces and repeat. Move an additional ten paces, approximately to the end of the tape, and repeat. The bag should now have sediment from four locations along the tape and be at least  $\frac{1}{2}$  to  $\frac{2}{3}$  full.
8. If additional transects, representing various tidal heights, along the beach are to be surveyed, place the sample bag in a cool, shady place and repeat the above procedures at these additional locations. If no additional samples will be taken, move on to wet sieving and winnowing the sample as described in the companion protocol "Procedures for recovering "winnowed light fractions" subsamples of forage fish egg-sized material from bulk beach substrate samples."
9. If you have a camera, take several photos of the survey area showing sampling locations. Be sure to take photos from several perspectives (i.e., both up and down, as well as along, the beach). For each photo, record the cardinal direction you are facing on the data sheet in the comments field.



## Appendix C: Sediment Analysis Protocols (PSEP 1986)

TABLE 2. RECOMMENDED SAMPLE SIZES, CONTAINERS, PRESERVATION TECHNIQUES, AND HOLDING TIMES FOR SEDIMENT CONVENTIONAL VARIABLES

Variable	Minimum Sample Size (g) <sup>a</sup>	Container <sup>b</sup>	Preservation	Maximum Holding Time
Particle size	100-150 <sup>c</sup>	P,G	Cool, 4° C	6 mo <sup>d</sup>
Total solids	50	P,G	Freeze	6 mo <sup>d</sup>
Total volatile solids	50	P,G	Freeze	6 mo <sup>d</sup>
Total organic carbon	25	P,G	Freeze	6 mo <sup>d</sup>
Oil and grease	100	G only	Cool, 4° C, HCl; Freeze	28 days <sup>d</sup> 6 mo <sup>d</sup>
Total sulfides	50	P,G	Cool, 4° C, 1N zinc acetate	7 days <sup>d</sup>
Total nitrogen	25	P,G	Freeze	6 mo <sup>d</sup>
Biochemical oxygen demand	50	P,G	Cool, 4° C	7 days <sup>e</sup>
Chemical oxygen demand	50	P,G	Cool, 4° C	7 days <sup>e</sup>

## PARTICLE SIZE

### USE AND LIMITATIONS

Particle size is used to characterize the physical characteristics of sediments. Because particle size influences both chemical and biological variables, it can be used to normalize chemical concentrations according to sediment characteristics and to account for some of the variability found in biological assemblages. Particle size is also an important variable for marine engineering purposes. In addition to Plumb (1981), a variety of other references discuss the uses and measurement of particle size (e.g., Krumbein and Pettijohn 1938; Folk 1968; Buchanan 1984).

Particle size can be characterized in a wide range of detail. The grossest divisions that generally are considered useful for characterizing particle size distributions are percentages of gravel, sand, silt, and clay. However, each of these size fractions can be subdivided further so that additional characteristics of the size distribution (e.g., mean diameter, skewness, kurtosis) can be determined.

Particle size determinations can either include or exclude organic material. If organic material is removed prior to analysis, the "true" (i.e., primarily inorganic) particle size distribution is determined. If organic material is included in the analysis, the "apparent" (i.e., organic plus inorganic) particle size distribution is determined. Because true and apparent distributions may differ, detailed comparisons between samples analyzed by these different methods are questionable. It is therefore desirable that all samples within each study (at a minimum) and among different studies (if possible) be analyzed using only one of these two methods.

### FIELD PROCEDURES

#### Collection

Samples can be collected in glass or plastic containers. A minimum sample size of 100-150 g is recommended. If unrepresentative material is to be removed from the sample, it should be removed in the field under the supervision of the chief scientist and noted on the field log sheet.

### Processing

Samples should be stored at 4° C, and can be held for up to 6 mo before analysis. Samples must not be frozen or dried prior to analysis, as either process may change the particle size distribution.

### LABORATORY PROCEDURES

#### Analytical Procedures

- Equipment
  - Sieve shaker
    - Ro-Tap or equivalent
  - Drying oven
  - Constant temperature bath
  - Analytical balance
    - 0.1 mg accuracy
  - Desiccator
  - Clock
    - With second hand
  - Standard sieves
    - Appropriate mesh sizes
  - Sieve pan and top
  - Sieve brush
  - Funnel
  - 1-L graduated cylinders
  - 50-mL beakers
  - 20-mL pipets
  - Water pique or squirt bottle
  - Glossy paper
  - Dispersant
    - 1 percent sodium hexametaphosphate = 1 percent commercially available Calgon
  - Distilled water.
- Sample preparation
  - Allow samples to warm to room temperature.
  - Homogenize each sample mechanically, incorporating any overlying water.
  - Remove a representative aliquot (approximately 25 g) and analyze for total solids content. This information can be used to estimate the dry weight of the aliquot used for particle size analysis. The efficiency of the entire analysis can then be evaluated by adding the dry weights of all sample fractions and comparing this sum with the estimated dry weight of the original aliquot.
  - Remove a second representative aliquot for wet sieving. The aliquot can range from 20 g for muddy sediments to 100 g for sandy sediments. The critical factor for sample size determination is

