# Arctic Shorebird Demographics Network
# Breeding Camp Protocol

## Version 4 – April 2013


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Cover photo: Semipalmated Sandpiper, taken on the Canning River, Alaska. Brad Winn.

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# INTRODUCTION

Recent shorebird trend analyses indicate that many North America shorebirds are declining, but we do not know why (Morrison et al. 2006, Bart et al. 2007). The goal of the Arctic Shorebird Demographics Network (hereafter termed "Network") is to collaboratively conduct demographic studies on several shorebird focal species that will help determine factors limiting their population size. The Network will measure demographic rates such as adult apparent survival, annual productivity, apparent nest survival, population age-structure, etc., on the Arctic breeding grounds. Additionally, site-specific ecological and environmental variables (e.g. food resources, prey and predator abundance, weather, etc.) that influence demographic rates and are influenced by climate change and other anthropogenic forces will be measured and incorporated into the analyses. Finally, the Network substantially increases our ability to address a wide variety of other science and conservation goals that can only be examined at a regional or global level (e.g. migratory connectivity studies that require marking individuals over a large area, the collection of tissue samples, analyses of contaminants, etc.).

## *Rationale for a demographic approach*

The existing large-scale monitoring efforts developed under the Program for Regional and International Shorebird Monitoring (PRISM) are aimed at providing population size and trend estimates, along with collecting accompanying environmental data to assess habitat use and help infer range and distribution. However, the current PRISM program does not provide information on the mechanisms behind apparent declines (e.g., poor nest success and survival, chick survival, or adult survival) and when (e.g. breeding, migration, non-breeding) shorebird populations are most likely to be limited. Determining the stage of the annual cycle when shorebird populations are most negatively impacted will allow targeted conservation actions in the future to address population declines. Therefore, these two programs complement each other, and together provide information critical to large scale shorebird conservation.

## *Network participation*

The Network is open to participation by any collaborators who are actively conducting shorebird studies in the sub-Arctic and Arctic regions of the North American or Russian Arctic, and can implement the protocols designed by the group as a whole. Current participants are leading field work in the Russian, Alaskan, and Canadian Arctic (Figure 1). Study sites include camps near Nome, Cape Krusenstern, Barrow, the Ikpikpuk River, the Colville River, Prudhoe Bay, and the Canning River in Alaska, the Mackenzie River Delta, Bylot Island, East Bay, Coats Island, Igloolik, Burntpoint Creek, and Churchill in Canada, as well as the Chaun Delta and the Lower Khatanga River in Russia.

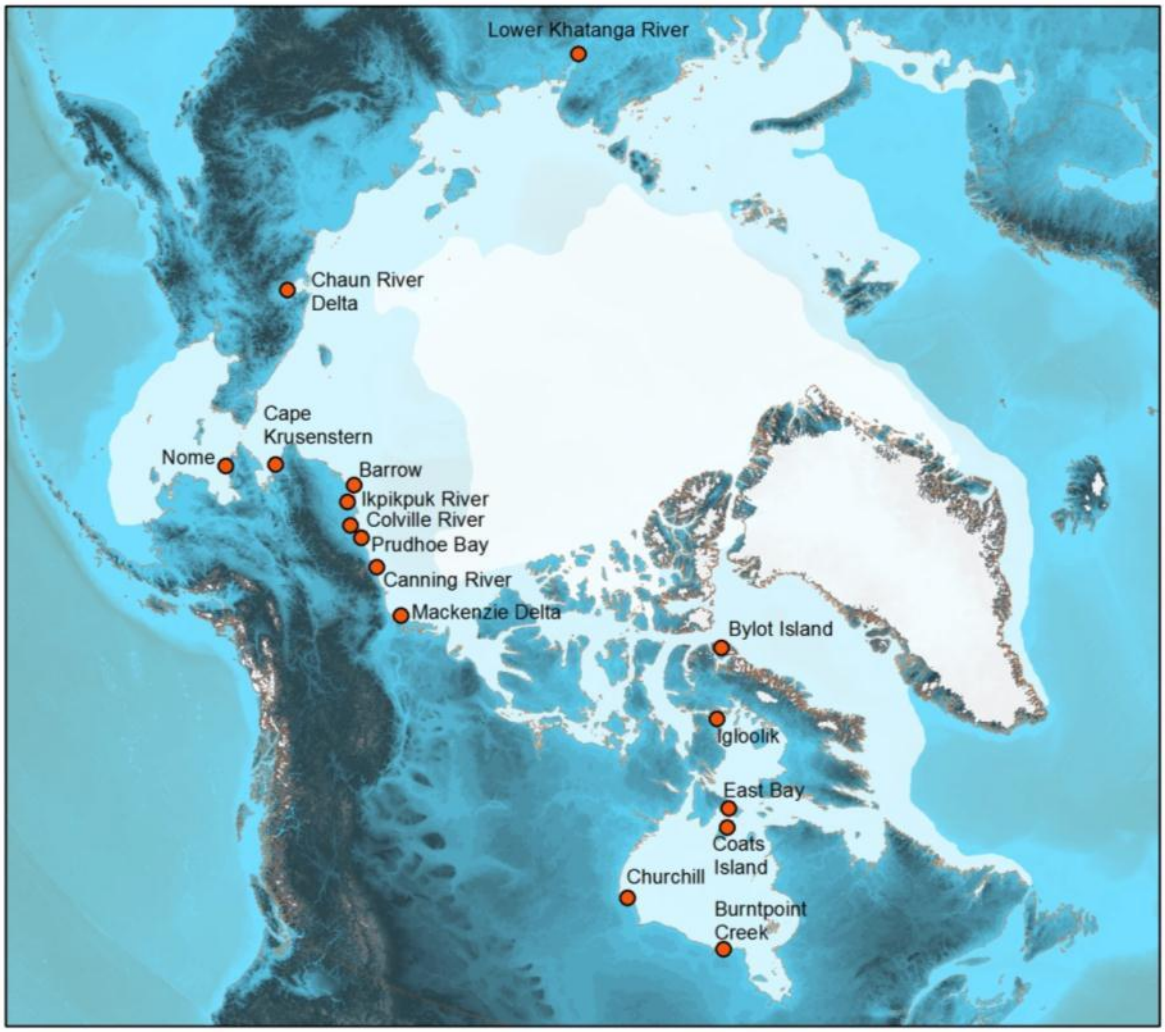



Figure 1. Arctic Shorebird Demographics Network study sites across the North American Arctic for 2012. Map prepared by J.F. Lamarre.

## *Network objectives*

Our overall objective is to assess factors limiting population growth of shorebirds through an international, large-scale collaborative effort that spans sites from Western Alaska to the Eastern Canadian Arctic and Russian arctic, and includes field work for up to 5 years per study site. At each site, we will also collect information on environmental and ecological variables that may influence these factors. This approach will allow a large temporal and spatial perspective on factors limiting shorebird populations in the Arctic. Our specific objectives include:

1) Collect demographic data (primary focus is on adult survival and nest survival) on a select subset of Arctic-breeding shorebirds (hereafter termed “focal species”).
2) For common species at a subset of sites, collect data on egg hatchability, adult and brood survival, mate and site fidelity, juvenile survival/recruitment, and age at first breeding on species.

3) Document contemporary patterns of species presence and abundance of shorebirds, and when possible, assess how species assemblages and abundance have changed historically.
4) Document breeding chronology and habitat use.
5) Collect information on the phenology and abundance of avian and mammalian predators of shorebirds and alternative prey (e.g. lemmings).
6) Collect data on local weather and snow conditions, the timing of insect emergence and changes in abundance throughout the season, inter-annual patterns in habitat characteristics, and other variables that will help assess impacts of climate change on shorebird breeding ecology.
7) Maximize the biological capacity of the Network by participating in projects that span large geographic and temporal scales. This includes investigations of shorebird health, migratory connectivity, and ecotoxicology.

## *Focal species*

Prior to formation of the Network, site leaders indicated a variety of species were present at planned Network sites (Table 1). Based on the number of nests likely to be found and the number of adults expected to be captured (ideally 50 nests monitored and 30 birds captured and marked/site), we initially chose to focus our efforts on Semipalmated Sandpiper (*Calidris pusilla*), Dunlin (*C. alpina*), Western Sandpiper (*C. mauri*), Red-necked Phalarope (*Phalaropus lobatus*) and Red Phalarope (*P. fulicarius*), designating them as first tier focal species. Pectoral Sandpiper (*C. melanotos*), Whimbrel (*Numenius phaeopus*) and Semipalmated Plover (*Charadrius semipalmatus*) were initially chosen as second tier focal species due to lower nesting densities or fewer sites with adequate numbers. We anticipated that project leaders at each field site would wish to study other species and other topics. Being part of the Network does not prohibit this, but only requires that efforts are made to collect all or portions of the data outlined in Tables 2 (a, b). Since the project's inception, additional sites have joined the Network and new species have become Tier 2 species (e.g., White-rumped Sandpiper and American Golden-Plover).

Table 1. Original Network focal species, avian target species, and relative abundance of each species at each Network site.

| | **Alaska** | | | | | | | **Canada** | | | |
|---|---|---|---|---|---|---|---|---|---|---|---|
| | **NOME** | **CAKR** | **BARR** | **IKPI** | **COLV** | **CARI** | **PRBA** | **MADE** | **BYLO** | **EABA** | **CHUR** |
| SESA | 1 | 1 | 1 | 1 | 1 | 1 | 1 | 2 | 0 | 2 | 2 |
| DUNL | 2 | 1 | 1 | 1 | 2 | 1 | 2 | 0 | 0 | 2 | 2 |
| WESA | 1 | 1 | 2 | 0 | 0 | 0 | 0 | 0 | 0 | 0 | 0 |
| RNPH | 1 | 2 | 2 | 2 | 1 | 1 | 0 | 2 | 0 | 0 | 2 |
| REPH | 1 | 0 | 1 | 2 | 2 | 1 | 0 | 1 | 2 | 1 | 0 |
| PESA | 2 | 2 | 1 | 0 | 2 | 2 | 0 | 1 | 2 | 0 | 0 |
| AMGP | 0 | 0 | 2 | 2 | 0 | 2 | 0 | 2 | 2 | 0 | 2 |
| SEPL | 2 | 0 | 0 | 0 | 2 | 2 | 0 | 0 | 0 | 2 | 1 |
| WHIM | 0 | 0 | 0 | 0 | 0 | 0 | 0 | 1 | 0 | 0 | 2 |
| WRSA | 0 | 0 | 0 | 0 | 0 | 0 | 0 | 0 | 2 | 2 | 0 |

1 = common breeders (*n*=30 pairs/year), 2 = low abundance (*n*<30 pairs/year), or represents transient populations that have large annual variation in site fidelity, 0 = not present at site in sufficient numbers. Network sites: NOME: Nome, CAKR= Cape Krusenstern, BARR= Barrow, IKPI= Ikpikpuk River, COLV=Colville River, CARI=Canning River, PRBA= Prudhoe Bay, MADE= Mackenzie Delta, BYLO=Bylot Island, EABA= East Bay, CHUR=Churchill. Sites that joined the Network after it

was underway and are not shown include Lower Khatanga River, Chaun River Delta, Igloolik Research Station, Coats Island, and Burntpoint Creek.

