

Hakai Nearshore Marine Ecology Rocky Intertidal Survey Methods



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Introduction

The Hakai Nearshore Research Program documents long term observations of ecosystem, community and population change in nearshore habitats, and addresses the factors driving spatial and temporal variation in these systems. Rocky shores account for more than 33% of the world's coastal shorelines (Johnson, 1988), and are amongst the dominant coastal habitats in British Columbia, extending along over 50% of the BC coastline (Reshitnyk, pers. comm.). Rocky shore intertidal benches are biodiverse, serving as important habitat for sessile invertebrates, nursery and feeding habitat for fish and crustaceans (Rilov and Schiel, 2006; Dias et. al, 2016) and feeding and roosting ground for birds (Marsh, 1986). Characterized by strong environmental gradients and interacting species competing for space and other resources, rocky shores provide an excellent backdrop to evaluate climate change effects, scaling between individual, population, community and ecosystem level effects. Many rocky shore intertidal species are indicators of environmentally driven change to marine ecological communities. As such, they act as sentinels to the biological costs of climate change (Denny, 1987; Helmuth et. al., 2002, Harley, 2011).

The Hakai Institute Rocky Intertidal Program aims to evaluate the trends and key drivers of change within the rocky shore intertidal ecosystem on Calvert Island, BC. We are dedicated to documenting annual trends in biodiversity and annual and seasonal trends of indicator species abundance to achieve focal research objectives. These data provide an essential resource for us to determine changes in the intertidal community under continual exposure to a changing climate. This document focuses on methods for documenting temporal and spatial dynamics within the rocky intertidal ecosystem.

Hakai Rocky Intertidal Research Objectives

- Track seasonal and annual changes in indicator species productivity and biodiversity
- Determine the drivers of change in rocky intertidal ecosystems
- Assess resilience of rocky intertidal species to disturbance, stress and disease
- Understand connectivity between rocky intertidal ecosystems and other coastal ecosystems

Focal Research Questions:

1. What are the seasonal and interannual drivers of variability in abundance and distribution of rocky intertidal indicator species (barnacles, mussels, *Fucus* spp., surfgrass)?

2. Does rocky intertidal habitat complexity drive mobile invertebrate biodiversity?
3. Are rocky intertidal habitats on Calvert Island resilient to disturbance, stress and disease?
4. What are the costs of asteroid idiopathic wasting syndrome for intertidal sea stars and their community?

Hakai Central Coast Rocky Intertidal Study Sites

Nearshore Marine Ecology rocky intertidal study sites on exposed coast were established in 2016 to encompass sufficient habitat in the low, mid and high intertidal zones in order to conduct long-term research. Sites were selected to monitor sessile invertebrate and macroalgal substrate coverage, surfgrass meadow characteristics, macro-invertebrate biodiversity, density, abundance and disease status and water chemistry characteristics. Methods were developed with a goal of data comparison capability across Hakai Institute projects and habitats. Proximity to the Calvert Island Ecological Observatory was also a factor in site selection, as the exposed rocky coastline complicates boat access.

Seasonal observations of indicator species abundance and the diversity of their associated mobile invertebrate communities were conducted (February, May, June, August, November) at rocky intertidal sites ($n=3$) on Calvert Island (Fig. 1, red squares).



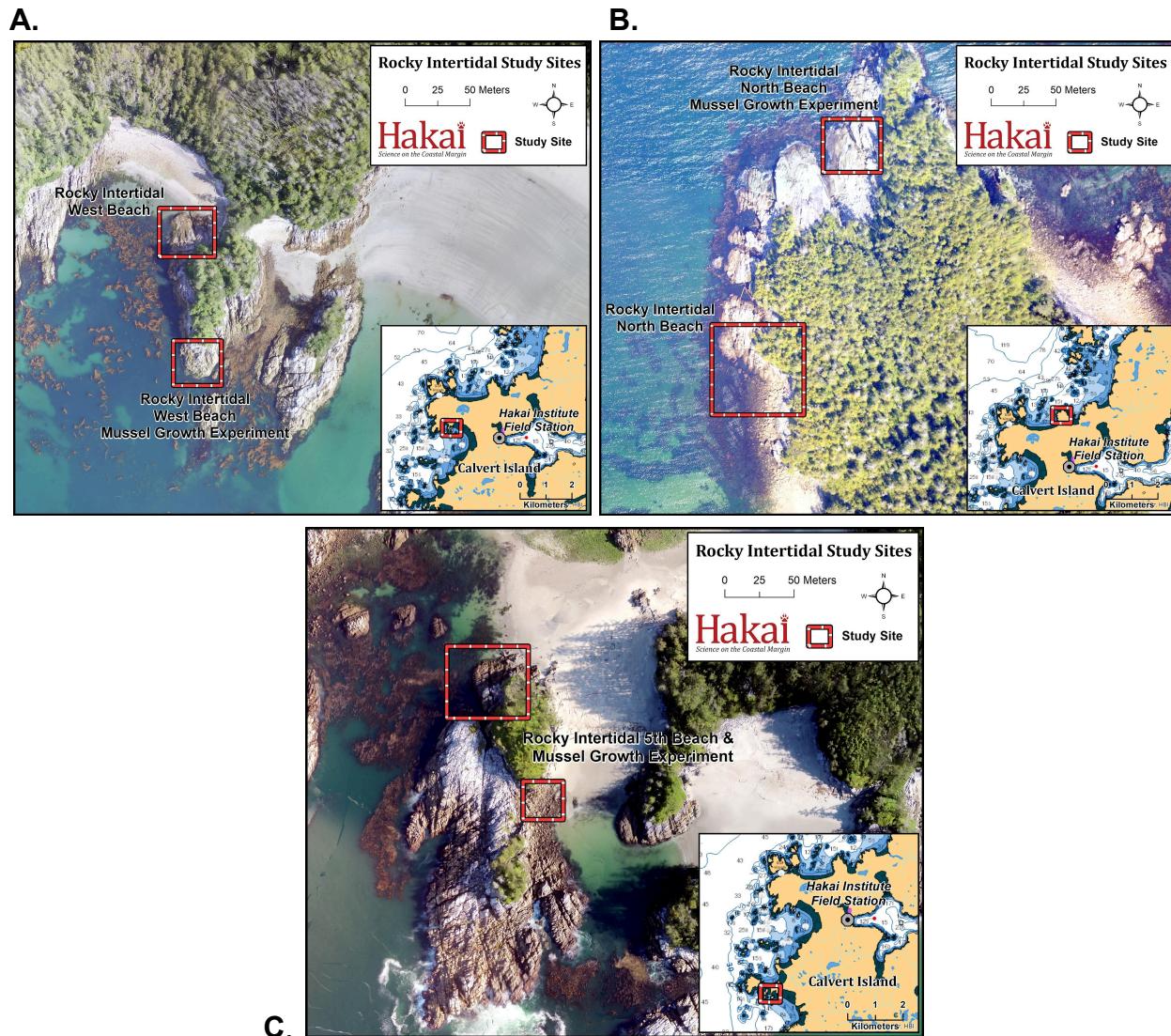


Figure 1. Map of rocky intertidal study sites and mussel growth experiments (red) on Calvert Island, British Columbia. West Beach (A), North Beach (B) and Fifth Beach (C) study sites have been surveyed since 2016, while the mussel growth experiments began in 2017.



Figure 2. Map of Fifth Beach rocky intertidal and mussel growth experiment study site (51.64068° N, -128.156779° W) including bolt locations with modeled elevations (m) on Calvert Island, British Columbia. Pink lines indicate permanent plots (75cm x 50cm). **Top right:** Overview of full site with species specific survey locations indicated in subset boxes. Blue box indicates Fucus spp. permanent plots and temperature logger. Green box indicates barnacle permanent plots, surfgrass bolts and Martone Lab permanent bolts. Red box indicates mussel permanent plots and mussel experiment. **Top left:** Fucus spp. plots 1-5 and temperature logger location with modeled elevations (m). **Bottom:** Barnacle permanent plots 1-5, surfgrass transect and Martone Lab permanent bolts with modeled elevations (m). **Bottom inset (red box):** Mussel permanent plots 1-5 and mussel growth experiment locations with modeled elevations (m).

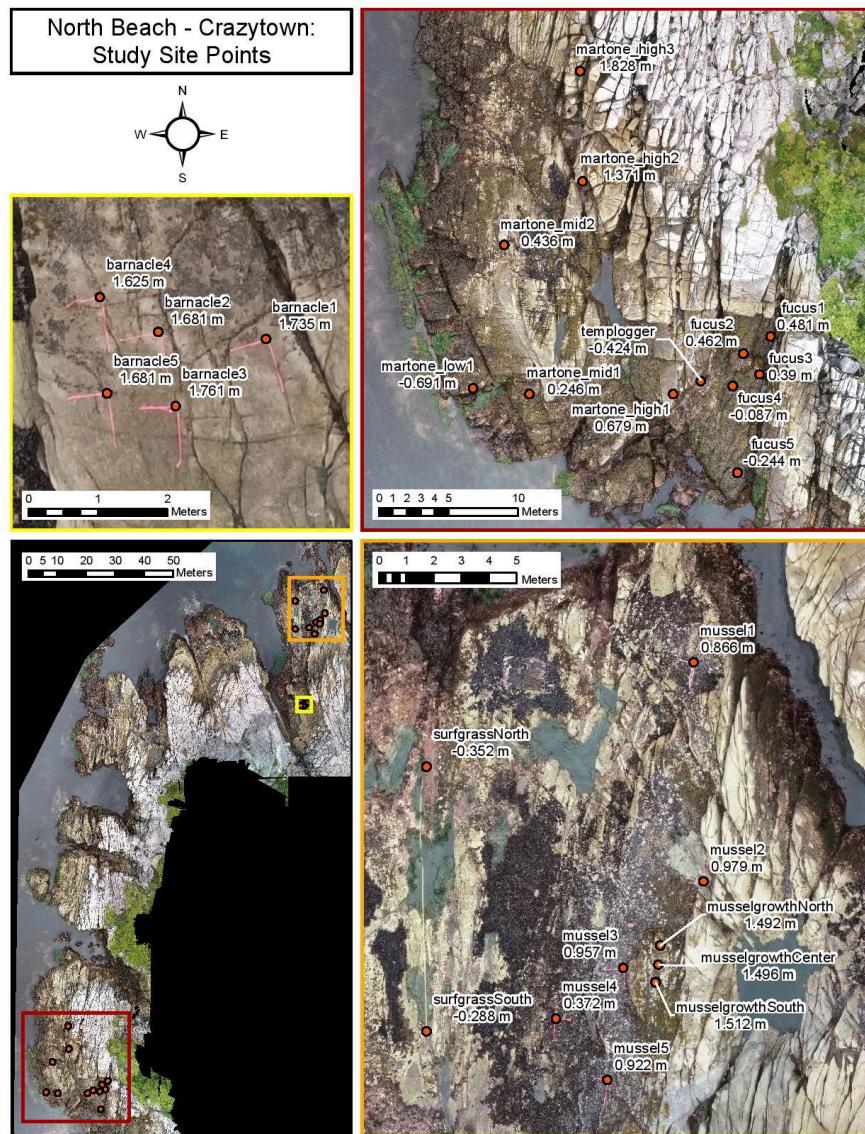


Figure 3. Map of North Beach and Cazytown rocky intertidal and mussel growth experiment study site (51.66508° N, -128.1347° W) including bolt locations with modeled elevations (m) on Calvert Island, British Columbia. Pink lines indicate permanent plots (75cm x 50 cm). **Bottom left:** Overview of full site with species specific survey locations indicated in subset boxes. Red box indicates *Fucus* spp. permanent plots, temperature logger and Martone Lab permanent bolt locations at North Beach. Yellow box indicates barnacle permanent plots. Orange box indicates mussel permanent plots, surfgrass transect and mussel growth experiment locations. **Top left:** Barnacle plots 1-5 with modeled elevations (m) at Cazytown. **Top right:** *Fucus* spp. permanent plots, temperature logger and Martone Lab permanent bolt locations with modeled elevations (m) at North Beach. **Bottom right:** Mussel plots 1-5, surfgrass transect and mussel growth experiment locations with modeled elevations (m) at Cazytown.