the weight of fine-grained material that will be used for the pipet analysis. Ideally the total dry weight of fine-grained material in the 1-L graduated cylinder should equal approximately 15 g. However, total weights between 5 and 25 g are considered acceptable. Total weights outside this range are not considered acceptable and it is recommended that aliquot size be modified to bring the amount of fine-grained material into the acceptable range.

- Weigh the wet sample to the nearest 0.01 g.
- Organics oxidation - this step removes organic material from the sample. It is optional and depends upon the objectives of each study.
  - Place the sediment sample in a large beaker ( $\geq 2$  L).
  - Add 20 mL of 10 percent hydrogen peroxide solution and mix.
  - Let the sample stand until frothing stops.
  - Once frothing stops, add an additional 10 mL of hydrogen peroxide solution.
  - Continue adding 10-mL portions of hydrogen peroxide solution until no frothing occurs on addition.
  - Boil the sample to remove any excess hydrogen peroxide.
  - Be careful that material is not lost from the beaker during frothing and boiling.
- Wet-sieving - this step separates the sample into size fractions greater than 62.5  $\mu\text{m}$  (i.e., sand and gravel) and less than 62.5  $\mu\text{m}$  (i.e., silt and clay)
  - Place the 62.5- $\mu\text{m}$  (4 phi) sieve in a funnel, with a 1-L graduated cylinder underneath. Moisten the sieve using a light spray of distilled water.
  - Place the sample in a beaker, add 20-30 mL of distilled water, and stir to suspend fine-grained material.
  - Pour the sample into the sieve and thoroughly rinse the beaker and stirrer with distilled water.
  - Wash the sediment on the sieve with distilled water using a water pique or squirt bottle having low water pressure. Aggregates can be gently broken using a rubber policeman.
  - Continue wet sieving until only clear water passes through the sieve. Try to ensure that the rinsate does not exceed approximately 950 mL. This can generally be accomplished by sieving a sample quantity that is not too large and by efficient use of the rinse water. Both of these techniques may require experimentation before routine wet sieving is started.
- Gravel-sand fraction - this fraction is subdivided further by mechanically dry sieving it through a graded series of screens.
  - Wash the coarse fraction into a preweighed 50-mL beaker using distilled water. Rinse the sieve thoroughly.

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- Dry the coarse fraction to constant weight at  $90 \pm 2^\circ \text{C}$ . The drying temperature is less than  $100^\circ \text{C}$  to prevent boiling and potential loss of sample.
  - Cool the sample to room temperature in a desiccator.
  - Weigh the cooled sample to the nearest 0.1 mg.
  - Set up a nest of sieves that will divide the coarse fraction into the desired number of subfractions. Set up the sieves in a graded series of mesh sizes, with the coarsest mesh on top and the finest mesh on the bottom. The bottom sieve always should have a mesh size of 62.5  $\mu\text{m}$  (4 phi). Place a solid pan on the bottom of the stack and a lid on top of the stack. At a minimum, the coarse fraction should be separated into gravel and sand fractions, using a sieve with a mesh size of 2 mm (-1 phi).
  - Add the sample to the uppermost sieve. Complete transfer can be ensured by using a sieve brush to remove any material adhering to the beaker. The sieve brush can also be used to gently break up aggregated sediment.
  - Shake mechanically for exactly 15 min using the Ro-Tap (or equivalent). A shaker having an automatic timer is preferable.
  - After shaking, empty the contents of each sieve onto a glossy piece of paper (e.g., wax paper). To empty a sieve, invert it and tap it on the table several times while ensuring that all edges hit the table at the same time. If the sieve is not tapped evenly, the meshes may be distorted. After tapping the sieve, ensure complete removal of the sample by brushing the back of the screen. After brushing the back of the screen, turn the sieve over and brush out any particles adhering to the sides of the sieve or the inside of the screen.
  - Add the fraction that passed through the bottom sieve (e.g., 4 phi) and was retained by the solid pan to the silt-clay fraction of that sample.
  - Weigh each remaining size fraction to the nearest 0.1 mg.
  - Sum the weights of all size fractions and compare the result with the initial weight of the coarse fraction. Losses and inaccuracies should be less than 5 percent of the initial weight. Losses and inaccuracies tend to increase with increasing number of fractions.
  - Large amounts of organically derived fragments (e.g., wood debris, grass, shells) or any unusual material in any size fraction should be noted on the laboratory log sheet.
- Silt-clay fraction - this fraction is subdivided further using a pipet technique that depends upon the differential settling rates of different particles. Because additions to this fraction may be made after mechanical sieving of the gravel-sand fraction (see above), it is recommended that the silt-clay analysis for each sample not be conducted until the gravel-sand analysis has been completed.
    - Add 10 mL of the dispersant to 990 mL of distilled water. Determine the weight of dispersant in a 20-mL aliquot of this