## *General framework and Network monitoring strategies*

The Network uses on-going projects and field camps and helps support new sites that are willing to contribute by collecting some level of demographic data. The Network maintains flexibility by encouraging project leaders to choose the intensity of effort that is reasonable for each camp. Two priority levels have been established to accommodate different field and logistic efforts occurring at camps, while providing standardized data that are still comparable. Priority level 1 includes minimum Network monitoring efforts, whereas priority level 2 requires more intensive sampling methods. Table 2a provides a summary of the difference between minimum and intensive efforts for assessing adult and nest survival. Additional plots or a more intensive nest search effort are needed in low density nesting areas to achieve adequate sample sizes.

## *Minimum nest search effort*

This level of nest searching effort will focus on finding a sufficient number of nests necessary for adult survival estimates (n = 30). A loosely defined search area should be established the first year. Care should be taken in area selection, since the search area ***must remain*** consistent across years (Figure 2). This area will be searched, focal species nests will be located and monitored to determine fate, and adults will be captured and marked for adult survivorship. The goal is to find 20 to 30 nests/focal species inside the study area and to uniquely mark 30-50 adults/year/species. If you capture less than this, or if you only have 30 pairs of a particular focal species at your site, this is still okay – the uncertainty around your survival estimates will just be greater. This effort will yield adult and nest survivorship estimates for the focal species.


Figure 2. Example of a Network site layout that follows the intensive approach for deriving nest survival estimates. Figure includes two intensive nest search plots (white squares), a greater search area for capturing adults off plot, and ecological monitoring components (e.g. predator surveys, food resources and lemming transects)

## *Intensive nest search effort*

Intensive nest search effort will require establishing permanent nest survival plots where we attempt to find and monitor nests of all shorebirds and other avian species. Nest survival plots (Figure 2, white squares) will have standardized intensive nest searching methods and effort. Plots can vary in size from 10 –ha to approximately 36 –ha depending on the nest density of the area. Plot size and shape may be variable across Network sites but each site's plot ***must remain the same across years***. There are 2

approaches to the intensive nest search effort: 1) banding all individuals both within the intensive nest search plots and the larger study area or 2) banding individuals in the larger study area but **not within** the intensive nest search plots. Both efforts will yield adult and nest survival estimates for the focal species in addition to standardized species diversity and nest density of tundra-breeding birds. The second approach will yield comparative analysis between nest survival estimates where adults were and were not banded so that the influence of human disturbance on nest survivorship can be assessed.

For both the minimum and intensive nest searching approaches, we will gather ecological and environmental monitoring data on potential shorebird nest predators, alternative prey (lemming counts), terrestrial and aquatic food resources, weather, snow melt, and snow and surface water cover. Details of how each parameter is monitored are illustrated in Table 2b.

## *Personnel considerations and seasonal work responsibilities*

The minimum goals of the Network can be accomplished with a 3-4 person crew that arrives shortly before the snow has begun to melt and departs approximately 1 to 2 weeks after peak hatch. However, a crew size of 4 to 5 is more suited to accomplish all of the objectives. Ideally, two people work for the first month of the season as primary nest searchers before switching to helping two other people who resight banded birds from prior years, band new birds, assist with rope dragging and help with Network side projects. All staff work to collect the environmental variables. Table 3 illustrates an abbreviated work schedule for the season.

Start dates are dependent on annual variation in snow melt and approximate time when the tundra becomes snow-free. If possible, camps should be established shortly before or when snow melt is just beginning (typically a 4-7 day period). Field season lengths vary by site and at a minimum are 6 weeks (this includes 1 week of set up, 3 weeks of incubation and 2 weeks of hatch). Seven weeks is better to capture the variation in initiation and hatching dates and a maximum of 10 weeks is needed if brood-survival or post-breeding studies are conducted.

Table 2a. Priority level (1 = primary, to meet minimum goals; 2 = secondary, to meet intensive), Network objectives, methodology, metrics, and workforce required to accomplish objectives for shorebird demography studies.

| Priority level | Objective (s) | Methods | Example metric (s) | Workforce required for min. Network site | Workforce required for intensive Network site |
|---|---|---|---|---|---|
| ***Shorebird adult demography, migratory connectivity and other species-specific ecological traits*** | | | | | |
| 1 | Monitor apparent annual and within-breeding season adult survival of focal species | Capture and color band adults for resighting | Adult survival, mate and site fidelity, natal philopatry | Mark at least 30 individuals within or off nest plots.<br><br>Resight opportunistically within season.<br><br>1 Resight/Bander: 8-10 hrs/day for 4-6 weeks | Mark ≥ 30 – 50 individuals within or off nest searching plots.<br><br>Resight opportunistically within season.<br><br>2 Resight/Banders: 8 - 10 hrs/day for 4-6 weeks |
| 2 | Determine age structure of focal species populations for demographic modeling (applies primarily to WESA, SESA, and DUNL) | Age determination of captured birds via plumage in the field or stable isotope techniques in the laboratory | Age at first breeding, population age structure | | |
| 2 | Collect appropriate samples from individual birds or equip birds with instruments for Network side-projects and collaborative studies | Collection of feathers and blood. Equip birds with instruments, etc. | Disease or contaminants, migratory connectivity | | |
| ***Shorebird breeding ecology*** | | | | | |
| 1 | Document nest survival | Nest fate, regular nest monitoring | Hatch success, nest survival | Only monitor nests (for survivorship) on those where birds are trapped. | Intensive and standardized nest searching effort with regular monitoring visits. |
| 2 | Document species assemblages and abundance | Standardized nest searching effort / unit area – must be consistent across years at a given site. | Identify species' presence/absence, nesting propensity, nest density<br><br>Territory mapping | Depends on size of search area and site density of focal species. | Depends on # of plots, 1 full time nest searcher/ 16 ha plot in high density areas ~ 8 hours/day for 4 weeks |
| 2 | Document breeding investment | Count and measure eggs | Clutch size, egg size | | |
| 2 | Document breeding phenology | Nest age determination | Initiation date | | |

Table 2b. Priority level (1 = primary, to meet minimum, 2 = secondary, to meet intensive), Network objectives, methodology, metrics and workforce required to accomplish objectives for ecological monitoring.

| Priority level | Objective | Methods | Metric(s) | Workforce required for minimum | Workforce required for intensive |
|---|---|---|---|---|---|
| ***Ecological monitoring*** | | | | | |
| **1** | **Predator – lemming index:** Document relative abundance of avian and mammalian predators | Regular (daily or weekly) counts within study area. Point counts for some sites | Abundance estimates or minimum counts. | Min seasonal survey -- 3 times/season (early, mid, and late) | Daily predator/lemming counts |
| **1** | **Lemming index:** Document relative and seasonal variation in abundance of lemmings | Winter nest line transect, daily observations quantified by person-hour effort during "low" lemming years, line transects in "high" lemming years, live trapping at some sites | Index to over-winter abundance, seasonal variation in relative abundance in live animals | 1 early season nest survey<br><br>Seasonal (early, mid, late) abundance checklist counts, or weekly abundance transects on "high" lemming years | 1 early season nest survey<br><br>Weekly lemming checklist counts, or weekly abundance transects on "high" lemming years<br><br>Live trapping |
| **1** | **Daily avian/mammal species list:** Document presence/absence of species; and large changes in number of individuals | Technicians enumerate observations throughout day | Daily check list with estimates of numbers seen, effort, and locations visited. | Daily 20 mins. | Daily 20 mins. |
| **1** | **Snow and surface cover:** Monitor snow melt progression and changes in surface water | Plot surveys | % snow / water/ land cover at fixed interval | 2 hrs/search area every other day, first 5 to 15 days of season, then once/week rest of season | 2 hrs/ plot every other day, first 5 to 15 days of season, then once/week rest of season |
| **1** | **Food resources:** Document seasonal change in insect emergence and abundance: Terrestrial (wet and dry locations) and aquatic (surface sampling). | Modified Malaise pitfall traps, aquatic sweep nets or passive pop bottle sampling in tundra ponds | Three-day estimatesof insect mass, abundance, species richness. | 1 -2 hours/day every 3rd day for sample collection and numeration during peak emergence, less frequent early and later | 1 -2 hours/day every 3rd day for sample collection and numeration throughout field season |
| **1** | **Weather:** Document within and inter-annual variability in weather conditions | Establish and monitor automated weather stations or obtain data from nearby (within 5km) established stations | Daily Min/Max temp, precip., wind speed/ direction | 1 day installation. ½ hr weekly download of data | 1 day installation. ½ hr weekly download of data |

Table 3: Seasonal work schedule for minimum and intensive efforts. Gray indicates time needed for both minimum and intensive effort sites, and black lines indicate intensive effort sites only.

| | Week 1 | Week 2 | Week 3 | Week 4 | Week 5 | Week 6 | Week 7 | Week 8 |
|---|---|---|---|---|---|---|---|---|
| Plot setup | | | | | | | | |
| Nest searching | | | | | | | | |
| Nest monitoring | | | | | | | | |
| Resighting | | | | | | | | |
| Banding | | | | | | | | |
| Food resources | | | | | | | | |
| Predators | | | | | | | | |
| Lemmings | | | | | | | | |
| Snow / Surface | | | | | | | | |

## *Geographic information datum and coordinate requirements*

Geographic information must be collected for study locations (plot locations, weather station, grid stakes within plots, transects lines, predator count locations, etc.) and boundaries of the study areas using a GPS unit. To ease future GIS applications, we insist all data be collected (or converted prior to data submission) in **latitude/longitude decimal degrees** (e.g., 56.3643°N; -145.78675°W) and the **WGS 84 (or NAD 83)** datum. These data should be saved in the geographic meta-database that accompanies each camp (*see data forms Network site establishment form).*

Adjust time on all GPS units (e.g., -9 hours from Greenwich Time for Alaska). Refer to: http://wwp.greenwichmeantime.com/time-zone/north-america/canada/time-zones.htm to obtain Greenwich Time zone for your camp.

# ADULT RESIGHT METHODS

## *Objectives*

1. Estimate annual survival of individually color-marked focal species at each Network site
2. Obtain between season resighting rates of individually color-marked focal species at each Network site

To estimate survival and resighting rates, we need to record the presence of as many birds marked in previous years as possible. The primary way we do this is to recapture them at nest sites or resight them either near their nest or when found opportunistically as people conduct work around the study area. Obtaining survival estimates is one of the primary goals of the Network and subsequently a fair amount of effort needs to be directed towards this task.