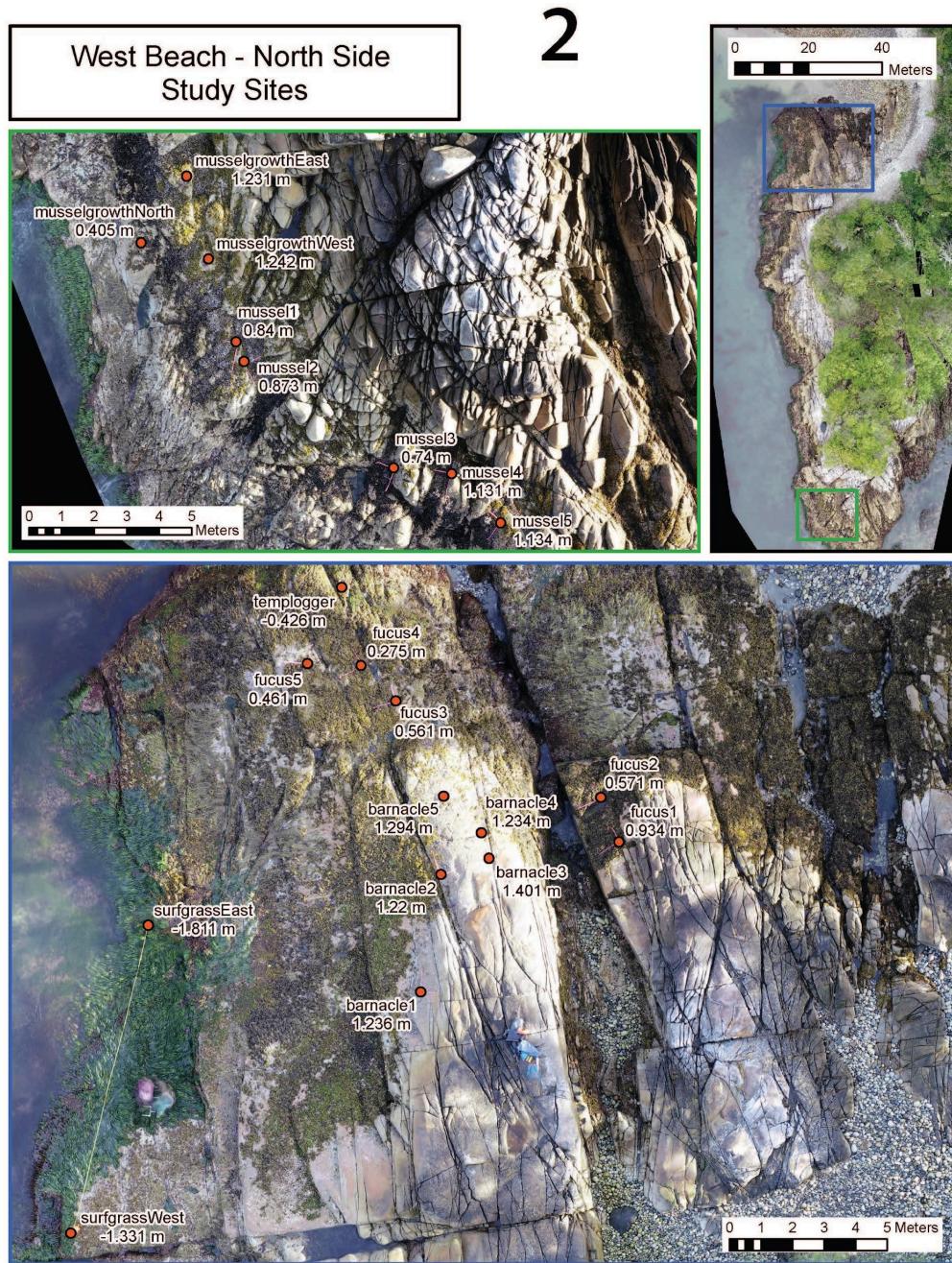


Figure 4. Map of West Beach rocky intertidal and mussel growth experiment study site ($51.657423^{\circ}\text{N}$, $-128.148814^{\circ}\text{W}$) including bolt locations with modeled elevations (m) on Calvert Island, British Columbia. Pink lines indicate permanent plots (75cm x 50 cm).

Top right: Overview of full site with species specific survey locations indicated in subset boxes. Blue box indicates barnacle and fucus permanent plots, surfgrass transect and temperature logger. Green box indicates permanent mussel plots and mussel growth experiment. **Top left:** Mussel plots 1-5 and mussel growth experiment locations with modeled elevations (m). **Bottom:** Barnacle plots 1-5, Fucus spp. plots 1-5 and surfgrass transect bolt locations with modeled elevations (m).

Survey Design

At Hakai Nearshore Marine Ecology rocky intertidal study sites ($n=3$), field technicians conduct plot and transect-level surveys at permanent locations targeting specific environmental conditions (e.g. tidal heights, exposure). To evaluate change over time, permanent 50cm x 75cm plots ($n=15$ per site) delineated by bolts drilled into intertidal rock have been installed in 3 zones classified by sessile invertebrate and macroalgal substrate coverage: barnacle ($n=5$), mussel ($n=5$) and *Fucus* spp. ($n=5$). Surfgrass communities are observed along fixed 10m transects that run parallel to shore ($n=1$ transect per site). Seastar and other macro-invertebrate swath surveys are conducted in the high, mid and low intertidal zones within a 30m boundary horizontal to the shoreline. Intertidal temperature loggers associated with each habitat type ($n=4$ per site) have been installed to monitor changes in ocean conditions (Figure 5).



Figure 5. Temperature logger installation at Fifth Beach. Protective cages have been installed to prevent damage. Temperature loggers record temperature every 10 minutes year round. Data is downloaded regularly.

Survey Protocol Overview

During each seasonal rocky intertidal site visit, Hakai Institute field technicians conduct plot and transect-level surveys to record sessile invertebrate and macroalgae substrate coverage, *Phyllospadix* spp. meadow characteristics, macro-invertebrate biodiversity, density, abundance and disease status and water chemistry characteristics (Table 2).

Table 2. Surveys conducted at rocky intertidal sites and respective metrics obtained.

Survey	Frequency	Level	Parameters Collected	
			Field	Lab
Phyllospadix spp. cover and biomass	Seasonal	Transect	Record: percent cover, bed thickness, shoot density, flowering shoot density, epiphytes, canopy height	
<i>Pisaster ochraceus</i>	Seasonal Annual	Swath	Record: density, abundance, size, wasting disease status	
<i>Katharina tunicata</i>	Seasonal	Swath	Record: density, abundance, size	
All Macro-invertebrates	Seasonal	Swath	Record: density, species, abundance, size	
Barnacle abundance and diversity	Seasonal	Plot	abundance and diversity of <i>Balanus glandula</i> , <i>Semibalanus cariosus</i> , and <i>Chthamalus</i> spp.	
Mussel abundance	Seasonal	Plot	Record: abundance, length, bed depth, growth of <i>Mytilus californianus</i>	
Fucus cover & biomass	Seasonal	Plot	Record: <i>Fucus distichus</i> percent cover and diversity of other algae Collect: algal specimens that technicians are unable to properly identify in the field	Prepare herbarium specimens for assistance with identification at a later date

Mobile invertebrate diversity	Seasonal	Plot	Record: abundance, diversity, patchiness, substrate associations in barnacle, mussel, Fucus and Phyllospadix surveys
Water Chemistry	Seasonal	Site	Record: YSI measurements: temperature, conductivity, salinity, pH Collect: water samples

Field Protocols

Point Intercept Plot Monitoring

Research Objectives:

Do Fucus spp., barnacle (*Balanus glandula* and *Chthamalus dalli*), and mussel coverage change over time (seasonally and annually)?

Equipment

75 x 50 cm strung quadrat (x2)

Clipboard (x2)

Pencils

Data sheets (barnacles, mussels, fucus)

Knee pads

Site and plot number labels

Monitoring Parameters

- Photographic record of plots
- Percent cover of macrophytes (understory and overstory)
- Percent cover of sessile invertebrates (understory and overstory)

Set Up

At each site, point intercept surveys will be conducted on a total of 15 plots (target species: barnacle plots (x5), mussel plots (x5), fucus plots (x5)). Each plot is marked by 3 corner bolts and zip ties. The plot numbers are labelled by notches (1-5) engraved into the corner bolt of each plot, where the number of notches indicates the plot number. Each 75 x 50 cm strung quadrat consists of 100 point intercepts where the nylon string has been woven through to make a taught grid.

Point Intercept Plot Survey

Surveys require two technicians; an observer and a recorder. These surveys are **focused on sessile invertebrates and macrophytes, so motile invertebrates are not recorded.** If a motile invertebrate is present at a point-intercept, record what is underneath the motile invertebrate (e.g. rock). Avoid removing organisms from the

surface. Invertebrates such as limpets and chitons have lower survival rates after being pried off of rocks.

Photograph all plots (barnacle, mussel and Fucus; #1-5)

1. Photograph each permanent point intercept plot with the 50 x 75 cm quadrat on top. Place the 50 x 75 cm quadrat on top of the three corner bolts that denote the plot number for each plot type. The corner bolts should line up with the corners of the quadrat.
2. Write the plot number, plot type, site and date on the label plate with the dry erase marker. **If it is raining, make labels using duracopy paper beforehand.** Ensure the label is visible in the photo and the long (75 cm) side of the quadrat with 2 bolts is at the top of photo frame.

Other field technicians are encouraged to begin sampling while one technician begins taking photos.

Barnacle Dynamics: Point Intercepts

1. Place a 75 cm x 50 cm strung quadrat on top of barnacle point intercept plot #1 so that the 3 corner bolts line up with the corners of the quadrat, with the long (75 cm) side of the quadrat at the top.
2. Starting at the top left point intercept of the 75 x 50 cm strung quadrat, record the understory species that is present directly underneath the intersection of the strings for 50 points on the grid. **The understory species refers to the organism that is attached to the substrate.** For barnacle plots, the understory species must be a sessile invertebrate or algae (e.g. *Balanus glandula*, *Chthamalus dalli*, *Semibalanus cariosus*, *Fucus distichus*, *Pyropia spp.* etc.). Dead remnants of sessile species are also recorded (e.g. dead *Balanus glandula*). If there is no invertebrate or algae present, record the substrate (eg. rock). Do not record the presence of mobile invertebrates (record the substrate underneath the organism (eg. rock)), except if mobile invertebrates with limited mobility (e.g. limpets and chitons). Do not move mobile invertebrates with limited motility, in this case record the organism present as the understory species.
3. If there is another species (sessile invertebrate or macrophyte) growing on top of or covering the understory species at a point intercept, record this as the **overstory species**.