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mixture by pipeting a 20-mL aliquot into each of five tared beakers, drying the samples to constant weight at  $90 \pm 2^\circ \text{C}$ , cooling the samples in a desiccator, weighing the cooled samples, and calculating the mean weight of dispersant in the five samples. This weight multiplied by the number of 10-mL additions of dispersant to each sample will be subtracted from the weight of each sediment fraction at the end of the pipet analysis.

- Add 10 mL of the dispersant to each sample suspension in the 1-L graduated cylinders.
- Mix each suspension by either stoppering and inverting the cylinder or by using the up and down motion of a perforated disc plunger.
- Allow the mixed suspension to stand for 2-3 h and check for signs of flocculation. Flocculation can be recognized by a curdling and rapid settling of lumps of particles or by the presence of a thick soupy layer on the bottom of the cylinder passing abruptly into clear water above.
- If flocculation occurs, add dispersant in 10-mL increments until no noticeable flocculation occurs. Record the volume of dispersant added.
- When ready to conduct the pipet analysis, bring the sample volume to 1 L by adding distilled water, mix the suspension thoroughly, and place the cylinder in a constant-temperature water bath. If the volume is greater or less than 1 L, the factor for converting the weight of the sediment in each 20-mL aliquot to that in the total volume must be modified accordingly.
- After 20 sec, withdraw a 20-mL aliquot from a depth of 20 cm below the surface of the suspension using a pipet. The pipet should be marked for the specified sampling depths and should be inserted vertically into the settling cylinder when the aliquot is taken. A suction bulb may be used on the open end of the pipet to facilitate sampling. It is critical that the suspension be disturbed as little as possible when pipet aliquots are taken.
- Transfer the 20-mL aliquot to a preweighed 50-mL beaker. Rinse the pipet into the beaker using 20 mL of distilled water.
- Withdraw 20-mL aliquots at a depth of 10 cm below the surface of the suspension at the appropriate time(s) listed in Table 3. A formula for calculating withdrawal times is given by Folk (1968) and Buchanan (1984). If a withdrawal is missed, the suspension can be stirred again and the missed withdrawal can be taken at the appropriate time after settling begins. It is not necessary to withdraw the initial 20-mL aliquot when this corrective action is conducted.
- Transfer these additional 20-mL aliquots to 50-mL preweighed beakers, each time rinsing the pipet into the respective beaker using 20 mL of distilled water.
- Dry all aliquots to constant weight at  $90 \pm 2^\circ \text{C}$ . A drying temperature less than  $100^\circ \text{C}$  is used to prevent boiling and potential loss of sample.

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- Cool dried samples to room temperature in a desiccator.
- Weigh cooled samples to the nearest 0.1 mg.

- Calculations

- The total weight of a phi-size interval in the 1-L graduated cylinder is determined as follows:

$$\text{Phi weight (g dry weight)} = 50[(A-C)-(B-C)]$$

Where:

- A = weight (g) of residue in a 20-mL aliquot for a given phi-size boundary
- B = weight (g) residue in a 20-mL aliquot for the next larger phi-size boundary
- C = mean weight (g) of dispersant in a 20-mL aliquot.

QA/QC Procedures

It is critical that each sample be homogenized thoroughly in the laboratory before a subsample is taken for analysis. Laboratory homogenization should be conducted even if samples were homogenized in the field.

After dry-sieving a sample, all material must be removed from the sieve. This can be accomplished by tapping the rim of the sieve evenly on a hard surface and by brushing the screen.