## *Recapture or resighting at nests*

Many times it will be unknown if an adult attending a nest is marked already. It could be that one or two adults are already marked. Some individuals may become quite skittish and will not stay around the nest site as you approach and thus you never know they are marked until you capture them (e.g.

phalaropes and Dunlin). Other individuals will stay very near the nest, especially later in incubation when they become protective of the nest itself. For the latter individuals, it is possible to read the color band combinations of the adults instead of recapturing them. This can be particularly useful if you are having trouble recapturing a bird. Not recapturing the bird, however, prevents you from obtaining valuable data including confirmation of band colors and metal band number, body size measurements, molt, and samples (blood, fecal material, feathers, etc.). It is best to recapture birds if time allows. For those birds not recaptured, b**e sure to transfer all resightings obtained at a nest to the Resighting Form.**

## *Opportunistic resightings away from nest sites*

Whenever an observer sees a color-marked individual, an attempt should be made to record and verify the unique color combination (hereafter color combo) of the bird. These opportunistic resightings will be used to confirm annual survival if you are unable to recapture individuals on the nest (i.e., adult either does not nest or the nest fails before you can recapture adults). Resight birds as often as you can, and attempt to obtain ***at least 2 visual 100% confident resights*** – more if the resighting is not 100% confident per sighting. If birds have been captured they do not need to be resighted repeatedly afterwards. Opportune times to resight color-marked adults include during initial visits to plots when available open habitat is limited and few birds are nesting, and when you are carrying out other tasks (snow/surface cover, lemming winter nest survey and other regular surveys). You can also resight birds during regular nest searching or banding should a marked bird present itself. Good times and places to search for marked birds are during early break-up when birds are concentrated near the first open water areas to feed and there are few nests to find. Another good time is after most birds have lost their nests but are still present on the study area. They will frequently feed in the study area for several days and are quite approachable. Observers should also record band combinations of birds seen off plot when traveling to and from camp or during other activities. Reading color band combos is much easier during the early morning or evening hours (04:00 -08:00, 17:00 – 21:00) when the sun is low in the horizon and at your back.

Some species return to breed near their previous breeding sites, and knowledge of past breeding locations for marked birds may therefore help to focus search efforts. Similarly, a list of birds recently marked or resighted in recent years will help to determine whether additional resighting effort is needed for certain species. **Summaries of birds that are expected to be present in the plots could serve as a useful resource for workers in the field, and should be prepared ahead of the field season**. Similarly, it is useful for workers to keep track of the location and color schemes of birds banded in the current season in their field notebooks, so that undue effort is not expended to read bands that were applied in the current year. Standardized method for banding birds in each year can help field personnel know what color schemes to look for (i.e., a form of cohort banding where the flag and site color band code are always placed on the same part of the bird's legs in a given year).

If you do resight a marked bird at a nest, be sure to enter its color band combination in the following places:

- For camps using ASDN nest and banding books:
    - Record color combination in the nest data book to indicate that you have identified the parent(s) of this nest
- For camps that do not use these forms, notes can be taken in field notebooks
    - Transfer all resights at the end of each day from field note books to the *resight data form*. It is probably best to put one person in charge of verifying the daily resighting data and ensure it gets completed by all personnel.

## *Suggestions for how to resight shorebirds during the breeding season*

Shorebirds can be difficult to resight during the breeding season because their legs are concealed by vegetation or terrain. Consequently, we rely on both engraved flags (at least for focal species) and unique color combinations. The engraved flags are easy to read if the bird is in the open and the flag is oriented correctly, but difficult to read in vegetation. The unique color band codes are easier to read in the vegetation. Below we describe how to collect resighting data; data should be recorded in the *resight data form*. These forms can be put into the banding and nesting record books or used by themselves. We encourage people to write on the forms as opposed to a Rite-in-the-Rain notebook because the form prompts people to record all the data required for easy editing and data entry later.

Before people go into the field they should familiarize themselves with the colors and unique study code (e.g., dark green flag above red band) used at a particular study site. The most efficient way to gather resighting data is to either take pictures of the birds or establish a short hand system for taking notes in the field that allows you to record data as you gather it. For example, you can make quadrants in your field notebook in order to keep track of the colors you have figured out. Each quadrant represents different parts of the legs and breaks the color combination into 4 quadrants (Figure 3). Record colors from top to bottom, from the **bird's left** to the **bird's right**. Color codes are listed in Table 4. The following graphics show the standard shorebird short hand for recording marked shorebirds via resighting, with examples of the color-band combinations from the pictured bird.

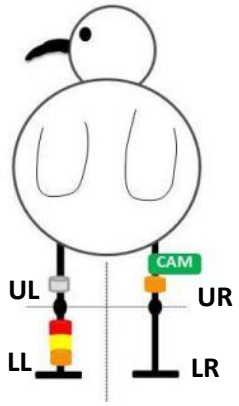



Figure 3. Example of ASDN color marked bird as seen from behind.

**Key to codes for recording color band combos:**
UL = Upper left - *left tibia tarsus*
LL = lower left - *left tarsus*
UR = upper right - *right tibia tarsus*
LR = lower right - *right tarsus*
\- = no bands were present
?? = unknown color or bands
/ = separates tibia tarsus from tarsus
: = divides left and right leg

*Example of short hand notation*:
_UL_/_LL_:_UR_/_LR_

This bird's colors can be noted as such:
**m /r,y,o :gfe, o/- engraved flag code: CAM**

Table 4. List of color band marker types and standard color codes for unique Darvic colors.

| Code | Marker type | Code | Marker type |
|---|---|---|---|
| m | metal band | r | red |
| y | yellow | w | white |
| bk | black | gy | gray |
| dg | dark green | gf | dark green flag |
| db | dark blue | bkf | black flag |
| lg | light green | wf | white flag |
| lb | light blue | wfe | white flag engraved |
| o | orange | gfe | green flag engraved |

**Data to record for each bird resighted**

1. Date: dd-mm-yyyy
2. Time: 24-hr
3. Observer: initials
4. Species: 4-letter BBL/CWS code
5. Plot or study area: code specific to each site
6. Reference plot stake/landmark: code specific to each site
7. GPS Location (lat/long): NAD83 Decimal degrees
8. Color-band combo upper left (UL)
9. Color-band combo lower left (LL)
10. Color-band combo upper right (UR)
11. Color-band combo lower right (LR)
12. Engraved flag code (if observed):
13. Observer confidence scale: (select one)

    **3)** All bands observed and recorded. Confident that location and colors of bands are correct on all parts of legs OR confident in engraved flag code (if present). Observed bird and checked notebook several times to make sure written down correctly.

    **2)** Most or all bands observed and recorded; if all bands recorded, exact location/order not certain, or colors not certain (i.e. red (r) or orange (o); read part of alpha-numeric flag code (if present); recorded in notes of all known/unknown parts of leg.

    **1)** Locations of several color bands observed, although not all bands seen; exact location of some bands unknown; recorded in notes of all known/unknown parts of leg.
14. Notes: (please record all that apply)
    Paired with color combo
    Behavior: territorial, singing, other
    General location
    Notes of colors seen or not seen very well (i.e.: "confidence "3" on all but UL. Could see 1 yellow, but couldn't see what was above it…" or "confident of band combo but R and L legs may be switched")

## *How to record resighting data*

The *resight data form* is for recording opportunistic observations of marked shorebirds **at or away** from the nest. DO NOT use it for recording captures of birds. These will be in the banding database and be recorded as a recapture.

### *Resight database and verification of resighting records*

Each sighting needs to be verified by the banding records to confirm that this color-band combination has been used on an individual of that particular species either this year or in a prior year. At the end of the day resighting records should be verified by confirming the color combo in the master banding database (accumulation of all banding data from that site in one file). Daily verification allows the observer who recorded the combination to help determine a plausible combination should the one written down not exist in the banding database. For example, an observer may have confused a white band for a yellow band (due to fading). The observers' recent memory can help verify resighting records. Care should be taken to retrieve all records from field personnel if they don't enter the data on a standard sheet in the banding or nest record data books (the recommended approach). See "readme" file in electronic database for more specific instructions on verifying records.

## ADULT SURVIVAL CAPTURE METHODS

### *Objective*

We would like to capture and uniquely mark at least 30 (but more is better – 50 to 100 is ideal) individuals of each focal species per year and make a concerted effort to resight individuals in subsequent breeding seasons. Our efforts to mark birds on the breeding grounds will also create opportunities for resighting individuals during migration and on the wintering grounds.

### *Unique marking scheme*

All birds captured for Network projects will be banded with a unique government-issued metal band, plain or alpha-numeric flag, and color combination. Each site will also have a unique site code color associated with the alpha flag (Table 5). Four of the focal species (e.g. SESA, WESA, DUNL, and PESA) will be marked with engraved alpha-numeric flags in addition to unique color combinations (some sites have discontinued marking PESA). Other species captured will be marked according to the project leaders' discretion, although all banding should be coordinated through the appropriate country's bird banding laboratory. The Network will help organize the engraved alpha-numeric codes but will not organize the unique color bands for individual birds (see instructions and tips below for generating color combinations). Each project leader is responsible for coordinating color band codes for species in their study area and for using site-specific codes that are coordinated through the Pan American Shorebird Program (PASP) currently supported by the Canadian Bird Banding Office (bbo_cws@ec.gc.ca), and the East Asian Australasian Flyway Partnership (for species migrating from the Arctic to Asia).

Unique site codes are listed in Table 5. Keep in mind that there are other people banding all of the species we work on so you must stick to your assigned site code to avoid banding multiple birds at different sites the same way. In years where you band many birds, you can develop more color combinations by separating the three unique color codes placed on the leg opposite to the cohort color scheme (e.g. place one color band above and two color bands below the knee joint).

### *Problem species*

There has been some concern about placing flags on phalaropes due to their use of cold, marine waters during much of their life. Icing of these bands could be potentially life-threatening to these species. Because the bands are so difficult to read at any time of the year (i.e., the birds need to be captured to determine their band combination), we suggest not placing flags on these birds but simply using your

site color band code, along with unique band combinations on the opposite leg (i.e., still use the template below but remove the green or white flag).

Table 5. Unique site-specific color band combination codes for Network Sites.

| Study site location | Flag color | Site code |
|---|---|---|
| ***Alaska*** | | |
| Nome | dark green | dk green flag and dark blue |
| Cape Krusenstern | dark green | dk green flag over orange |
| Barrow | dark green | dk green flag and yellow thru 2009, red in 2010 |
| Ikpikpuk | dark green | dk green flag and light blue |
| Prudhoe Bay | dark green | dk green flag and light blue |
| Canning River | dark green | dk green flag and dark green |
| Colville | dark green | dk green flag and white |
| ***Canada*** | | |
| East Bay/Coats Isl. | White | white flag over red/orange |
| Churchill | White | white flag over dark blue |
| Bylot Island | White | white flag over dark green |
| Mackenzie Delta | White | white flag over light green |
| Burntpoint Creek | White | white flag over yellow |
| Igloolik | White | white flag over light blue |
| ***Russia*** | | |
| Chaun River Delta | Light blue | light blue flag over/above orange |
| Lower Khatanga River | Light blue | |

## *How to apply metal and color bands to a bird*

Metal bands are applied with special banding pliers (not needle nose pliers). To remove metal bands from the wire string, use a band spreader or if your banding pliers have a split pin built into the side, insert split pin into center of band and open handles evenly. Try to open the band evenly. Place open band in the proper sized hole on pliers, and then slide around leg where specified for your site's color marking scheme and close gently. Bands should be placed on birds in sequence if possible to make reporting data to the Bird Banding labs easier. Incoloy bands (a more durable metal type) are better than aluminum bands. A special request can be sent to the banding offices to obtain incoloy bands.