It is important to record these data in the correct order so the grid on the data sheet matches the grid on the strung quadrat. Record odd point intercepts for columns 1, 3, 5, 7 and 9. Record even point intercepts for columns 2, 4, 6, 8 and

10. Start at the top left point intercept and continue on downwards, working to the right.

Note: During winter sampling, the number of point intercepts recorded is scaled back to 25 points. Record odd point intercepts for columns 1,5 and 9. Record even point intercepts for columns 3 and 7.

4. Repeat steps 1 and 2 with barnacle point intercept plots #2, #3, #4 and #5.



Figure 6. Barnacle Plot within 50 cm by 75 cm quadrat.

Mussel Dynamics: Point Intercepts

1. Place a 75 x 50 cm strung quadrat on top of mussel point intercept plot #1 so that the 3 corner bolts line up with the corners of the quadrat, with the long (75 cm) side of the quadrat at the top.
2. Starting at the top left point intercept of the 75 x 50 cm strung quadrat, record the understory species that is present directly underneath the intersection of the strings for 50 points on the grid. **The understory species refers to the organism that is attached to the substrate.** For mussel plots, the understory species must be a sessile species (e.g.. *Mytilus californianus*, *Balanus glandula*, *Chthamalus dalli*, *Semibalanus cariosus*, *Pollicipes pollicipes*, etc.). Dead remnants of sessile species are also recorded (e.g. dead *M. californianus*). If there is no animal present, record the substrate (eg. rock). Do not record the presence of mobile invertebrates (record the substrate underneath the organism (eg. rock)). Do not move mobile invertebrates with limited motility (e.g. limpets and chitons). In this case, record the organism present as the understory species.

It is important to record these data in the correct order so the grid on the data sheet matches the grid on the strung quadrat. Record odd point intercepts for

columns 1,3,5, 7 and 9. Record even point intercepts for columns 2,4,6, 8 and 10. Start at the top left point intercept and continue on downwards, working to the right.

Note: During winter sampling, the number of point intercepts recorded is scaled back to 25 points. Record odd point intercepts for columns 1,5 and 9. Record even point intercepts for columns 3 and 7.

3. If there is another species (sessile invertebrate or macrophyte) growing on top of or covering the understory species at a point intercept, record this as the **overstory species** (e.g. *Endocladia muricata*, *Fucus distichus*, *Semibalanus cariosus*).
4. Repeat with mussel point intercept plots #2, #3, #4 and #5.



Figure 7. Mussel plot within 50 cm by 75 cm quadrat.

Fucus Dynamics: Point Intercepts

1. Place a 75 x 50 cm strung quadrat on top of fucus point intercept plot #1 so that the 3 corner bolts line up with the corners of the quadrat, with the long (75 cm) side of the quadrat at the top. Do not disturb the algae within the plot prior to quantification.
2. Starting at the top left point intercept of the 75 x 50 cm strung quadrat, record the overstory species (eg. *Fucus spp.*) that is present directly underneath the intersection of the strings. In the same location, gently move the overstory species aside and record the understory species (eg. non-coralline crust, *Endocladia muricata*, *Chthamalus spp.*, rock). The understory species may be a macrophyte or a sessile invertebrate. *If the point intersect falls on Fucus spp. with a stipe or holdfast as the understory, record Fucus spp. as the overstory and the understory for this point.* Continue recording overstory and understory for all 100 point intercepts on the strung quadrat. **It is important to record this data in**

the correct order. Start at the top left point intercept and continue on downwards, working to the right.

Note: During winter sampling, the number of point intercepts recorded is scaled back to 50 points. Record odd point intercepts for columns 1, 3, 5, 7 and 9. Record even point intercepts for columns 2, 4, 6, and 8.

3. Repeat with *Fucus distichus* point intercept plots #2, #3, #4 and #5.



Figure 8. *Fucus* plot within 50 cm by 75 cm strung quadrat.

Workflow Guidelines

- The recorder is encouraged to use the program wide species codes, but can use different codes in the field if the recorder defines them on the data sheet.
- For mussel and *Fucus* spp. point intercept plots, the sampler and the recorder must make sure to communicate that the point intercept locations match the locations on the corresponding datasheet for all points on the grid. Check in with each other as you go!
- For each point intercept on the datasheet, the overstory species is recorded in the top left of the square and the understory species is recorded in the bottom right of the square.
- *Data management:* When entering Rocky Intertidal data post field monitoring, we have changed these species codes so they are consistent with other Hakai Nearshore Ecology habitat data for data management purposes. Please see the “Species Codes” tab on the Rocky Intertidal Data Entry Spreadsheet (in Google Drive).

Scoring Guidelines

1. Record the species code for the top-most organism/substrate first (see rules below for weedy species and cases where there are >2 layers).

2. Record the species code for the bottom-most organism/substrate second. If there is only a single layer, record this species code as the bottom organism/substrate. If the same species occurs in the top-most and bottom-most layers (e.g. "Fucus" canopy over "Fucus" holdfast), then record Fucus as the bottom and top layers (e.g. Fucus over Fucus)..
3. Only record rock or sand under a species if you can actually see it. For example *Fucus* on rock is acceptable (as long as you don't hit a holdfast), but *Chthamalus* on rock is just recorded as "NA" on Chthamalus.
4. If there are >2 layers, record only the **uppermost BIOLOGICAL layer (not ephemeral) and lowermost Biological species;** Some examples to illustrate how to record data for points with >2 layers:
 - a. *Fucus* on *Chthamalus* on *Mytilus*, record top layer=*Fucus* and bottom layer=*Mytilus*.
 - b. Dead *Balanus glandula* on *Semibalanus* on *Mytilus*, record top layer=Dead *Balanus glandula*, bottom layer=*Mytilus*.
 - c. *Mastocarpus* over *Endocladia* over *Semibalanus*, record top layer=*Mastocarpus*, bottom layer=*Semibalanus*.
5. Do not score mobile invertebrates (e.g. *Tegula*, *Nucella*). Instead, score the organism/substrate underneath them. Exception: large or immovable motile invertebrates such as *Pisaster* or chitons, where the individual takes up too much space for the sampler to be sure what substrate is under the animal at that point.
6. Epoxy corner markers and bolts should be scored as "rock".
7. When sand is present under a point, if you can identify what is under the sand, then score the underlying species or substrate; otherwise score as "sand".

Motile Invertebrates: Biodiversity

Research Objectives:

Are the infaunal mobile invertebrate communities in *Fucus* spp. beds, mussel beds and barnacle-inhabited substrate changing over time (seasonally and annually)?

Does the primary substrate holder (i.e. *Fucus* spp. beds, mussel beds and barnacle) alter the invertebrate community size and abundance?

Equipment

75 x 50 cm quadrat (unstrung)

20 x 20 cm string quadrat

Calipers

Clipboard

Clickers

Pencil

Data sheets (3 per site)

Knee pads

Parameters

- Counts and sizes (cm) of mobile invertebrates associated with three substrates (barnacle, mussel, Fucus)

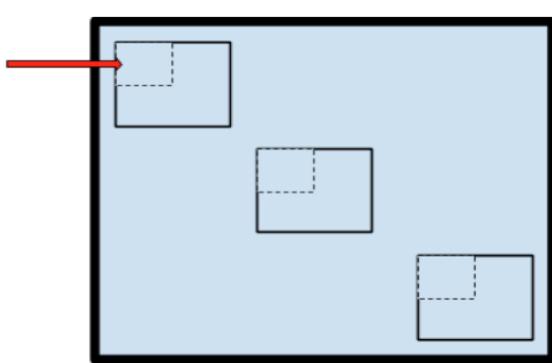
Set Up

Motile invertebrate biodiversity counts and measurements will be conducted on a total of 15 permanent plots (5 barnacle plots, 5 mussel plots, 5 fucus plots) per site.

Motile Invertebrate Biodiversity and Density

- Place a 75 x 50 cm quadrat (unstrung) on top of the three corner bolts that denote the plot number for each plot type. The corner bolts should line up with the corners of the quadrat.
- Count all the motile invertebrates specified in the topmost section of the datasheet within the entire 75 x 50 cm quadrat.
- Count and measure (first 10 encountered only) the species in the second section of the datasheet within the entire 75 x 50 cm quadrat.
- Using the 20 x 20cm string quadrat, subdivide the plot into 3 subsections in the top left, middle and bottom right of the 75 x 50 cm quadrat (see diagram for approximate locations). Within each subsection, sub-sample all small limpets (<5 mm) and medium limpets (5-15 mm) and record the counts in the corresponding boxes.

Note: If small and/or medium limpets are extremely abundant in the 20 x 20 cm quadrat, subsample a 10 x 10cm subsection within the top left quarter of the 20 x 20 cm quadrat and note this on the datasheet. Alternatively, if there are no limpets in the 20 x 20 cm quadrat and limpets are present in the 75 x 50 cm quadrat, count the limpets in entire quadrat.



75 x 50 cm quadrat with three 20 x 20 cm quadrats
(dotted line indicates 10 x 10 subplot)



5. Within each 20 x 20cm subsection, count all *Littorina spp.* within the top left quarter (10 x 10cm area) of the quadrat (visually estimate area for subsample)
Note: If there are no *Littorina spp.* in the 10 x 10cm subsection and are present in the 20 x 20cm quadrat, count the *Littorina spp.* 20 x 20cm quadrat and record this on the datasheet.
6. Repeat steps 1-6 for all 15 point permanent plots (*Fucus spp.*, mussels and barnacles).

Guidelines

- Take photos and sample point intercept plots prior to conducting mobile invertebrates survey to prevent rearrangement of foliose algae (especially important for fucus plots).
- Mobile invertebrates may be gently removed from the plot for measuring if it is too difficult to measure them (be sure to put them back!). Some whelks may be lodged in crevices in the rocks.



Figure 9. Mussel, barnacle and Fucus plots within 50 cm by 75 cm unstrung quadrat.

Mussel Dynamics: Length and Bed Depth

Research Objectives:

Does mussel bed depth change over time?

Does average mussel size change over time?