The total amount of fine-grained material used for pipet analysis should be 5-25 g. If more material is used, particles may interfere with each other during settling and the possibility of flocculation may be enhanced. If less material is used, the experimental error in weighing becomes large relative to the sample size.

Before pipet extractions can be made, the sample must be homogenized thoroughly within the settling cylinder. Once the pipet analysis begins, the settling cylinders must not be disturbed, as this will alter particle settling velocities. Care must be taken to disturb the sample as little as possible when pipet extractions are made.

After a pipet extract has been transferred to a drying beaker, any sample adhering to the inside of the pipet must be removed. This can be accomplished by drawing 20 mL of distilled water into the pipet and adding this rinse water to the drying beaker.

Dried samples should be cooled in a desiccator and held there until they are weighed. If a desiccator is not used, the sediment will accumulate ambient moisture and the sample weight will be overestimated. A color-indicating desiccant is recommended so that spent desiccant can be detected



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easily. Also, the seal on the desiccator should be checked periodically, and, if necessary, the ground glass rims should be greased or the "O" rings should be replaced.

It is recommended that triplicate analyses be conducted on one of every 20 samples, or on one sample per batch if less than 20 samples are analyzed. It is also recommended that the analytical balance, drying oven, and temperature bath be inspected daily and calibrated at least once per week.

DATA REPORTING REQUIREMENTS

The weight of each sediment fraction should be reported to the nearest 0.0001 g dry weight. The laboratory should report the results of all samples analyzed (including QA replicates) and should note any problems that may have influenced data quality.

## TOTAL SOLIDS

### USE AND LIMITATIONS

Total solids are the organic and inorganic materials remaining after a sample has been dried completely. This variable is commonly used to convert sediment concentrations of substances from a wet-weight to a dry-weight basis. It typically is measured in conjunction with other variables.

Total solids values are operationally defined, because results depend on drying temperatures. For example, temperature-dependent weight losses occur from volatilization of organic matter, mechanically occluded water, water of crystallization, and gases from heat-induced chemical decomposition. By contrast, weight gains may result from oxidation processes. To provide data that are comparable among different studies, it is therefore critical that drying temperatures be standardized.

### FIELD PROCEDURES

#### Collection

Samples can be collected in glass or plastic containers. Samples can be collected in the same containers as samples for other variables, if total solids is to be measured in conjunction with those variables. A minimum sample size of 50 g is recommended. If unrepresentative material is to be removed from the sample, it should be removed in the field under the supervision of the chief scientist and noted on the field log sheet.

#### Processing

Samples should be stored frozen and can be held for up to 6 mo under that condition.

### LABORATORY PROCEDURES

#### Analytical Procedures

- Equipment
  - Muffle furnace  
550° C capacity
  - Drying oven
  - Desiccator

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- Analytical balance  
0.01 g accuracy
- 100-mL evaporating dishes  
Porcelain, platinum, or Vycor.
- Equipment preparation
  - Ignite clean evaporating dishes at 550 + 10° C for 1 h in a muffle furnace to remove any remaining organic material.
  - Cool ignited dishes to room temperature in a desiccator.
  - Weigh each cooled dish to the nearest 0.01 g and store in the desiccator.
- Sample preparation
  - Allow frozen sediment samples to warm to room temperature
  - Homogenize each sample mechanically, incorporating any overlying water.
  - Transfer a representative subsample (approximately 25 g) to a preweighed evaporation dish.
  - Weigh the undried sample to the nearest 0.01 g.
- Analytical procedures
  - Dry the sample to constant weight at 103 + 2° C.
  - Cool the dried sample to room temperature in a desiccator.
  - Weigh the cooled sample to the nearest 0.01 g.
- Calculations
  - Total solids content is determined as follows:

$$\text{Percent solids} = \frac{(A-B)(100)}{C-B}$$

Where:

A = weight (g) of dish and dry sample residue  
B = weight (g) of dish  
C = weight (g) of dish and wet sample.

QA/QC Procedures

It is critical that each sample be thoroughly homogenized in the laboratory before a subsample is taken for analysis. Laboratory homogenization should be conducted even if samples were homogenized in the field.

Evaporating dishes must be ignited at 550° C before being used for total solids analysis. This step ensures that dishes are free from organic contaminants.

Dried samples should be cooled in a desiccator and held there until they are weighed. If a desiccator is not used, the sediment will accumulate

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ambient moisture and the sample weight will be overestimated. A color-indicating desiccant is recommended so that spent desiccant can be detected easily. Also, the seal on the desiccator should be checked periodically and, if necessary, the ground glass rims should be greased or the "O" rings should be replaced.