Color bands for smaller species (size 1B to 3) are usually 'butt-end' bands, similar to metal bands, while those for species size 3A and larger are usually 'wrap-a-round' bands. Butt-end bands are applied with a thin metal 'shoehorn' applicator: a smaller size applicator is used for bands up to 1A, and larger size applicators for size 2 and larger bands. The band is placed on the applicator with the opening in the band towards the depression in the shoehorn, and the band is slid up the applicator until the band is sufficiently open to fit on the leg. The applicator is laid against the leg, and the band is slid off the small end of the applicator onto the bird's leg. It is important to stretch these bands no more than is necessary to put them on the leg, and to ensure that the color band is completely closed on the leg. It may be necessary to click the edges of the band under each other with one's fingers to ensure that the

band is completely closed. Wrap-a-round bands are twirled carefully onto the bird's leg, ensuring that the leg is not injured and the bands are not opened more than necessary. Again, these bands may be tightened with the fingers after they are on the bird. Ensure that the bands rotate freely around the leg, but are not so loose that they can pass over the 'knee' joint or 'ankle'. Wrap-a-round and butt-end bands should be permanently sealed with a battery-operated soldering gun. Only solder on the flat edge of the band, not the top or bottom areas near the bird's leg.

Always put an entire color combination on a bird (including the country flag and unique site code on each as it is banded). **Never** make up a color combo or band a bird with a partial combo. If a bander is short on color bands in the field, the bander should place a metal band on the bird, release it, and return later to recapture the bird after obtaining necessary supplies for correct color banding.

If the bird you capture has been banded previously, be sure to check your "double-banded bird" cheat sheet and correct the bands if necessary (see below). DO NOT change the bands on a bird banded uniquely by someone else (e.g., a bird banded at another location), and only add your site code color to the bird's leg should this previously banded bird not be uniquely marked (e.g. bird has a single white flag with no engravings).

## *Applying focal species alpha-numeric flags*

Use shoehorn applicators to place the flag on the bird (**with the engraved numeric code upright – very important since certain codes can be read in either direction – e.g., E6 or 9E**) or use one's nails to open the flag slightly. Open the flag as little as necessary, so that the flag is not stretched (otherwise, remove and reshape later). Flag tabs should be sealed along the edges with a battery or propane operated soldering pen (http://www.all-spec.com/products/BP860MP.html). If the gun malfunctions, the flag can also be sealed with Marley Solvent Cement or Superglue (e.g. cyanoacrylate). Glue is applied to the tabs of very slightly opened flags with an object such as the tip of a small screwdriver. Pliers or clothespins are used to hold the flag tabs closed for about 20 seconds until the glue is set. Then the pliers are carefully removed to prevent the flag from opening. **For alpha-numeric flags, be very careful not to damage the letters when soldering. If you cannot do this with the solder gun, then use glue to seal them.**

## *Instructions and tips for a color marking shorebird program*

Generating color band combinations to identify birds uniquely becomes increasingly tricky as more birds are color-banded in your study area through time. It is very helpful to have your color banding scheme well planned and organized, both before and during the field season, to accomplish a successful multi-year color-marking program. This will help you avoid having birds that either have duplicate or similarly banded combinations, making it hard to assess survival rates and other breeding parameters (mate and site fidelity). Below are a few helpful tips for generating, distributing, and managing color band combinations.

## *Generating templates for color combos*

Go to the Manomet website (http://www.manomet.org/arctic-shorebird-demographics-network) and download the example excel file ASDN_color_combo_template.xlsx. Each site may be able to use this spreadsheet by simply replacing the site-cohort mark color (this consists of a unique country flag and site color assigned to your location listed in Table 5). Each year, a new file should be created by moving the site-specific code to a new position on the bird's legs. By doing this, you essentially create cohorts for each year. If you have a lot of band combinations remaining from a prior year, you can also use

these although you will forego the cohort marking and it is essential that you check that color combinations remaining on your lists have not been used previously. The color combo Excel worksheets are printed and cut into unique cards by species and laminated on both sides to protect the paper. Compile a set of color combos for all species (see example below) and use a binder ring to hold them together, and place one of these inside each banding kit. **Banders must cross off combinations as they are placed on birds and write down the band number during banding to avoid banding two birds the same way.** You may wish to randomize the order in which the bands are applied to birds by simply moving excel cells around in a random fashion since it has been shown in some passerines that band colors can influence mate choice. For sites with <100 birds banded, you may wish to make a list of all banded birds that were previously banded and carry this with you in the field. It will be helpful for people resighting color banded birds (i.e., they will know possible combinations) and for banders who capture a bird that has lost a band (i.e., so they know how to fix the band combination).

Example template for a set of color band combinations

| **DUNL – IA** | | | | |
|---|---|---|---|---|
| **Upper Left** | **Lower Left** | **Upper Right** | **Lower Right** | **Band number** |
| **GFE,O** | **-** | **m** | **O,DG,O** | |
| **GFE,O** | **-** | **m** | **O,DG,Y** | |
| **GFE,O** | **-** | **m** | **R,O,R** | |
| **GFE,O** | **-** | **m** | **R,O,LB** | |
| **GFE,O** | **-** | **m** | **R,O,Y** | |
| **GFE,O** | **-** | **m** | **R,LB,O** | |
| **GFE,O** | **-** | **m** | **R,LB,R** | |
| **GFE,O** | **-** | **m** | **R,LB,Y** | |
| **GFE,O** | **-** | **m** | **R,LB,DG** | |

Issues to consider when generating codes (which the template should already take into account in most cases):

a) Avoid placing two bands of the same color next to each other - *it will be hard to determine from a distance if there are one or two bands*
b) Avoid using two bands that are similar in color in your banding plan (e.g., light blue and light green) - *it will be hard to differentiate these in poor lighting conditions, and they will fade and appear similar to one another in the future.*
c) Avoid placing three bands on the upper leg of small species – *feathers will obscure top color(s) when placed on the upper leg*
d) Do not use **just** a green or white flag in one location (e.g., lower or upper portion of a leg) on your bird - always put your site code next to the flag (either above or below the flag depending on your location color assignment – see below). If two people do this in different locations, we will get two birds with the same combination. This is only really problematic on the breeding grounds for species that are not site-faithful or when one of these birds is seen on migration or on the wintering grounds. Of course, if you do this once it will be the bird that is resighted in Mexico later that year (and you won't know if the bird was banded in Churchill or Bylot Island for example). Thus if you run out of site code color bands while banding, opt for placing on a single metal band as opposed to a partial band combination on your bird.
e) Consider carefully the placement location of bands on the bird's legs – we advise thinking about each species and where the bands are most easily observed. For species with small tibia, you

might place bands preferentially on the tarsus. For species that are frequently in tall vegetation, you might place bands preferentially on the tibia. Of course, as you move bands around across years to create cohort colors it will become increasingly difficult to place bands in the optimal spot. Ideally you should plan out how you will place bands on each species for the entire span of your study area to avoid problems one, two, or more years down the line.

f) Use of engraved flags is currently restricted to SESA, WESA, DUNL, and PESA. Engraved flags are helpful for identifying individuals on the open mudflats on the wintering grounds but should not be considered sufficient for identifying individuals on the breeding grounds. Engraved flags are difficult to read for birds moving through vegetation.

If there are a lot of band combinations not used, you could reuse the sheets in succeeding years. Be sure that people remember to cross off all band combinations used (i.e., double check the unused codes on leftover banding sheets were not put on birds in a prior year).

## *Pre- field season identification of birds with duplicate or problem band combinations*

Before field work begins each year, each camp should determine whether they have two birds banded with the same color band combination. This typically occurs when people fail to cross off a combination from their assigned list. Sort all of your banding records from prior years by upper left and lower left and upper right and lower right band combinations to find these problem birds. Then make a list of them and put the list (be sure to include metal band #) in all of your banding boxes. When people capture a bird that was previously banded in the field, the first thing they should do is check to see if this bird has a duplicate. If it does, the bands should be changed to remove a duplicate color combination. Of course, this only has to be done on one of the two birds.

This is a good time to also look for birds that have problem combinations. Perhaps people have left a band off accidentally or neglected to put a site color combination on a bird. Should you recapture these birds, you can place a full combination on the bird and avoid problems with resightings in the future.

## *Changes to color band combinations and metal bands.*

ANY CHANGES IN COLOR COMBINATIONS NEED TO BE CAREFULLY NOTED BY RECORDING THE BIRD COLOR COMBO AND METAL BAND WHEN RECAPTURED AND HOW YOU MODIFIED THE COMBINATION. THIS IS ESPECIALLY IMPORTANT IF YOU CHANGE THE METAL BAND IF IT IS WORN OUT AND NO LONGER READABLE. There are special data fields for these entries at the end of the electronic database file.