Equipment

50 x 75 cm quadrat (unstrung)

20 x 20 cm quadrat

Calipers

Clipboard

Pencil

Data sheet (1 per site)

Depth measuring skewer

Knee pads

Lumber chalk

Parameters

- Mussel bed depth to approximate biomass
- Mussel length (subsample)

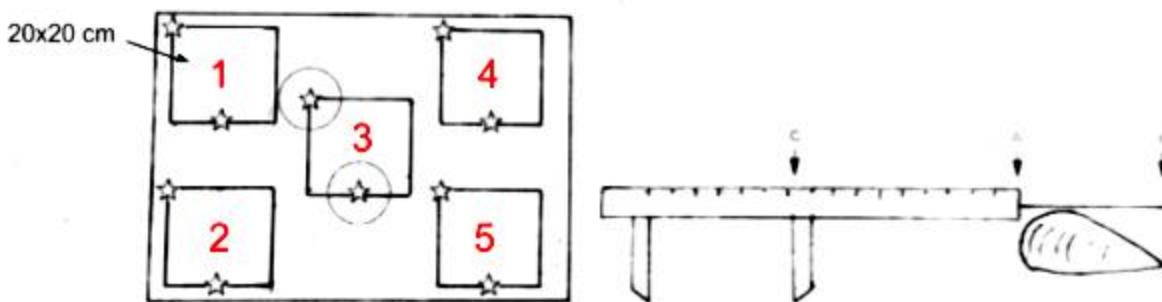
Set up

At each site, mussel length and bed depth measurements will be conducted on 5 plots. The same permanent plots for the mussel point intercept surveys will be sampled. Each plot is marked by 3 corner bolts and zip ties. The plot numbers are labeled by notches (1-5) engraved into the corner bolt of each plot, where the number of notches indicates the plot number.

Mussel Length Sampling

1. Place a 50 x 75 cm quadrat (unstrung) on top of the mussel point intercept plot #1 so that the 3 corner bolts line up with the corners of the quadrat, with the long (75 cm) side of the quadrat at the top (which is denoted by two bolts). Within the 50 x 75 cm quadrat, place the 20 x 20 cm quadrat in each of the 5 locations detailed in the diagram below, moving in order from the top left (1), bottom left (2), middle (3), top right (4) then bottom right (5).
2. In each of the 5 locations of the 20 x 20 cm quadrat, measure the length (to the nearest cm) of mussel(s) visible directly under each of the 2 designated points

(indicated by stars in diagram below). There will be a total of 10 measurements per 50 x 75 cm quadrat. If there is no mussel under a point, measure the length of the mussel nearest to the point that can be found within the 50 x 75 cm photoplot (also measure bed/patch depth at this mussel- see Mussel Bed Depth section). A mussel is only measured once, so if a mussel is also closest to another designated point, the next closest non-measured mussel is chosen. Use lumber chalk to distinguish measured mussels. *For example, if there are only 16 mussels in a 1 layer clump in the 50 x 75 cm plot, 10 of these 16 mussels would be measured no matter where the clump occurs in the plot.*

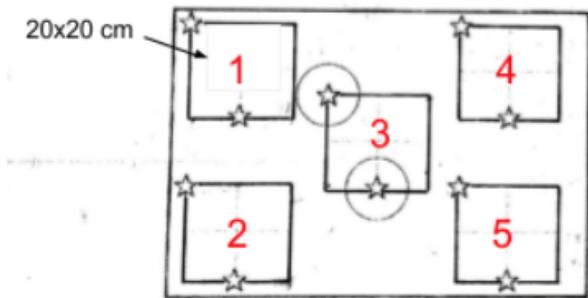


3. To measure a mussel, rest the end of the caliper (**A** in diagram) against one end of the mussel. Slide the caliper open until the tip of the rod (**B**) reaches the end of the mussel. Read mussel length on the scale (**C**) to the nearest centimeter. *For extra thick mussel beds, a long rod/skewer and ruler can be used in place of the caliper.*
4. On the datasheet, tick the box in the “Top” column that accords to the size of the mussel.
5. If the bed/patch has multiple layers, also measure any deeper mussel under the point that can be reasonably measured without undue disturbance to the bed. On the datasheet, tick the box in the “Under” column that accords to the size of the mussel (note that all understory measurements are pooled). There is no need to indicate whether a mussel is attached to another mussel or to the substrate.
6. Repeat steps #1-5 for mussel photoplots #2, #3, #4 and #5.

NOTE: For plots where mussels are rare or absent, there may be <10 or even 0 measurements. For plots containing 10 or more mussels, a total of at least 10 length measurements will be recorded (more if multiple measureable mussels occur below any of the 10 designated).

Mussel Bed Depth Sampling

- For all 50 x 75 cm plots with more than 10 mussels, measure the bed depth (to the nearest cm) of the mussel(s) directly under the upper left point (corner star—see diagram). A total of 5 bed depths will be measured per 50 x 75 cm quadrat (unless there are fewer than 10 mussels in the 50 x 75 cm quadrat).



- The measurement is taken perpendicular to the substrate orientation under the point, upwards from the substrate to the top of the mussel that lies directly under the top point. There is no need to record the layering status. Since the layering rod cannot go through the mussels, place it in a nearby gap and project across to the point location (a small ruler can help with this projection).

Note: For single layer mussel oriented vertically by the longest length, the mussel length and bed depth measurements will be the same. However, since mussels vary in orientation, and multilayering may occur, the 2 measurements will rarely be the same. If the entire plot is an obvious monolayer, then put a check in the monolayer box, if not, leave it blank, but still measure the bed depth.

Guidelines

- Mussel bed thickness is sampled to approximate population biomass (when combined with separately scored mussel cover in photoplot). *There is no need to measure bed depth if less than 10 mussels occur in plot (we can get approximate biomass from length measurements).*
- In cases where no mussel is present under the designated point (would have been previously noted in Mussel Length Sampling), measure bed depth where the next closest mussel in the 50 x 75 cm photoplot is located.

Surfgrass Community Structure

Research Objectives:

Does Phyllospadix coverage change over time?

Is the Phyllospadix infaunal community changing over time?

Equipment

50m transect tape

Tape measure

25cm x 25cm strung quadrat

25cm x 25cm unstrung quadrat

Clipboard

Pencil

Datasheets

Monitoring parameters

- Phyllospadix spp. shoot density
- Reproductive status of surfgrass (flowering shoot density)
- Phyllospadix average canopy height
- Patchiness of Phyllospadix spp. cover
- Surfgrass epiphyte cover
- Mobile invertebrate diversity within Phyllospadix spp. bed

Set up

One permanent surfgrass transect per site is marked by 2 stainless steel bolts at either end of a 10m transect. Lay 10m transect tape (refer to site maps to denote 0m bolt at each site).

Motile Invertebrates

1. Place 25cm x 25cm unstrung quadrat on right side of transect at 1.5m mark.
2. Estimate % cover of Phyllospadix spp. within 25cm x 25cm quadrat
3. Identify and count all mobile invertebrates within 25cm x 25cm quadrat.
4. Record presence/absence (P/A) for colonial invertebrates.
5. Continue steps 1-3 for a total of 5 quadrats along the 10m transect. Prioritize sampling the 5 quadrats on the right side of the 10m transect (1.5m, 3.5m, 5.5m, 7.5m, 9.5m). If surfgrass cover within quadrat is less than 50%, sample next

surfgrass quadrat on **left** and note on datasheet. (Eg. move to left at 2.5m if right at 1.5m contains <50% surfgrass). A total of 5 quadrats must be sampled.

Notes:

If fewer than 5 quadrats on the left **and** the right contain >50% surfgrass cover, randomly select additional quadrat locations on the transect.

To randomly select a distance on the transect, refer to the milliseconds display on a digital watch. Pause the watch to determine the meter mark. Pause the watch a second time to determine the decimeter mark. Pause the watch a third time to determine the centimeter mark. Pause the watch a fourth time to determine whether the quadrat will be placed on the left or right side of the transect. If an even number is selected, choose the right side. If an odd number is selected, choose the left side.

Length and Density

1. Place 25cm x 25cm strung quadrat on left side of transect at 0.5m mark.
2. Determine *Phyllospadix* species (*P. torreyi*, *P. scouleri*), count number of shoots and record number of flowering shoots in **12.5cm x 12.5cm subplot** (lower corner closest to transect; see diagram). *If shoot density in 12.5cm x 12.5cm subplot is very dense, subsampling further is an option. Make sure this is recorded on the datasheet!
3. Using retractable tape measure, record average canopy height (height at which shoots taper off) of all shoots measured.
4. If present, record epiphyte species and epiphyte species percent cover.
5. Alternating sides at one meter intervals, repeat steps 1-4.

Distances: 0.5m (L), 1.5m(R), 2.5m(L), 3.5m(R), 4.5m(L), 5.5m(R), 6.5m(L), 7.5m(R), 8.5m(L), 9.5m(R) (total 10 quadrats)

Notes:

At 1.5m(R), 3.5m(R), 5.5m(R), 7.5m(R) and 9.5m(R), complete invertebrate diversity protocol **prior to** surfgrass bed dynamics protocol. Mobile invertebrates flee from quadrat area in response to disturbance caused by counting and measuring shoots.

Record 0 if no surfgrass observed in a quadrat.

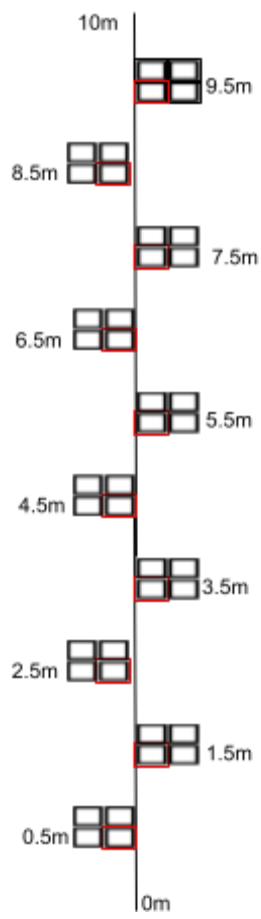


Diagram 1. Surfgrass transect survey design. Along 10m transect, 25cm x 25cm quadrat is placed every meter on alternating sides of the transect line. Surfgrass bed dynamics are monitored within 12.5cm x 12.5cm subplot (red).

Seastar and Macroinvertebrate Dynamics: Swaths

Research Objectives:

Is the abundance and size of *Pisaster ochraceous* changing over time?

Is the incidence of seastar wasting disease changing over time?

Is the abundance and size of *Katharina tunicata* changing over time?

Are the abundance and sizes of other large invertebrates changing over time?

Equipment

50 m transect (x7)

15 cm rulers (1 per technician)

Clipboard

Pencil

Datasheet

Lumber chalk

Camera

Clickers (x3)

Parameters

- *Pisaster ochraceous* counts, size distribution (from radius measurements) and disease status (wasting disease category according to the UCSC classification system)
- *Katharina tunicata* counts and size distribution
- Counts and sizes of other large invertebrates (other sea star species, urchins, *Cryptochiton stelleri*)

Set Up

At each site, 6 bolts have been permanently drilled into the rock to delineate three 30m horizontal transect lines in the high, mid and low intertidal zones. At Fifth Beach and North Beach, these are the bolts originally drilled for the Martone Lab annual monitoring. Two bolts have been drilled at an approximate distance of 30m in each zone. Refer to maps and coordinates to locate bolts.

1. Lay 3 transects horizontally through the two bolts in each zone.
2. Lay 4 transects vertically from the high zone transect line to the water line at 0m, 10m, 20m and 30m. The area between the low zone transect line and the water line will be searched even though the low zone bolts have been drilled above the water line (at approximately 1.0m). In order to

maximize sampling area, search the lower intertidal zone at the time of low tide.

By creating a grid, the area searched can be divided into subplots so that technicians can be assigned a set search area. The area searched will change with respect to the low tide levels during the day of the survey, however the counts of invertebrates/m² can be extrapolated using the bolt locations and the low tide height at the time of the survey.