It is recommended that triplicate analyses be conducted on one of every 20 samples or on one sample per batch if less than 20 samples are analyzed. It is also recommended that the analytical balance and drying oven be inspected daily and calibrated at least once per week.

DATA REPORTING REQUIREMENTS

Total solids should be reported as a percentage of the wet weight of the sample to the nearest 0.1 unit. The laboratory should report the results of all samples analyzed (including QA replicates) and should note any problems that may have influenced sample quality.

## TOTAL VOLATILE SOLIDS (TVS)

### USE AND LIMITATIONS

Total volatile solids represent the fraction of total solids that are lost on ignition at a higher temperature than that used to determine total solids. Total volatile solids is used as a crude estimate of the amount of organic matter in the total solids.

Total volatile solids is operationally defined by the ignition temperature. Total volatile solids content does not always represent the organic content of a sample because some organic material may be lost at the drying temperature and some inorganic material (e.g., carbonates, chlorides) may be lost at the ignition temperature. Because of the temperature dependence of total volatile solids, valid interstudy comparisons require the use of standardized drying and ignition temperatures.

### FIELD PROCEDURES

#### Collection

Samples can be collected in glass or plastic containers. A minimum sample size of 50 g is recommended. If unrepresentative material is to be removed from the sample, it should be removed in the field under the supervision of the chief scientist and noted on the field log sheet.

#### Processing

Samples should be stored frozen and can be held for up to 6 mo under that condition.

### LABORATORY PROCEDURES

#### Analytical Procedures

- Equipment
  - Muffle furnace  
550° C capacity
  - Drying oven
  - Desiccator
  - Analytical balance  
0.01 g accuracy
  - 100-mL evaporating dishes  
Porcelain, platinum, or Vycor.

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- Equipment preparation
  - Ignite clean evaporating dishes at  $550 \pm 10^\circ \text{C}$  for 1 h in a muffle furnace to remove any remaining organic material.
  - Cool ignited dishes to room temperature in a desiccator.
  - Weigh each cooled dish to the nearest 0.01 g and store in the desiccator.
- Sample preparation
  - Allow frozen sediment samples to warm to room temperature.
  - Homogenize each sample mechanically, incorporating any overlying water.
  - Transfer a representative subsample (approximately 25 g) to a preweighed evaporating dish.
- Analytical procedures
  - Dry the sample to constant weight at  $103 \pm 2^\circ \text{C}$ .
  - Cool the dried sample to room temperature in a desiccator.
  - Weigh the cooled sample to the nearest 0.01 g.
  - Ignite the sample at  $550 \pm 10^\circ \text{C}$  to constant weight. Make sure that the samples do not flare up when placed in the oven, as sediment may be lost from the crucibles. If sample flashing is a problem, it is recommended that the muffle furnace be cooler than  $550^\circ \text{C}$  when samples are placed inside, and that the temperature gradually be increased to  $550^\circ \text{C}$ .
  - Weigh each cooled sample to the nearest 0.01 g.
- Calculations
  - TVS content is determined as follows:

$$\text{Percent TVS} = \frac{(A-C)100}{A-B}$$

Where:

A = weight (g) of dish and dry sample residue  
B = weight (g) of evaporation dish  
C = weight (g) of dish and ignition residue.

QA/QC Procedures

It is critical that each sample be thoroughly homogenized in the laboratory before a subsample is taken for analysis. Laboratory homogenization should be conducted even if samples were homogenized in the field.

Evaporating dishes (or crucibles) must be ignited at  $550^\circ \text{C}$  before being used for total volatile solids analysis. This step ensures that the dishes are free from volatile contaminants.

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Dried and combusted samples should be cooled in a desiccator and held there until they are weighed. If a desiccator is not used, the sediment will accumulate ambient moisture and the sample weight will be overestimated. A color-indicating desiccant is recommended so that spent desiccant can be detected easily. Also, the seal on the desiccator should be checked periodically and, if necessary, the ground glass rims should be greased or the "O" rings should be replaced.

It is recommended that triplicate analyses be conducted on one of every 20 samples or on one sample per batch if less than 20 samples are analyzed. It is also recommended that the analytical balance, drying oven, and muffle furnace be inspected daily and calibrated at least once per week.

#### DATA REPORTING REQUIREMENTS

Total volatile solids should be reported as a percentage of the dry weight of the uncombusted sample to the nearest 0.1 unit. The laboratory should report the results of all samples analyzed (including QA replicates) and should note any problems that may have influenced data quality.