**Data recorded with each bird**

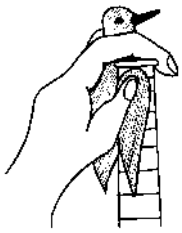
1. Book number
2. Page number
3. Year
4. Site name (use codes in readme file)
5. Bander (first name initial and last name e.g. loring for Lewis Oring)
6. Date (dd-mm-yy)
7. Time (24 hr)
8. Nest ID (unique code of spp and nest number)
9. Plot identification (unique plot code, alpha-numeric)
10. In or off plot (Was the bird captured on or off the plot)
11. How captured (bownet, mist net, walkin, standing decoy, other [define])
12. Capture status (laying, incubation, brood, post-breeding, unknown)

13. Band number (USGS/CWS 9 digit unique number)
14. Species (4-letter BBL species code)
15. Recapture between years
16. Recapture within years
17. Color combo: a: Upper Left b. Lower Left c. Upper Right d. Lower Right
    Use "," between color band colors or flags on one portion of the leg
    Flags: gf = dark green flag, gfe = engraved dark green flag wf = white flag, wfe =engraved white flag, engraved flag codes listed in an additional field
    Colors: o = orange, y = yellow, r = red, db = dark blue, bk = black, dg = dark green, lg = light green, lb = light blue, m = metal band;
18. Picture taken (yes or no, be sure to first take a picture of the banding sheet and then of the bird so it is possible to determine the identity of each bird)
19. Flight feather molt (score 0- 5, primaries 1-10, secondaries 1-10)
20. Tail feather molt (score 0- 5)
21. Body molt (score 0, 2 – 5, for head, neck, back, breast, and abdomen)
22. Exposed culmen (nearest 0.1 mm)
23. Total head (nearest 0.1 mm)
24. Diagonal tarsus (nearest 0.1 mm)
25. Flattened straightened wing (nearest 0.5 mm)
26. Bird Weight (nearest gram or nearest 0.1 if digital)
27. Bag Weight (nearest gram or nearest 0.1 if digital)
28. Final weight (calculated automatically in the banding electronic database)
29. Fat (score 0 – 7)
30. Blood for avian malaria, Amount of blood (in micro liters), type of capillary tube (Plain)
31. Blood for ASDN genetics in Longmire (Y/N) Amount of blood (in micro liters), type of capillary tube (Plain)
32. Blood for Methyl Mercury (Y/N) Amount of blood (in micro liters), type of capillary tube (Hep)
33. ASDN archive feathers sampled (Specify which feather was pulled and from where according to standard feather codes e.g. "brst, 10sL + 10sR= 10 secondary on the left and right sampled)
34. Moss dispersal feathers sampled (Y/N)
35. Fecal sample (Y/N), preservative used
36. Sex (Male, Female, unknown)
37. Method of sex (culmen, morphology, plumage, brood patch, cloaca size, wing, overall size, egg in oviduct)
38. Age (chick, HY, SY, AHY, ASY)
39. Method of age (e.g. plumage, weight, recapture)
40. Release status: band and release, band and escape, release unbanded, injured, band and release, mortality
41. If geolocator, record exact date and time removed from the bird (VERY IMPORTANT).
42. GPS location of nest
43. Band change (1/0 for Y/N)
44. Old band number: Numb. of the band that was removed from the recapture, if the band was changed.
45. UPPER left color combo: Color combo that was on the recaptured bird.
46. LOWER left color combo: Color combo that was on the recaptured bird.
47. UPPER right color combo: Color combo that was on the recaptured bird.
48. LOWER right color combo: Color combo that was on the recaptured bird.
49. Comments

## *Morphological measurements*

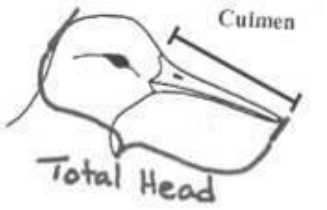
Figures from Prater et al 1977 or Gratto-Trevor 2004, Photos B. Lewis and S. Sapora/USFWS.

**Wing length**: maximum length with the wing *flattened and straightened*, measured with a wing ruler (to nearest mm) from the bend in the wing to the end of the last primary. Be sure to hold the wing close to the body, not at a right angle to the body when measuring.



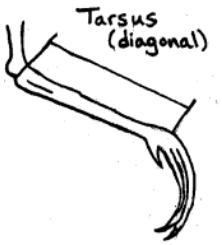
**Exposed culmen**: measured with calipers (to the nearest 0.1 mm) from the edge of feathering to the tip of the bill. Since bills of most shorebirds are very sensitive, hold bill lightly with the fingers, with the calipers resting on one's fingers and not the bird's bill. The calipers must remain **perpendicular** to the bill and not angled to measure a downturned bill. This is especially important for long-billed species such as Dunlin and Whimbrel.


**Total head**: measured with calipers (to the nearest 0.1 mm) from tip of the culmen to the notch at the back of the head at a perpendicular angle.





**Diagonal tarsus length**: measured from the slight indentation below the 'knee' joint to the indentation above the longest toe (to the nearest 0.1 mm).



**Mass:** measured with a hanging Pesola scale or a digital scale (to nearest 0.1 g). If using a Pesola scale hold by the top ring or hook and allow to dangle freely, providing protection from the wind. Place the bird in a weighed cloth bag, or plastic cone with the bill protruding from the bottom. The cone should be firmly attached to the teeth of the clip at the bottom of the scale. It is very easy to release shorebirds from plastic cones or cloth bags, by sliding them out into the palm of the hand until one can hold them in the banding grip. Mass should be the last measurement taken (or first before you take them out of the bag) as it is the best opportunity for a bird to escape during handling.

## *Molt*

Examining birds for body and flight feather molt can indicate age as well as provide information on timing and extent of molt, which is poorly known for most shorebirds. To describe **body molt** the bird is normally divided into five regions: head, neck, back, breast, and abdomen. Look for feathers emerging to detect molt, although be careful for cases where new feathers have already fully emerged. This is obvious when, for example, Red Phalaropes are no longer red but are gray. The extent of replaced body feather codes are as follows:

**0**: all old body feathers, **NOTE: there is no score of 1 here.**
**2**: a few new body feathers
**3**: about half body (30- 50%) replaced
**4**: most replaced (60 -90%)
**5**: all new (100%).

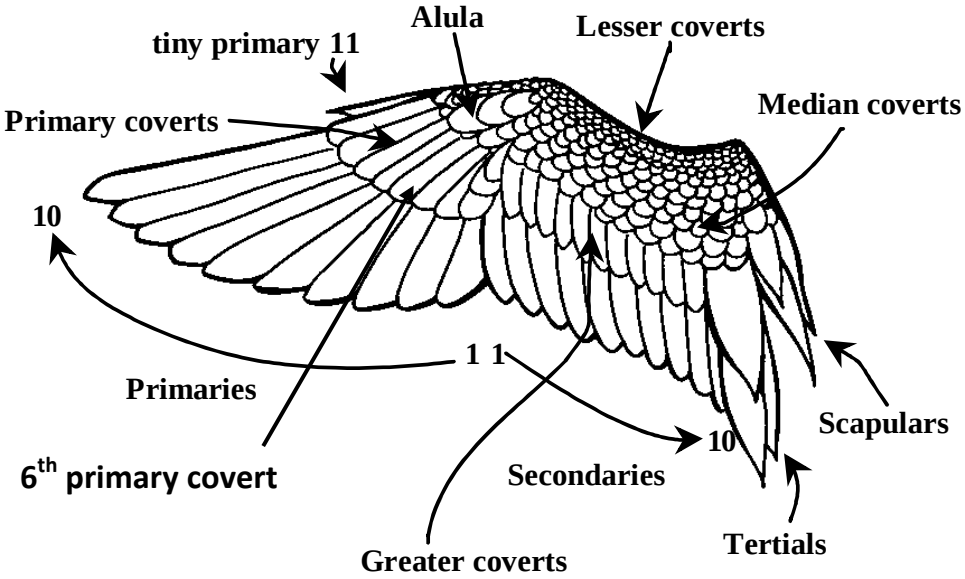
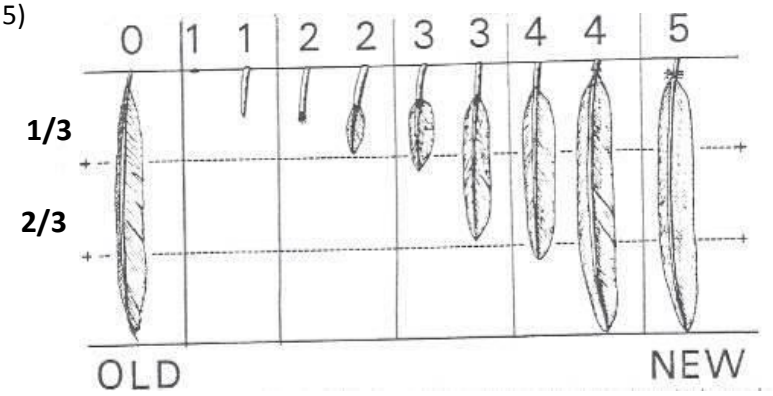



Figure 4. Specific names of each flight feather and their standard abbreviations are as following (pp=primaries, ss= secondaries). Figure from Gratto-Trevor 2004.

**Flight feather molt** scores are usually more complicated, with the condition of every primary, secondary, tertial and tail feather described (Figure from Ginn and Melville 1983):

Their condition is noted as follows (see Figure 5)

**0**: old feather
**1**: feather missing or completely in pin
**2**: just emerging from sheath to one-third grown
**3**: one to two-thirds grown
**4**: more than two-thirds grown but still with waxy sheath at base
**5**: new feather fully developed and without waxy sheath



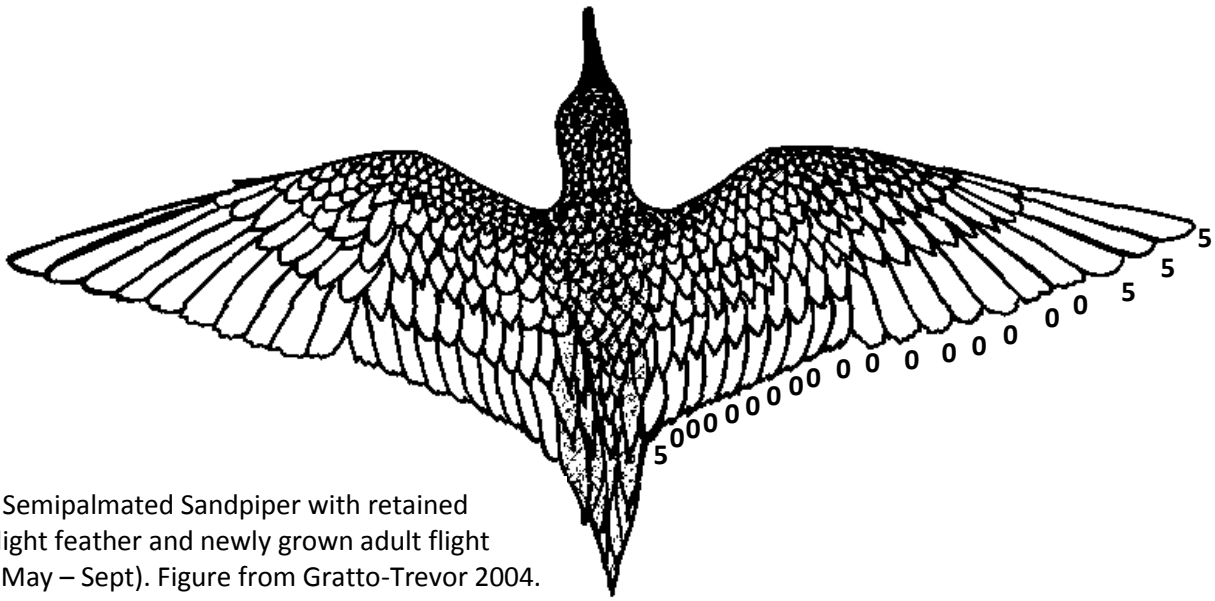




Figure 5. Semipalmated Sandpiper with retained juvenile flight feather and newly grown adult flight feathers (May – Sept). Figure from Gratto-Trevor 2004.