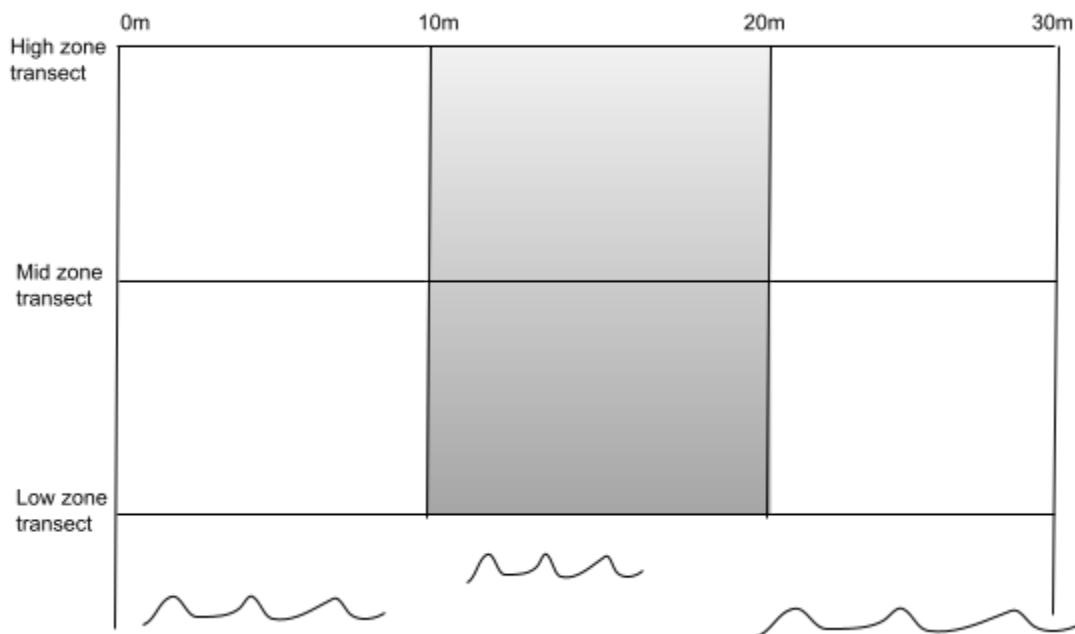


Figure 9. Diagram of *P. ochraceous* and *K. tunicata* swath set up. Grey area indicates *K. tunicata* search area. Searching for all target species will continue above high zone transect line and below low zone transect line to water line within 30m swath.

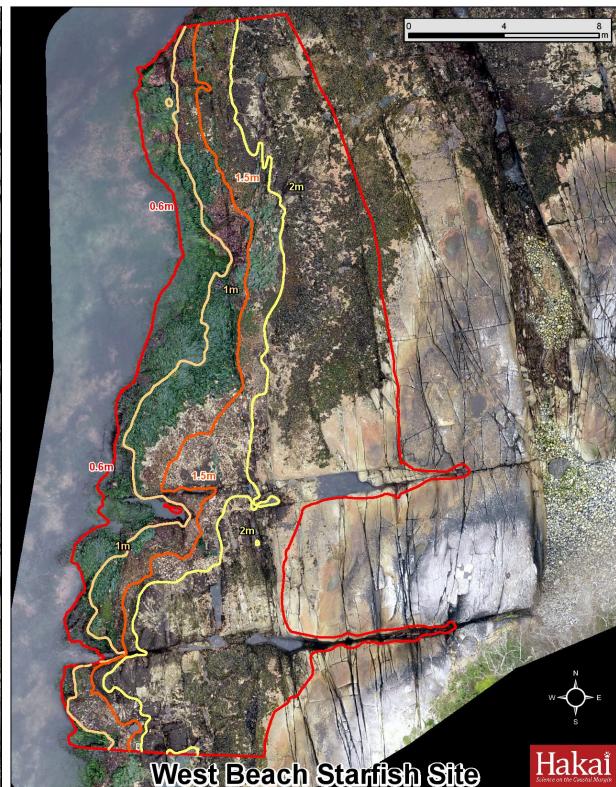
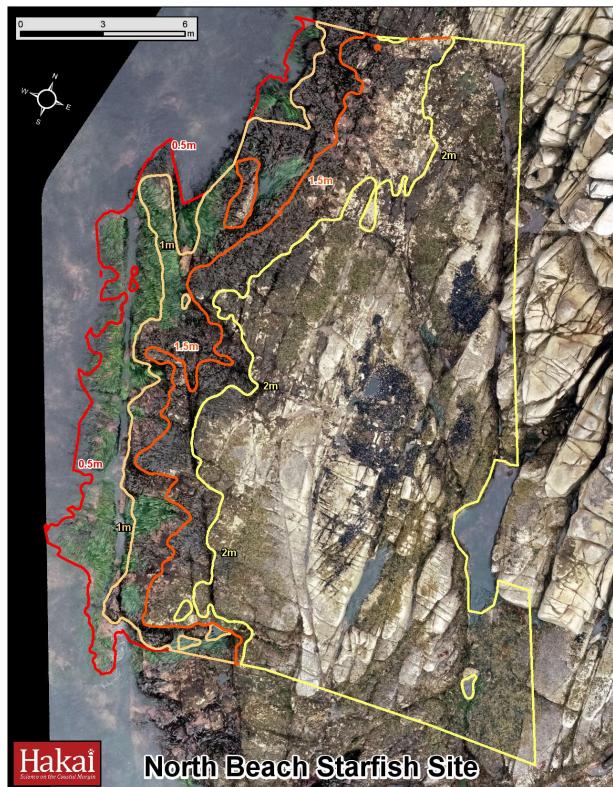
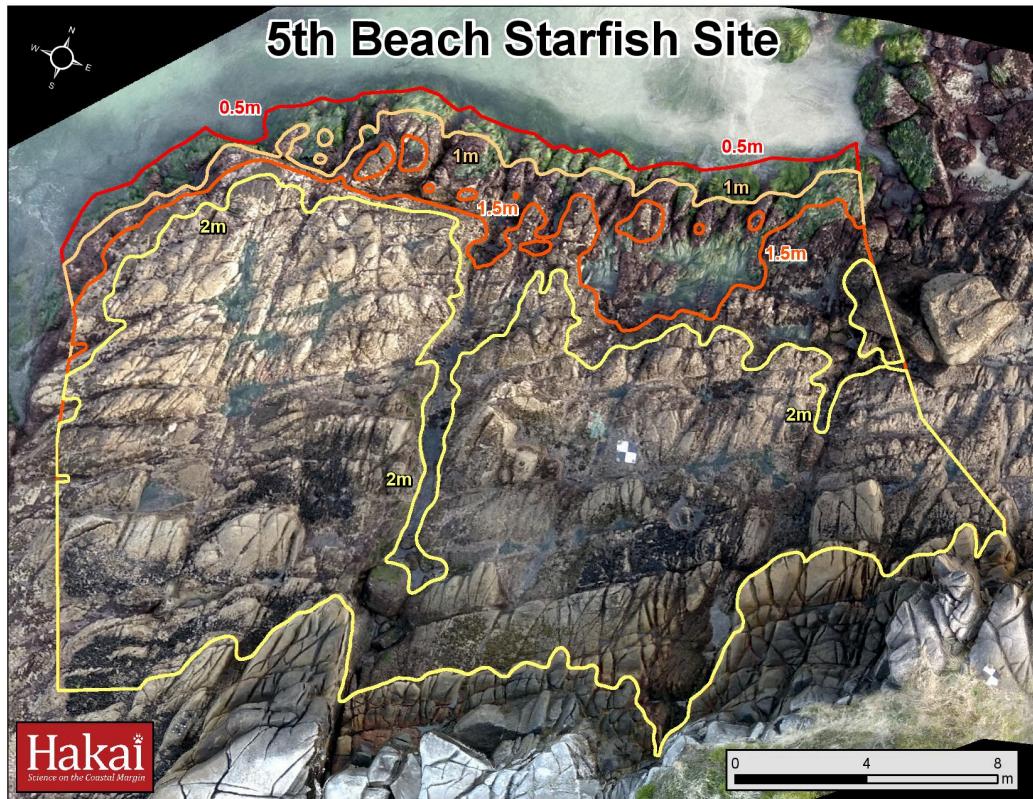


Figure 10. Sea star swath elevation maps of Fifth Beach, North Beach and West Beach.

1. Search within the plot parameter for ochre stars. Assign technicians a set search area. In order to maximize sampling area, search the lower intertidal zone at the time of low tide. Measure the first **100** encountered individuals (includes juveniles and adults for a total of 100 measurements). Measure size to the nearest 10mm from the center of the disc to the tip of the longest ray. Sizes may be estimated if sea stars are wedged in crevices with rays curved.
2. Record the corresponding disease category (0-4) for each individual (refer to UCSC Wasting Disease Categories). Technicians are encouraged to mark the substrate **next to** (not on- to avoid potential for spreading disease) the sea star with lumber chalk to ensure individuals are not measured twice. Sea stars should never be “straightened” or removed from the rock.
3. After 100 individuals have been measured, count the remainder of the ochre stars within the plot parameter. Technicians are encouraged to use clickers. Record adults and juveniles separately. Continue to record corresponding disease categories for all individuals.

Adults = center of the disc to the tip of the longest ray is **greater than 30mm**

Juveniles = center of the disc to the tip of the longest ray is **less than 30mm**
4. Measure all (to a maximum of 30 per species) other large invertebrates. This includes gumboot chitons (*Cryptochiton stelleri*), Northern Abalone (*Haliotis kamtschatkana*), urchins and other sea stars (eg. *Henricia* spp., *Dermasterias* spp., *Leptasterias* spp. and any other species observed). Measure other sea stars from the center of the disc to the tip of the longest ray. Measure chitons and abalone lengthwise from tip to tip. Measure test diameter of urchins.
5. Report all measurements to the recorder.

Leather Chiton Dynamics: Swaths

1. Within the middle 10m segment of the plot parameter (see Figure 9), measure 50 leather chitons (*Katharina tunicata*).
2. Count all remaining leather chitons within the 10m subplot and report counts to the recorder. To avoid counting individuals twice, individuals can be marked directly with lumber chalk. Technicians are encouraged to use clickers.

Mussel Growth Experiment

Adapted from: PISCO Intertidal, Oregon State University

Research Objectives:

Are *Mytilus californianus* growth rates changing spatially or seasonally?

Equipment (for set up)

Wood file or dremel

Vexar mesh

Zip ties

Hammer drill

Bolts (8 per replicate)

Safety glasses

Set Up

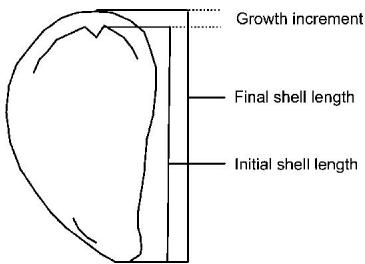
Outplant mussels at rocky intertidal sites around Calvert Island to monitor their growth seasonally and yearly. Mussels are initially strapped to the rock with Vexar cages to encourage byssal attachment. Vexar cages are slowly removed following mussel attachment. Select experiment locations on sections of bare rock placed within the middle of the mussel bed vertical extent at the site (half way between the upper and lower limit of the mussel bed). For each replicate, install 8 bolts in order to attach a rectangular piece of Vexar mesh (approximately 30cm x 30cm). To monitor yearly and seasonal mussel growth outplants will be reset annually in October.

Winter replicates (n=3): Begin protocol in October. Mussels will grow for 6 months over winter.

Summer replicates (n=3): Begin protocol in April. Mussels will grow for 6 months over summer.

Mussel Collections

1. Collect 50 mussels between 4-5 cm in total length. Attempt to limit collections to the middle range of the mussel bed.
2. Using a wood file or dremel, notch each mussel along the top of the shell (where the shell meets, opposite from the umbo). Example here is showing the notch after several weeks/months of growth, initial notch will be in the same location, but at the edge of the shell..



3. On bare rock approximately mid-mussel band, drill bolt holes for 3 plots to secure the Vexar mesh that will cover the mussels (see below). Make sure that the mesh is secure to prevent the mussels from falling out, but that it is loose enough to allow them to grow.
4. After approximately 4 weeks, loosen the vexar mesh by a few centimetres to encourage the mussels to attach more byssal threads and to allow them to grow more.
5. After approximately 8 weeks, check if the mussels are securely attached. If they are, remove the Vexar mesh. If they are not, return in one week.
6. After approximately 6 months, collect 25 mussels (or half of the surviving mussels) from each plot and store them in a freezer @ -20°C. Leave the remaining 25 mussels (or other half of the surviving mussels) to grow for another 6 months.
7. Repeat steps 1-3 for the next treatment.
8. Repeat steps 3-4 for the three new plots.
9. At 12 months from initial outplant collect remaining mussels of the first treatment as well as 25 mussels from each plot of the second treatment. Restart the yearly outplant by restarting at step 1.



Lab processing

Equipment

Gloves
Lab coat
Calipers
Scalpel
Forceps
Tray
Aluminum tins (10 per replicate)
Pencil
Data sheet

Monitoring parameters

- Mussel measurements: total length, initial length, perisoticum lip length, girth, height
- Mussel gonad development and sex
- Mussel total wet tissue weight, dry tissue weight and shell weight

Lab Processing

1. Remove a bag containing a replicate of mussels (n=25 or less depending on survival rates) from the freezer and allow them to thaw.
2. Select a mussel and record the sample ID, site, deployment date (when mussel was notched), collection date, lab processing date, duration of experiment and season.
3. Scrape off any barnacles on the mussel shell with a scalpel and carefully pull off the byssal threads.
4. Using calipers, measure the total length (bottom to top; longest length), initial length (bottom to top of notch), Perisoticum lip (bottom to the black), girth (distance from side to side of shell, in line with the umbo), height (distance from ventral side to tallest part on dorsal side) of the mussel shell to the **nearest tenth of a mm**. Refer to mussel measurements images below.

Note: *The growth of the mussel will be calculated in excel by subtracting the initial length of the mussel shell by the total length of the shell.*

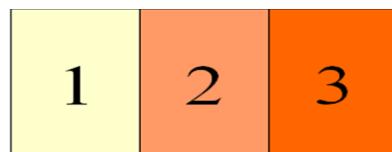
5. Cut open the mussel by slicing through the adductor muscle. Place it open side down on the tray, and allow any liquid to drain out.

6. Record sex category. **See Gonad Color Scoring below** and record category.
 - 1=creamy white
 - 2=white to peach-ish to orange-ish
 - 3=dark orange
7. Record gonad development category.
 - 0 = no gonad visible
 - 1 = small gonad
 - 2 = larger gonad confined to visceral mass
 - 3 = large gonad extending into mantle cavity
8. Record the total wet weight of the mussel in grams – to the **nearest hundredth of a gram**.
9. Record the weights of aluminum tins.
10. Place the wet tissue in a pre-weighed aluminum tin and record the wet tissue weight (tin weight + wet tissue weight).
11. Record the shell weight to the **nearest hundredth of a gram**.
12. Repeat step 10-11 for all mussels.
13. Record the weight of the shell for all mussels in each replicate to the **nearest hundredth of a gram**.
14. Place aluminum tins with wet tissue in drying oven at 60°C.
15. After a minimum of three days in the drying oven, record the dry weight of the soft tissues. Be sure that all tissue is crispy.

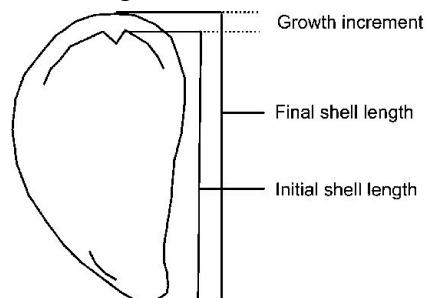
Mussel Measurements



Gonad Color Scoring



Mussel growth with notch



Surface Water Grab Samples

Field Protocols

YSI Measurements

Equipment

Calibrated YSI unit

Field Cover Sheet

Monitoring parameters

- Temperature (°C), specific conductivity, salinity and pH measurements
- 1. Calibrate the YSI in the lab before going to the study site.
- 2. At each site, submerge the YSI probe in the seawater near the surface. Allow readings to stabilize.
- 3. Record: time, temperature (°C), specific conductivity, salinity, pH

Water collection

Equipment

Weighted carboy and line

500mL brown Nalgene bottles (x3)

Nitrile gloves

Monitoring parameters

- do¹³C, nutrients, chlorophyll
- 1. Wearing nitrile gloves, deploy the weighted carboy from the shore and allow it to fill with seawater.
- 2. Using the attached line, bring the carboy back to the shore.
- 3. Using the water spigot, rinse one brown 500mL Nalgene **3** times. Fill it completely with the remaining seawater in the carboy.
- 4. Continue steps 1-3 for a total of 3 replicates.

Note: Samples should be processed upon returning to the lab, or within 2 hours from time sampled (see lab protocol).

Lab Protocols

Complete 'Marine Lab' iPad form with hyper nearshore water sample filtering lab protocol.

Processing surface water grab samples

Equipment

Gloves

Peristaltic pump

Flat forceps

60mL syringe

250ml beaker

Squeeze bottle of ethanol

Squeeze bottle of DI water

Filter tips (3 per site)

Non-combusted 47mm GFF filters

3um filters

20um filters

Mesh separators

Petri dish (x2)

Black ziplock bag

Stickers with UIDs (per site: DO13C x3, nutrients x1, Chl a x2)

Blank sticker labels

Falcon tubes (14mL) (3x per site)

Amber borosilicate glass vials (40mL) (3x per site)

Kimwipes

Permanent marker

Safety Glasses (for acidifying DO13C)

Lab Coat (for acidifying DO13C)

50mL beaker (x2) (for acidifying DO13C)

Pipette and tip (for acidifying DO13C)

Waste acid container (for acidifying DO13C)

6M HCL (for acidifying DO13C)

Pre-labelling stickers

1. Sheets of stickers containing UIDs have been pre-printed for each sample type: nutrients, DO13C and Chl a.
2. For each site, pre-label 3 amber borosilicate glass vials (40mL) for DO13C (bottles 1, 2 and 3), 1 falcon tube for nutrients (bottle 1 only) and 6 falcon tubes for Chl a (bottles 1 and 2; 3 filter size fractions). Pre-printed labels contain sample type, UID, and marine (M) or freshwater (F) designation. Chlorophyll a labels also contain filter size fraction. With a permanent marker, circle GFF on 2 falcon tubes, 3um on 2 falcon tubes and 20um on 2 falcon tubes.
3. On blank sticker labels, write site code, date and bottle number and place on all bottles and falcon tubes. See below for samples size and replicates per site.
4. Each amber borosilicate glass vials (40mL) and falcon tube should be labelled with sample type, UID, filter size fraction, site code, date and bottle number.

Sample sizes and replicates per site:

DO13C: n=3

Nutrients: n=1

Chl a: n=2

YSI: n=1

Bottle 1:

DO13C: n=1

Chl a: n=1 (GFF, 3um, 20um)

Nutrients: n=1

Bottle 2:

DO13C: n=1

Chl a: n=1 (GFF, 3um, 20um)

Bottle 3:

DO13C: n=1

Filtering nutrients

1. Wearing gloves, rinse a 60mL syringe 3 times with DI water.
2. Attach a filter tip and rinse the 60mL syringe 3 times with seawater from bottle 1.
3. Fill the 60mL syringe with seawater from bottle 1.
4. Filter ~10mL of the seawater into the sink to clean out the filter tip.
5. Rinse a pre-labelled nutrient falcon tube 3 times with the filtered seawater in the syringe.
6. Fill the falcon tube to 13 mL with the filtered seawater from the syringe.
7. Store the falcon tube in the chest freezer in the Griswold Lab.

Filtering DO13C

1. Wearing gloves, rinse a 60mL syringe 3 times with DI water.
2. Attach a filter tip and rinse the 60mL syringe 3 times with seawater from bottle 1.
3. Fill the 60mL syringe with seawater from bottle 1.
4. Filter ~10mL of the seawater into the sink to clean out the filter tip.
5. Rinse a pre-labelled DO13C amber borosilicate glass vial 3 times with the filtered seawater in the syringe.

6. Fill the amber borosilicate glass vial to the bottleneck with the filtered seawater from the syringe.
7. Repeat steps 1-6 for bottles 2 and 3.
8. Acidify samples immediately.

Acidifying DO13C

1. Wearing gloves, safety glasses and a lab coat, turn on the fume hood in the Britco Lab.
2. Fill an acid waste container with fresh water (there is likely already an acid waste container in the fume hood. This is diluted and emptied regularly by techs from other programs that use the lab more frequently).
3. Pour ~20 mL of DI water into a 50mL beaker and place it in the fume hood.
4. In the fume hood, pour a very small amount (~1mL) of 6M HCL into a 50mL beaker. Any unused acid cannot be poured back into the acid bottle, so be conservative.
5. At the fume hood, remove sample lids. Be sure that lids are organized so that they go back on the same bottle.
6. Select the appropriate pipette and pipette tip in order to add 200 μ L of 6M HCL.
7. Carefully pipette 200 μ L of 6M HCL into each sample. **As long as the pipette tip does not touch sample water, the same pipette tip can be used for all 3 DO13C samples.**
8. Replace the sample lids and invert the samples to mix the acid.
9. Rinse the acid tip by pipetting DI water and eject it into the waste acid container 3 times.
10. Eject the used tip into the “*Rinsed acid tips*” container in the fume hood.
11. Pour unused acid in the 50 mL beaker into the waste acid bucket. Rinse the beaker with DI water.
12. Mark the DO13C labels with an “A” (for acidified) with a permanent marker and store them in the Griswold fridge.