## *Sex determination*

For breeding shorebirds, sex can usually be differentiated by plumage, exposed culmen wing length, overall size, or behavior. However, there is significant geographic variation, prohibiting a standard morphological measurement across the Network. The Identification Guide to North American Birds, Part II (2008) can be used as a reference guide that includes subspecies measures to determine how the focal species can be sexed. For species with no observable unique traits, sex can be determined with genetic techniques. Evidence of recent egg-laying (extended cloaca) or even a bulge indicative of an egg inside a bird can also be used. This is especially helpful for sexually monomorphic species. There is a location on the data form to record how the bird was sexed – please be sure to fill this in as it allows us to ascertain confidence in the field sexing technique.

Culmen length is commonly the most sexually dimorphic measurement in sandpipers, with female culmen length averaging longer than males. Other measurements may provide more information in other species. Measurements must be used carefully, however, especially when a study begins in a new area or an area where different populations of the same species overlap. Moreover, the degree of overlap between sexes in measurements may vary from one breeding site to another. Field crew leaders may prefer to wait to determine the sex of a bird once both mates are captured and comparisons of morphometrics can be made. A final sex determination for each bird is typically done after the field season, and can incorporate knowledge of the size of the bird and its mate during this and previous field seasons, as well as information from genetic analysis and discriminant function equations.

**Appendix I** illustrates the specific criteria for determining age of three focal species (DUNL, SESA, and WESA). The Identification Guide to North American Birds, Part II (2008) can be used as a reference guide that includes subspecies measures to determine how the focal species can be aged. It is sometimes possible to determine the age of an individual by closely examining the condition of the flight feathers (primaries and secondaries) and the wing coverts (primary, secondary, median, and lesser coverts). During the breeding season adult birds can be separated into three groups. Second year (SY) can be identified by the presence of buffy edges on their innermost median coverts, and also by heavily worn primary feathers. After second year (ASY) have white edged innermost median coverts and, in general, will have less worn primaries and wing coverts. Birds that do not have these distinctive traits are aged as After Hatch Year (AHY). If you are uncertain about the age of adults, AHY is a conservative catch-all. Most birds are usually considered AHY rather than SY/ASY, please be conservative in assigning ages to all individuals. **Pictures of the wings will be used later to assign sex in a standardized fashion so be sure to take good pictures and know what parts of the wing are relevant**.

For *Calidris* sandpipers in general, most or all SY birds molt the most important outer primaries only, as well as inner secondaries. These birds may be identified as SY (between at least May through September) by the contrast between fresher outer primaries and more worn inner primaries. If all feathers had been molted the previous winter, outer primaries, which suffer the most wear, would be more worn than inner primaries. Note that the percentage of SYs in these species with this Partial Post Juvenal Wing (PPW, see Figure 6) molt can be variable among populations and years (e.g. Prater et al. 1977, Gratto and Morrison 1981, Nicoll and Kemp 1983). Individuals without the partial molt usually have not molted any primaries, but some undergo a complete molt.

To ensure consistency in aging of birds from feathers, we strongly advise taking pictures of all birds so that birds can be aged by one person at a later date (see below).

## *Fat*

Subcutaneous fat is yellow or orange in appearance and is stored just under the skin in three discrete areas with deposition occurring in the following order:

(1) The hollow in the furculum (wishbone) just below the throat at the top of the breast muscles;
(2) The hollow directly under the wing, essentially in the “wingpit”;
(3) The lower abdomen just anterior to the vent area.

Holding the bird on its back, gently blow the feathers away from the upper breast to expose the furculum. Then check under the wing and on the abdomen by blowing the feathers out of the way. Fat scores are subjective and require looking at many birds to be consistent. Blowing on the chest of a bird with a straw can be useful for concentrating wind in a particular location.

**Score fat as follows:**

**0)** No fat in the furculum or anywhere on the body

**1)** A very small amount of fat in the furcular hollow (less than 5% filled) but not enough to cover the bottom of the furculum. None or just a trace of fat under the wing, on the abdomen, or anywhere else on the body.

**2)** The bottom of the furculum is completely covered but the furcular hollow is less than 1/3 filled. A small amount of fat may be present under the wing, on the abdomen, or both.

**3)** The furcular hollow is about half full (from 1/3 – 2/3 full). A covering pad of fat is definitely present under the wingpit and, usually, on the abdomen.

**4)** The furcular hollow is full (2/3 to level with the clavicles). A thick layer of fat also occurs under the wing and on the abdomen.

**5)** The furcular hollow is more than full; fat is bulging slightly above the furculum. The fat under the wing as well as that on the abdomen is also well mounded. (Figure 6).

**6)** Fat is bulging greatly above the furculum. Large mounds of fat occur under the wings and on the abdomen.

**7)** The fat pads of the furculum, "wingpit," and abdomen are bulging to such an extent that they join. Nearly the entire ventral surface of the body is thus covered with fat, and fat even extends onto the neck and head. Such birds are nicknamed “butterballs.”

Figure 6. Magnolia Warbler with a great deal of fat in the furcular hollow and abdomen. This individual is a 5 on the fat score scale.

**<u>Photo documentation of all captured birds</u>**

<u>Objectives:</u>

1. Capture images of bird wings to verify age by plumage characteristics
2. Capture images of color bands for verification of color band combinations

Determination of shorebird age can be accomplished for some shorebird species by examining plumage characteristics (see Appendix I). In an effort to standardize our classifications and learn more about shorebird molt, we will take pictures of each adult’s wing once the bird has been banded. Ages will be assigned to birds in the field (see ageing section) and will be verified by the pictures. For species that have longer wings (i.e. DUNL, AMGP, LBDO) two pictures of the wings should be taken. It is very important to take a clear photo of **all inner wing coverts (greater, lesser, and median) and flight feathers (primaries and secondaries, see Figure 4)**. Using dark background and shading the bird while taking the picture improves the image quality (bright light dulls color richness).

At least 3 pictures should be taken of each individual, including the banding data form, the bird’s color combo, and the entire wing. Make sure your fingers are NOT covering the lesser, median and greater

coverts that are important for age determination. These photos should be taken in the same order each time, and labeled uniquely (see below).

| Content of image | File name | Example photo |
|---|---|---|
| Data sheet that includes the band number, date bird was banded and color combo | 230155644_dunl_data.jpg | Fig. 7a |
| Color bands and alpha-numeric coded flag | 230155644_dunl_combo.jpg | Fig. 7b |
| Inner wing: lesser, median and greater coverts (aka secondary coverts) | 230155644_dunl_innerwing.jpg | Fig. 7c |
| Outer wing: primary and greater coverts (aka secondary coverts) | 230155644_dunl_outerwing.jpg. | Fig. 7d |

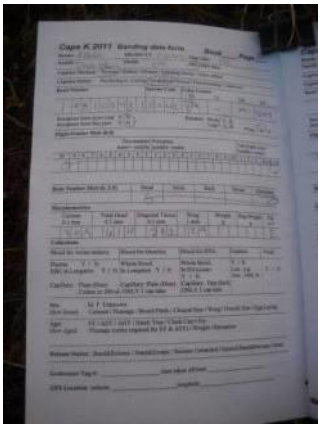


Fig.7a


Fig.7b

Fig. 7c

Fig. 7d

Figures 7 a – d. Series of photos taken to document adult marking including color banding and ageing characteristics of Dunlin. Photos: R. Gates/USFWS

## *Chick banding*

Nests are visited more regularly (typically daily) close to the expected hatch date to capture the chicks in the brief period that they remain in the nest cup. It usually takes about 48 hours for tapping chicks to break out of the egg shell and be dry enough to leave the nest (however, this process can be highly variable depending on species and weather conditions). Departure from the nest cup is also dependent upon weather conditions on the day of hatch. Chicks tend to leave the nest earlier on warmer hatch dates and can leave the nest in less than 24 hours. Several precautions are taken when working with nests that may contain hatching chicks. Observers should make sure predators are not watching them before they approach a nest and they should be sure they know the general location of the nest or chicks (if outside the nest). If hatch is not yet complete, banding should be delayed until all the young have emerged (unless two or three "fluffy" young are in the nest and the other eggs show no signs of hatching). Early banding may result in parents attempting to lead the few young that have hatched away from the nest prematurely.

The observer should check the nest notebook before approaching the nest to see how many eggs and thus how many chicks should be present; this also helps determine if any chicks are missing. Newly hatched chicks are cryptically colored and tend to disperse or hide if there is danger, so it is necessary to approach the nest carefully to avoid stepping on the chicks that are near the nest cup. Observers should look in the direction of the nest when approaching to detect a flushing adult's location. This is likely the location of the chicks. They may not all be together and they may not all be in the nest bowl. If not all of the chicks are initially present in the nest, the observer should gather up the chicks from the nest cup (place them in a chick bag), back away (about 5 to 10 meters), and watch the adult to find the remaining chicks. Brooding adults will typically make soft "gathering" calls to bring the chicks in; will look like a giant sitting puff ball as they move their feathers out to expose their brood patch; and their belly will often appear to be "moving" as the chicks rearrange themselves. Once you begin your approach to the brood, the adult will typically emit a "danger" call in which the chicks will either try to disperse or will lay flat on the ground and hide. The adult may do a distraction display but do not watch the adult once you begin your approach. Focus on the spot the adult flushed from and do not look away. As you approach the spot you think the chicks are in, walk slowly and look where you will place each foot.

Once all chicks have been located, move away from the area to band them. Newly hatched young are incapable of thermoregulation and are susceptible to chilling. During handling, the young should be placed in nylon insulated bags, and if cold outside, a chemical heat pack should be added (although do no place chicks directly on heat packs). Alternatively, chicks can be placed in typical bird bags and placed inside one's shirt. The bags should be loose enough to avoid suffocation, but the tops should be kept tightly tied to avoid escape. Weigh each chick and then place aluminum bands on each chick following the banding methods outlined in the adult banding section. Care should be taken to not close the band on the leg. If information on the sex or paternity of the chicks is needed, take a small blood sample from the metatarsal vein (prick with needle and soak up with filter paper). Return banded chicks to the nest cup (if this is where you found them) or near where the adult last was. Place all chicks next to each other in the same location (even if you found them in separate areas). It is best to work as quickly as possible to collect and band the chicks and get them back to their original location – however, take care to avoid stepping on or otherwise injuring chicks. The adults are typically the best indicator of chick locations so be sure to pay attention to them. Leave the area quickly after banding to allow parents to brood. To reduce negative impacts, we ask that people do not extend the time required to band chicks by taking pictures or by excessive handling.