Filtering chlorophyll

1. Wearing gloves, turn on peristaltic pump and rinse filter cups and filter holder with DI water 3 times. Turn off pump.
2. Using clean forceps, place a GFF filter on bottom of the peristaltic pump, followed by a mesh separator in the center of the filter. Place a 3um filter on top of the mesh spacer (shiny side up). Place a mesh spacer in the center of the 3um filter. Place a 20um filter on top of the mesh spacer (shiny side up).
3. Carefully screw the filter cup onto the pump, preventing the formation of wrinkles or air bubbles on the filters.
4. Measure 250mL of seawater from **Nalgene bottle 1** into a 250mL beaker.
5. Turn on the pump (pressure should not exceed -5 Hg). Pour the 250mL of seawater into the filter cup. Open the filter funnel valve. **Avoid letting the filter run dry- this will cause phytoplankton cells to rupture.**

6. When filtering is complete, close the filter funnel valve. Carefully unscrew and remove the filter funnel cup.
7. Using flat forceps, roll up the 20um filter (on top). The filter should be rolled up like a burrito with the contents of the sample on the inside. Avoid touching the center of the filters with the forceps, only the edges.
8. Place the rolled up filter in a pre-labelled falcon tube. Make sure it is below the 10mL marking on the falcon tube. Place falcon tube into a black ziplock bag. Label bag with site code, date and bottle number.
9. Remove the mesh separator. Place it in petri dish and spray with ethanol until it is covered. Move the mesh separator to another Petri dish and rinse with DI water.
10. Using flat forceps, roll up the 3um filter (in middle). Repeat steps 7-9 with the 3um filter and the mesh separator.
11. Using flat forceps, roll up the GFF filter (on bottom). Repeat steps 7-8 with the GFF filter.
12. Screw on filter cup and repeat steps 1-11 with **Nalgene bottle 2**.
13. The two black ziplock bags containing 3 falcons tubes from bottle 1 and 3 falcon tubes from bottle 2 should be stored in the -80°C freezer in the Britco Lab.

Appendix

Appendix A. Data Sheets

Appendix A1. Field Cover Sheet

Rocky Intertidal Coversheet

Site:

Date:

Field team:

Tide Sampled:

Time @ low	Low (m)	Wind (L/M/H)	Swell (L/M/H)	Hours Worked

YSI data:

Time	Temp	Spc	Salinity	pH

Completed:

	Barnacle	Mussel	Fucus
Point intercepts			
Mobile Inverts			
Bed depths			
Plot Photo			

Seastar swath			
Katharina swath			
Phyllospadix quads			
Phyllospadix inverts			

Comments:

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Appendix A2. Point Intercept Plot Survey Data Sheet

Site: _____ Plot 1

Recorder: _____ Plot 2

Invertebrates:

BAGL = *Balanus glandula*
CHDA= *Chthamalus dalli*
SECA = *Semibalanus car*
MYCA = *Mytilus californi*
PPOO = Pollicipes polymy

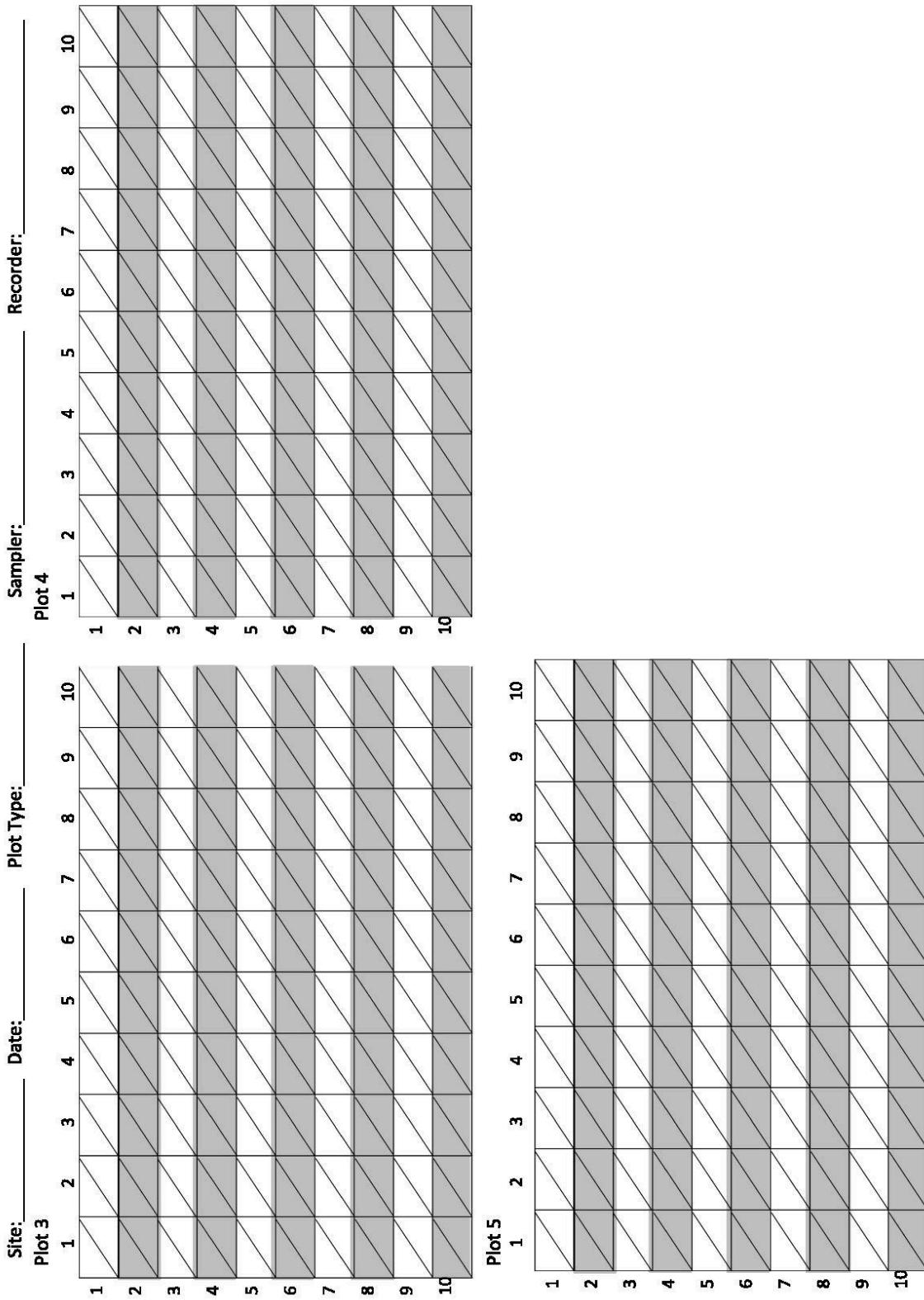
ANEI = *Anthopleura eleg*

Algae:

NNC	= Non-coraline crust
CCOR	= Coralline crust
ACOR	= Articulated corallines
CLAD	= Cladophora spp./A. f.
MMAST	= Mastocarpus spp.
FUDI	= Fucus distichus
HILD	= Hildenbrandia spp.
PETR	= Petrocellis.
ENMU	= Endocladia muricata
FRFD	= Filamentous red al.
ULVA	= Ulva spp.
HAGL	= Halosaccion glandiforme
MAZZ	= Mazzarella spp.

Dead/Other/Non-Biological:

ROCK = Rock
DBAGL = Dead *Balanus glandula*
DCHDA = Dead *Chthamalus dalli*
DSECA = Dead *Semibalanus cariosus*
DMyCA = Dead *Mytilus californianus*



Appendix A3. Motile Invertebrates: Biodiversity Data Sheet

Appendix A4. Mussel Dynamics: Length and Bed Depth Data Sheet

Mussel Length and Bed Depth Sampling

Site: _____

Sampler: _____

Date: _____

Recorder: _____

<10 mussels in plot? Yes _____ No _____

If <10 mussels, measure lengths. Do not measure "bcd" depths.

Top = topmost mussel layer. **Under** = understory mussel layer(s)

Record a tick mark in appropriate box for each mussel measured.

measure to nearest cm, but record in mm (i.e. 5cm=50mm)

Mussel

Bed depth- measure to nearest cm, but record in mm (i.e. 5cm=50mm)

Check box if entire plot is an "obvious" non-*layer*

Notes. _____

OCT 29 12

Appendix A5. Surfgrass Community Structure: Motile Invertebrate Data Sheet

Appendix A6. Surfgrass Community Structure: Length and Density Data Sheet

Rocky Intertidal: Surfgrass Survey
 Site _____ Date _____ Time _____ Recorder _____
 Count shoots & record flowering shoots in 12.5cmx12.5cm subplot (lower corner closest to transect). Record average canopy height, epiphyte spp. & % epiphyte cover.
 Record 0 if no surfgrass observed. If <5 mobile inverts quadrats sampled, randomly select additional quadrat location on transect.

Dist. (m)	L / R	Species	Quad Density	Flowering Shoot Density	Canopy Height (cm)	Epiphyte Species	Epiphyte Cover (% Cover)	Notes
0.5	L	<i>P. torreyi</i> <i>P. scouleri</i>						
1.5	R	<i>P. torreyi</i> <i>P. scouleri</i>						
2.5	L	<i>P. torreyi</i> <i>P. scouleri</i>						
3.5	R	<i>P. torreyi</i> <i>P. scouleri</i>						
4.5	L	<i>P. torreyi</i> <i>P. scouleri</i>						
5.5	R	<i>P. torreyi</i> <i>P. scouleri</i>						
6.5	L	<i>P. torreyi</i> <i>P. scouleri</i>						
7.5	R	<i>P. torreyi</i> <i>P. scouleri</i>						
8.5	L	<i>P. torreyi</i> <i>P. scouleri</i>						
9.5	R	<i>P. torreyi</i> <i>P. scouleri</i>						

Appendix A7. Seastar and Macroinvertebrate Swath Data Sheet

Sea Star Swath Survey

Site: _____ Date: _____ Time: _____ Tide: _____ Samplers: _____

Pisaster ochraceus Disease Categories: 0=healthy, 1=lesion(s) on 1 arm or body, 2=lesions on 2 arms or 1 arm and body and/or deteriorating arm(s), 3=lesions on most of body and/or 1-2 missing arms, 4=severe tissue deterioration/death and/or ≥3 missing arms.
Life Stage: radius (from tip of longest arm to central axis) < 30 mm = juvenile, radius ≥ 30 mm = adult.

Size	Disease Category				
	0	1	2	3	4
5					
10					
20					
30					
40					
50					
60					
70					
80					
90					
100					
110					

Sea Star Swath Survey

Site: _____ Date: _____ Time: _____ Tide: _____ Samplers: _____

Pisaster ochraceus Disease Categories: 0=healthy, 1=lesion(s) on 1 arm or body, 2=lesions on 2 arms or 1 arm and body and/or deteriorating arm(s), 3=lesions on most of body and/or 1-2 missing arms, 4=severe tissue deterioration/death and/or ≥3 missing arms.
Life Stage: radius (from tip of longest arm to central axis) < 30 mm = juvenile, radius ≥ 30 mm = adult.

Life Stage	Disease Category				
	0	1	2	3	4
Count					
Adults	0	1	2	3	4
Count					

Other echinoderms and *Cryptochiton stelleri*: Record total # and size.

Species	Size	Count

Appendix A8. Leather Chiton Swath Data Sheet

Katharina tunicata Swath Survey

Site: _____ Date: _____ Time: _____ Tide: _____ Samplers: _____

Measure length of first 50 individuals to nearest cm and record total count in middle 10m of sea star swath transect.

Size (mm)	
10	
20	
30	
40	
50	
60	
70	
80	
90	
100	

Count

Appendix B. Invertebrate Species List

Arthropoda

Amphipoda	<i>Pagurus hirsutiusculus</i>
<i>Balanus glandula</i>	<i>Pagurus</i> spp.
Caprellidae	<i>Petrolisthes cinctipes</i>
<i>Chthamalus dalli</i>	<i>Petrolisthes</i> spp.
<i>Hemigrapsus nudus</i>	<i>Pollicipes polymerus</i>
Idotea spp.	Porcellanidae
<i>Oedignathus inermis</i>	<i>Pugettia gracilis</i>
<i>Pachycheles rufus</i>	<i>Pugettia producta</i>
<i>Pachygrapsus crassipes</i>	<i>Pugettia richii</i>
<i>Pagurus beringanus</i>	<i>Pugettia</i> spp.
<i>Pagurus granosimanus</i>	<i>Semibalanus cariosus</i>
<i>Pagurus hemphili</i>	

Chordata

<i>Aplidium solidum</i>	<i>Styela</i> spp.
<i>Tunicata</i>	

Cnidaria

<i>Abietinaria greenei</i>	<i>Anthopleura xanthogrammica</i>
Actiniaria	Holothuroidea
<i>Anthopleura elegantissima</i>	Hydrozoa

Echinodermata

Cucumaria spp.	<i>Mesocentrotus franciscanus</i>
<i>Dermasterias imbricata</i>	<i>Pisaster ochraceus</i>
Echinoidea	<i>Pseudocnus</i> spp.
<i>Evasterias troschelii</i>	<i>Strongylocentrotus droebachiensis</i>
<i>Henricia pumila</i>	<i>Strongylocentrotus pallidus</i>
Henricia spp.	<i>Strongylocentrotus purpuratus</i>
<i>Leptasterias hexactis</i>	<i>Strongylocentrotus</i> spp.
Leptasterias spp.	

Mollusca

<i>Acmaea mitra</i>	<i>Diodora aspera</i>
<i>Alia carinata</i>	<i>Hermissenda crassicornis</i>
Alia spp.	<i>Homalopoma</i> spp.
Amphissa spp.	<i>Katharina tunicata</i>
<i>Amphissa versicolor</i>	<i>Lacuna</i> spp.
Anomiidae	<i>Lepidochitona dentiens</i>
<i>Bittium eschrichtii</i>	<i>Lepidochitona</i> spp.
<i>Calliostoma canaliculatum</i>	<i>Lepidozona</i> spp.
Calliostoma spp.	<i>Leukoma staminea</i>
<i>Ceratostoma nuttali</i>	<i>Littorina plena</i>
<i>Crasssodoma gigantea</i>	<i>Littorina scutulata</i>
Crepidula spp.	<i>Littorina sitkana</i>
<i>Cryptochiton stellerii</i>	<i>Littorina</i> spp.

<i>Lottia digitalis</i>	<i>Nucella lamellosa</i>
<i>Lottia paradigitalis</i>	<i>Nucella ostrina</i>
<i>Lottia pelta</i>	<i>Nucella</i> spp.
<i>Lottia scutum</i>	Nudibranchia
<i>Lottia</i> spp.	<i>Onchidella borealis</i>
<i>Mopalia</i> spp.	Polyplacophora
<i>Mytilus californianus</i>	<i>Searlesia dira</i>
<i>Mytilus</i> spp.	<i>Tegula funebralis</i>
<i>Mytilus trossulus</i>	<i>Tegula pulligo</i>
<i>Nucella canalicuta</i>	<i>Tonicella</i> spp.

Annelida

Nemertea
Platyhelminthes
Polychaeta
Spirorbis spp.

Bryozoa

Porifera

Haliclona permollis

Appendix C. Macroalgae Species List

Chlorophyta

Codium fragile

Ulva spp.

Rhodophyta

<i>Ahnfeltia fastigiata</i>	<i>Microcladia borealis</i>
<i>Bangiaceae</i>	<i>Neorhodomela larix</i>
<i>Callithamnion pikeanum</i>	<i>Odonthallia floccosa</i>
<i>Cryptosiphonia woodii</i>	<i>Odonthallia</i> spp.
<i>Dilsea</i> spp.	<i>Plocamium</i> spp.
<i>Gloiopeltis furcata</i>	<i>Polysiphonia</i> spp.
<i>Halosaccion glandiforme</i>	<i>Prionitis</i> spp.
<i>Hildenbrandia</i> spp.	<i>Pterosiphonia</i> spp.
<i>Mastocarpus</i> spp.	<i>Ptilota</i> spp.
<i>Mazzaella</i> spp.	<i>Smithora naiadum</i>
<i>Melobesia mediocris</i>	<i>Wildemania norrisii</i>

Appendix D. UCSC Wasting Disease Categories

Category 1

Lesion(s) on 1 arm or body tissue degradation in some of these photos may be the result of multiple lesions merging, but it is restricted to a single arm, or single location on the oral disk.

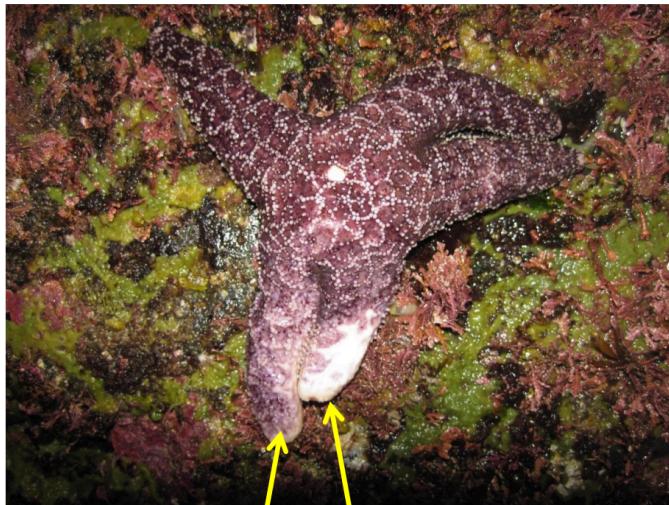


Category 2

Lesions on 2 arms or 1 arm and body, deteriorating arm(s).



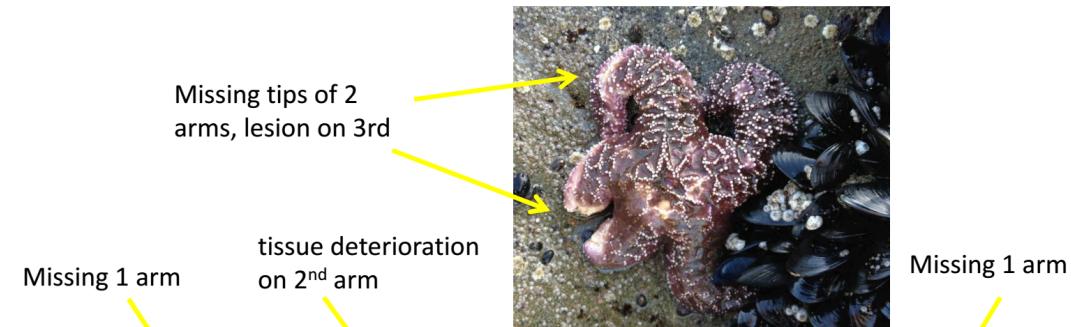
Arm starting to separate



Tissue deteriorating on 2 arms

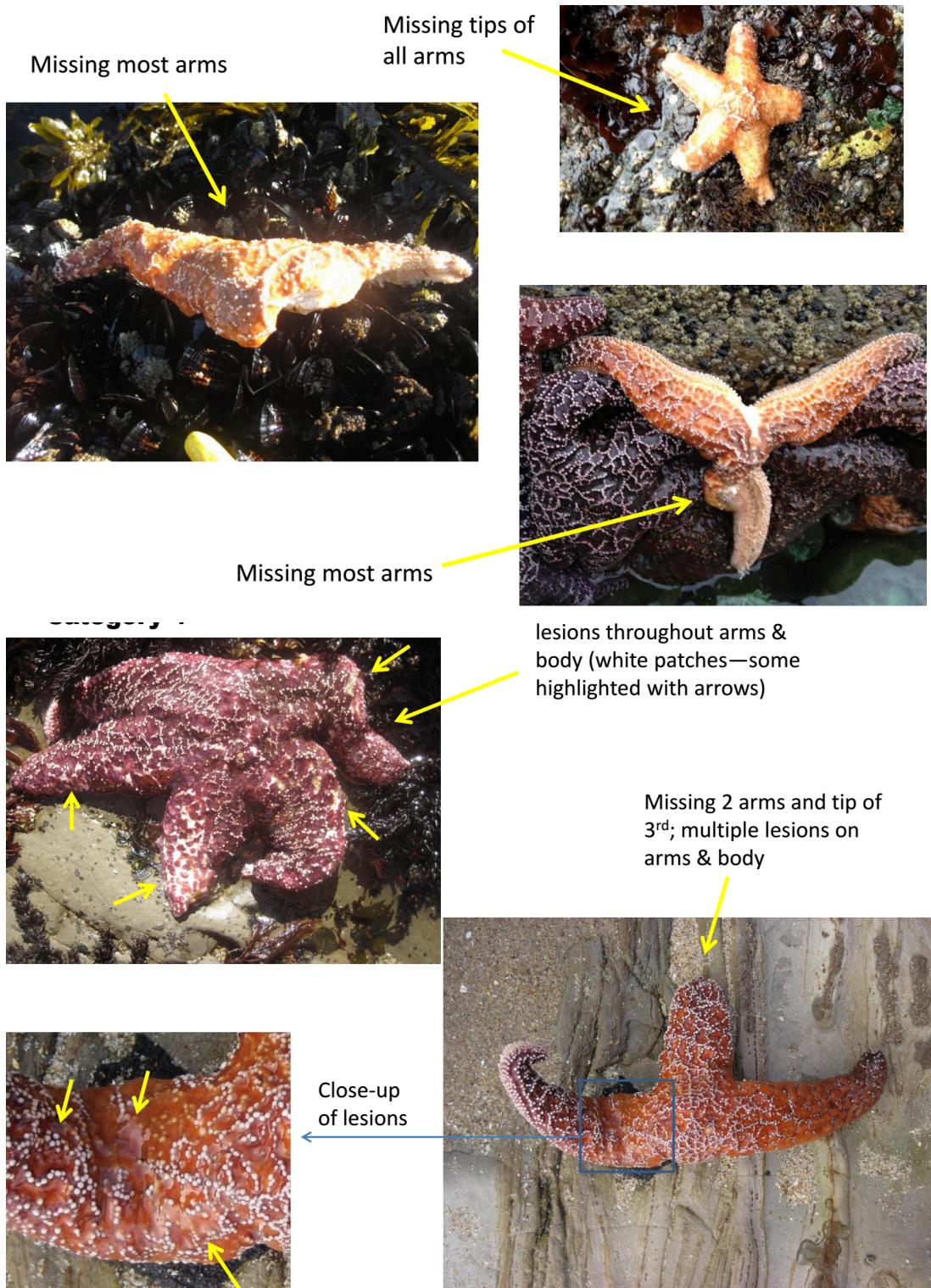
Category 3

Lesions on most of body, 1-2 arms missing.



Category 4

Severe tissue deterioration/death >/= 3 missing arms



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