**Data recorded with each chick**

| | | |
|---|---|---|
| 1. | book_page | number from raw data form |
| 2. | year | year of study |
| 3. | plot | unique plot code, alpha-numeric |
| 4. | plotLmt | Was the bird captured on or off the plot |
| 5. | species | 4-letter BBL species code |
| 6. | nestID | unique code of species and nest number nest searcher initials |
| 7. | EggChick# | 1-4 unique number of egg/chick |
| 8. | Date | dd-mm-yy |
| 9. | band# | USGS/CWS 9 digit unique number |
| 10. | UL | Color combo: Upper Left |
| 11. | LL | Color combo: Lower Left |
| 12. | UR | Color combo: Upper Right |
| 13. | LR | Color combo: Lower Right |
| 14. | Engraved | Engraved flag code |
| 15. | Weight | nearest gram or nearest 0.1 if digital |
| 16. | Cone_wt | nearest gram or nearest 0.1 if digital |
| 17. | Final_wt | Calculated by database |
| 18. | sample | was a sample collected? |
| 19. | sample_type | blood, tissue, feather |
| 20. | fate | dead, alive (should be confirmed by visiting nest on the next day) |
| 21. | Comments | |

## SAMPLE COLLECTION METHODS

See **Appendix A** for more specific details on the target sample sizes for this year, specific procedures, and what materials to order for collecting samples for ASDN core objectives and side-projects.

**Sample labeling is crucial for all of the collection methods. Inappropriate or difficult to read sample labels will require the sample to be thrown away. Please see labeling section for each sample listed below on page 67.**

### *Fecal samples*

Birds can be placed in plastic containers (small animal aquariums) until they poop. The bottom of the container should be lined with clean waxed paper (e.g., parchment/baking paper) to avoid sample cross-contamination from other birds. **Birds usually poop within 5 to 10 min of capture.** Samples will be taken as fast as possible after capture to minimize the effect of stress and to obtain a sample before the bird has voided the entire gut. We also recommend putting a chemical heat pack at the bottom of the container during cold days to keep birds warm. This will help you bleed the bird later.

- **Please wear gloves when handling the lining paper** while preparing the box for every new bird to prevent contamination of the sample with our skin microbiota.

- If birds trample through their fecal sample and it is not possible to collect an untouched portion of it, **please disregard the sample**. Bacteria present on the birds feet will be mixed in with the sample and can't be distinguished from gut microbiota. Phalaropes tend to do this, and it might help to reduce the

time they spend in the box to a few minutes. Also darkening the box (with tape or by draping a coat/sweater over it) really helps to keep the birds calm.

**Secure the bird fully before opening the lid to prevent escape.**

Once the bird has defecated, the bird is removed and the entire fecal pellet is transferred with a sterilized infant tongue depressor into a sterile 2 ml Eppendorf tube (screw-top) with 1 ml of 100% ethanol or scraping the pellet off the wax paper with the opening of the tube. For non-solid samples, the sample can be poured into the tube carefully. Pellets should be transferred to the tube within 10 minutes. Both the wax paper and the sterilized tongue depressor should be discarded after being used once, and the container should be wiped clean with a tissue soaked with antiseptic soap solution. **Keep fecal samples cool by placing in a fridge or in a location that is cool – do not place in freezer or cryoshipper.**

**Special consideration for determining samples size and recapture of birds sampled**
To assess inter-annual variation in gut microbiota, we will attempt to recapture birds that were fecal-sampled in 2011 or 2012. Before the field season begins, make a list of all individuals where fecal sampling occurred. Banders can reference this list when a bird is recaptured from a previous year to determine if a fecal sample needs to be taken.

**Materials provided by field camp:**

1. Plastic container with lid – to hold bird while waiting for it to poop. Wrapping the container with duct tape makes it dark and less stressful for the captured bird.
2. Pliable container top – to easily get the bird in and out of the container. A fine mesh type material (e.g. panty hose) wrapped with an elastic band works great.
3. Baking wax paper (1 per bird with large enough pieces to cover bottom of container) – to catch fecal material so that cross-contamination between birds is minimized
4. Chemical heat packs – to put in container to keep bird warm on cold days (put under a bird bag or some other cloth within the container to avoid direct contact with the bird)
5. Antiseptic solution and tissues for cleaning container
6. 100% ethanol –0.3 liter (1 ml per bird) – to preserve fecal material. AK camps will be supplied this item. Canadian camps will need to purchase own supply since the material is hazardous and shipping across the US/Canada border would be difficult.
7. 1.5 ml Eppendorf tube with screw top (1 per bird) – hold feces and ethanol
8. Sterilized tongue depressor (1 per bird) – to transfer fecal material to Eppendorf tube
9. Plastic pipette (5 per camp – can be used repeatedly) – for dispensing ethanol
10. Alcohol-proof pens (4 per camp) – to label tubes containing samples
11. Labels for Eppendorf tubes (1 per bird) – to place on tubes for writing sample information

Table 5. Requested maximum number of fecal samples for 2013 shown per species per ASDN field camp. R represents previously captured and banded individuals. Samples of recaptured birds are especially valuable as multiple samples of the same individual allows us to investigate if and how gut microbiota composition changes between years and if there is a correlation with the age of the individual. Therefore, if sampling effort is in any way limited, priority should be given to sampling of previously captured birds over new birds.

| | | | | | dunl | | | | | | | | | | |
|---|---|---|---|---|---|---|---|---|---|---|---|---|---|---|---|
| Site | amgp | basa | bbpl | bltu | *arct* | *pac* | *hud* | lbdo | pesa | reph | rnph | rutu | sesa | wesa | whim |
| East Bay | | | 10+R | | | | | | | | 10+R | 10+R | 10+R | | |
| Nome | | | | | | 10+R | | | 10+R | | 4+R | | 4+R | R | |
| Cape K. | | | | 10+R | | 3+R | | | | | 10+R | | R | 5+R | |
| Barrow | 10+R | 10+R | | | 10+R | | | 10+R | 10+R | 10+R | 10+R | 10+R | 10+R | 10+R | |
| Ikpikpuk | | | | | R | | | | | 8+R | 10+R | | R | | |
| Colville | | | | | 10+R | | | | 10+R | 10+R | 10+R | 10+R | R | | |
| Canning | | | | | 10+R | | | | 10+R | 10+R | 10+R | | 3+R | | |
| Mackenzie | | | | | | | | | 10+R | | 8+R | | 8+R | | |
| Bylot Island | 9+R | | | | | | | | | | | | | | |
| Churchill | | | | | | | 10+R | | | | | | | | 10+R |
| Igloolik<br>Burnt Point<br>Chaun Delta<br>Lower Khatanga | *If new Network sites are interested in collecting fecal samples for the shorebird gut microbiota side project, please collect 10 samples per species from your most common species.* | | | | | | | | | | | | | | |

## *Blood samples*

If you are conducting fecal sampling that should be done first, then after the fecal sample has been collected, the bird should be removed from the container by reaching through the lid, grabbing the bird, and then removing the lid. The bird can then be transferred to your free hand. Completely process and band the bird (see banding section) before collecting additional samples. Blood samples are collected from the basilic vein (also known as the brachial vein) under the wing of adult shorebirds (Figure 8). To collect blood, first use a small amount of Vaseline or water on a swab to move feathers away from the vein. Make sure there are no loose feathers nearby or else the blood may be soaked up by the feather and will be difficult to collect with the capillary tube. Once the vein is obvious, puncture it with a sterile small gauge (27.5) needle by holding the needle at an angle, bevel side up, and slowly insert into the vein (much like a person takes blood from a human arm). After removing the needle, a small drop of blood should materialize. Draw the blood into a capillary tube (the type of tube depends on the procedure – see below). Hold the capillary tube in such a way that blood flows downward into the tube. In this way, gravity will help you draw the blood into the tube. You may need to pump the wing to get extra blood to flow out of the vessel. Normally the blood will quickly clot, but if it does not, use a piece of a cotton ball to apply direct pressure on the wound to stop it. Do not puncture a vein repeatedly as injuries such as hematomas can occur. If performed correctly, the punctured area will not be visible within a couple of days. It is possible to take blood from both wings if need be. Note the size of the capillary tube – the ones we will provide typically hold 50 microliters (µl) of blood.


Figure 8. Illustration of basilic vein for blood withdrawal and the ulnar nerve. Picture of basilic vein before drawing a blood sample. (Figure: Evers 2008; Photo: S. Sapora/USFWS)

Store blood according to directions for each sample type. For genetics, avian malaria, and for those sites using cryoshippers for the methyl mercury side project; blood will be transferred from hematocrit tubes to other containers. For genetics do the following; given the recent avian influenza issues, use a plastic transfer pipette (Figure 9) to force air through the capillary tubes and move the blood into an Eppendorf vial pre-filled with buffer (see more details below about buffers, etc.). If the blood clots inside the capillary tube (or you have only a very small amount of blood), break off the tube inside the Eppendorf vial and leave it there. Label the outside of the tube with relevant information (see sample labeling section at end of protocol) and place inside nylon insulation bag with refreezable ice pack. Samples should be transported in a cooler during the day. Directions for other blood samples are below.

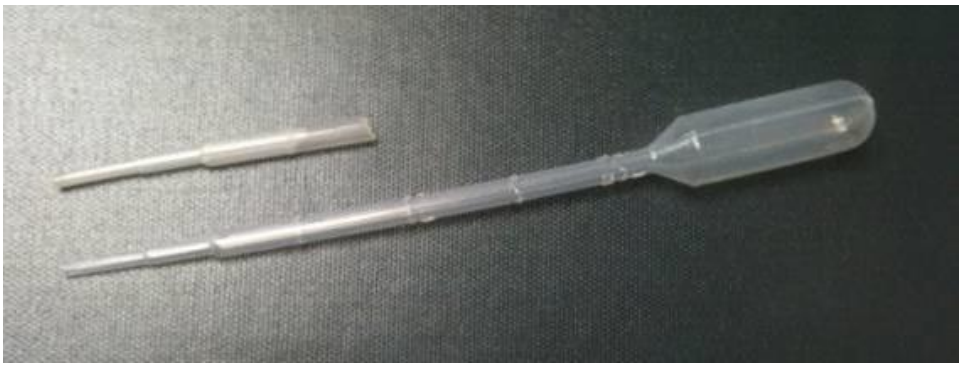


Figure 9. Plastic pipette used to transfer solutions (lower) and preservatives for sampling and modified pipette to transfer blood samples from capillary tubes into storage vials.

**Materials needed, provided by each field camp:**

1. Vaseline or water – to clear feathers from basilic vein
2. Refrigerator/freezer – to refreeze ice packs, store samples in short term
3. Small flexible cooler bags
4. Refreezable ice packs (or some way to have items kept cold while in the field)
5. Cotton balls – to stop bleeding if necessary
6. Small Nalgene with screw cap (200 ml) – for holding used needles and blood waste
7. Tissue preservation buffer – for preserving collected tissues (e.g. chicks or adults)
8. Banding kits
9. Longmire Buffer – 0.3 liter (500 µl per sample) – to preserve WHOLE blood. May be available from Rick Lanctot, request his help if needed.
10. 1.5 & 2.0 ml Eppendorf tube with screw tops (up to 3 per bird, genetic, avian malaria and whole blood samples)
11. Plastic pipettes
12. Transfer samples from capillary tube to Eppendorf tube (5 per camp) – will need to cut off one end of plastic pipette at the right location so it fits tightly over capillary tube, then cut off the large end so you can blow into this section (Figure 9.). Thus you will be able to blow air into capillary tube via this small plastic pipette to transfer blood to Eppendorf tube
13. Transfer bulk samples (10 per camp) – for putting Longmire or other buffers into Eppendorf tubes
14. 27.5 gauge needle (1 per bird + 20 extra) – to puncture vein to draw blood
15. Capillary tubes
16. PLAIN (100 µl size, 3 per bird) – to draw whole blood for avian malaria and genetic sample
17. HEPARINZED (100 µl size, 2 per bird) – to draw whole blood for methyl mercury sample
18. Marking pens (alcohol-proof, 4 per camp) – to label tubes containing samples
19. Critocaps®(4 per bird, plus extras) – to seal methyl mercury capillary tubes
20. 10 cc plastic vacutainers – storage for methyl mercury sample
21. Cyrovials (for camps with cryoshipper) – to store methyl mercury sample
22. Labels for Eppendorf tubes (2 per bird) – to place on tubes for writing sample information
23. Labels for Vacutainers and Cryovials (1 per bird) – to place on tubes for writing sample information
24. Dry Nitrogen cryoshipper (1 per camp for camps without a freezer) – for storage of methyl mercury samples. The shippers stay cold about 65 days.
25. Storage Boxes – (81 samples/box) – enough for storing Eppendorf tubes that do not need to be placed in cryoshipper

**ASDN genetic blood sample**

For basic genetic analysis (population structure, paternity, sex identification), we would like to get 25 µl for our analysis. This typically equates to a half of one capillary tube of blood. Samples for DNA analysis

can then be blown into labeled 1.5 mL plastic Eppendorf tube (screw top preferred) with buffer solution and stored at room temperature. For genetic analysis, it is prudent to save whatever amount of blood you collect, no matter how small. However, **do not take more than the recommended amount of blood** as this can overwhelm the buffer and keep the blood from being preserved. Remember you can now get DNA from the amount of saliva it takes to lick a postage stamp. Be sure to mix the buffer and blood by inverting the tube several times. Place label on Eppendorf vial and fill out relevant information. We advise using Longmire buffer and plain capillary tubes (or EDTA capillary tubes) for collecting these samples.

**Avian malaria blood sample**

Collect 100 µl of blood with PLAIN capillary tubes (blue line around top of capillary tube) and place into an Eppendorf vial filled with 500 µl of Longmire buffer. Place label on vial and fill out relevant information. Samples can be stored at room temperature in a dark space (e.g., inside a storage box) until the end of the field season and then refrigerated at 4$^{o}$C. Efforts should be made to ensure that the temperature is stable at the storage location. Changes in temperature are not good for the samples.

**Methyl mercury blood sample**

For sites participating in this study, the collection method will depend on what type of cold storage your camp has access to. Please refer to Appendix N: Methyl Mercury Sampling and Shipping Protocol 2013, for details about sample collection and shipping. Please be sure to accurately label each sample using labels provided, and cover with tape to prevent degradation of label during storage.

Freezer storage:

Collect two, 1/3-filled 100 µl HEPARINZED capillary tubes (red line around top of capillary tube) of blood. Each capillary tube will be sealed on both ends with Critocaps® and placed in a labeled 10 cc plastic vacutainer to prevent breakage. These blood samples will be stored on ice in a cooler during field collection and then stored in a freezer until shipping.

Cryoshipper storage:

Collect one 1/2-filled 100 µl HEPARINZED capillary tube of blood, which should then be transferred to a cryovial. The cyrovial should be kept cool during field collection, in a cooler with ice packs if possible, and placed in the cryoshipper as soon as possible upon return to camp.

## *Feather Samples*

**ASDN Archive Samples**

Collect reference feathers from all captured birds. Refer to Appendix A for feathers to collect. Place archive samples in a manila envelope, and label with collector name, species, nest ID, band number, date, lat/long location, and general location description or site name (e.g. Prudhoe Bay). ASDN Archive samples should be shipped to Rick Lanctot, USFWS Migratory birds, at address in the shipping section below.

**Methyl Mercury Feather Sample Collection**

Refer to Appendix N for details on collection, handling, and shipping of feathers for sites participating in this study. If you are not participating in the methyl mercury study, follow instructions above for ASDN archive samples. Please make an effort to collect feathers for every bird that you take a methyl mercury blood sample from.

**Feathers for the Moss Dispersal Study**

Sites participating in the Moss Dispersal Study should collect 3-5 feathers from the base of the breastbone for target species. See Appendix Q for details on target species, feather collection, labeling, and shipment for the moss dispersal study.

## *Tissue samples*

Tissues of embryos or chicks that have died should be collected if paternity analyses are desired. Samples from adult birds that have died accidentally can also be preserved. Place a small piece of tissue (pea-size that is minced up) in "tissue preservation buffer" (using a ratio of 1 part sample to 5 to 10 parts tissue preservation buffer). With birds that are freshly dead, it is also possible to get blood from the heart chambers – here we recommend obtaining up to 500 µl of blood and storing it in 1.3 ml Longmire. The blood can be drawn up with capillary tubes as described above. Be sure to sterilize any cutting instruments by dipping them in 100% ethanol and burning between each dissection.

Additional guidelines for using tissue preservation buffer **when preserving tissues**

- Use tissue preservation buffer with fresh tissue only; do not freeze tissues before immersion in tissue preservation buffer
- Before immersion in tissue preservation buffer, cut large tissue samples to ≤0.5 cm (5 mm) in any single dimension.
- Place the fresh tissue in 5–10 volumes of tissue preservation buffer solution.
- Most samples in tissue preservation buffer can be stored at room temp without compromising DNA quality for the duration people will be in the field, up to 1 month at 4°C (fridge door), or at –20°C or –80°C indefinitely.
- ***Important:*** Do not freeze samples in tissue preservation buffer solution immediately; store at 4 °C overnight (outside door on a standard fridge is fine) to allow the solution to thoroughly penetrate the tissue, remove supernatant (top layer of liquid above the tissue), then move to –20°C or –80°C for long-term storage.

**Materials provided by each field team:**

1. Cutting instrument for doing dissection (razor blades or from dissecting kit)
2. 1.5 ml Eppendorf tubes (2 per bird – estimate 10 birds/camp max) – for storing tissue and whole blood samples
3. Plain capillary tubes – take from stock supply mentioned above

## *Field Protocols for SESA Geolocator Deployment*

Questions about the equipment or procedures should be directed to Rick Lanctot (Richard_Lanctot@fws.gov, cell 907.440.9733) or Stephen Yezerinac (yezerinac@gmail.com, cell 506.224.0352). Address questions about participating in the project to Stephen Brown (sbrown@manomet.org, 508-224-6521)

Goal - Capture 30 SESA at each participating site and equip each bird with a geolocator, an engraved flag, and a metal band.

Strategy - Try to use a contiguous, small area for geolocator deployments, as an enlarged area will need to be searched next year to look for returned birds. Try to deploy a logger on both the male and female at each nest. This will provide a roughly balanced sex ratio and possibly yield data on pairs' migrations.

Geolocators - The loggers will be attached to flags, started, and calibrated in Sackville or Anchorage. Keep the loggers away from heat, which will negatively impact the battery life. Also avoid exposing the loggers to vibration and static electricity, which could damage the logger or weaken the attachment to the band. At the end of the field season send any loggers that are not deployed to Rick. They can be used to gather control data.

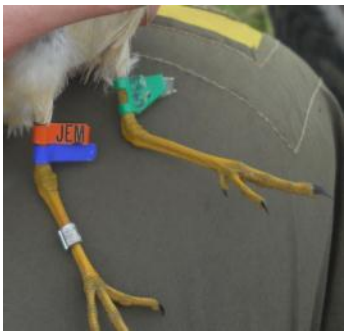
Banding - We want to minimize the added weight for the birds carrying loggers, so use minimal marking:

> Upper Left: dark green logger flag (must be oriented with slanted edge downwards, such that when the band points forwards, the logger faces laterally – see photo below)
> Upper Right: engraved dark green flag (USA) OR engraved white flag (Canada)
> Lower Right: USGS metal band
>
> Example notation for banding records: geo/- : gfe,KLA/m

where geo = geolocator, gfe = green flag engraved, KLA = combination on engraved flag, m = USGS metal band, "/" separates above and below the 'knee', and ":" separates left and right leg. Always write left leg first, and top portion of leg first.

Deployment Procedures - **Before you begin, be certain to note the logger serial number on the banding form, which is written on the flag, so it can be linked later to the band number of the individual. This is very important!!!!**

The logger should be installed early in the banding process, so that the glue has time to set before the bird is released. Place the logger flag on the left leg with the slanted edge downwards, such that when the band points forwards, the logger faces laterally. See the photo below. This positioning is key to proper exposure of the light sensor.




Using a small screwdriver or toothpick as a spatula, apply a small amount of cyanoacrylate glue between the flaps of the flag. Clamp the band shut with the modified clothes pin supplied. Be careful not to place any glue on the light sensor (silver square) or torque the logger-band connection.

If you have a very steady hand, you may choose to also solder the edges of flaps - just those that are furthest from the logger. Only do so if you're absolutely certain you will not touch the protective plastic jacket of the logger with the soldering tip, as doing so would breach the protective plastic casing and lead to failure of the logger.

If you can do so without adversely affecting the individual bird, make a special effort to collect ASDN blood and feather samples. In future years, when you recover geolocators, make a special effort to collect health samples for projects you are participating in (e.g., avian malaria, fecal, and methyl mercury), as they have the potential to elucidate differences among birds related to these health topics, as well as link geolocation, isotopic, and genetic analyses.

Before release, double check that the banding records are complete. The logger serial number from the back of the flag must be recorded in the banding record. **The banding record is the critical link between the logger number and the USGS band number.**

The banding record must contain the date and time the bird was released with the geolocator, so that the light records preceding this time can be ignored when the data are processed. Use local time.

At the end of the season include a note with your data submission that confirms the offset of your local time from GMT. Your GPS unit can provide local time and GMT.

Data from the logger should be included as part of the banding file record submitted to the ASDN science coordinator (Rick Lanctot) or the logistical field coordinator (position not filled at time of writing).

## *Field Protocols for AMGP Geolocator Deployment*

Questions about the equipment or procedures should be directed to Jean-François Lamarre (jflamarre@gmail.com, office 418-723-1986 ext 1909, cell 581-246-8844). **For guidance on collecting and shipping AMGP geolocators that you recover this year, see Appendix P**.

Goal - Capture American Golden-Plovers at each participating site and equip each bird with a geolocator, a metal band, and a unique banding combination of bands (either color band and/or engraved flag).

Strategy - Try to use a contiguous, small area for geolocator deployments, as an enlarged area will need to be searched next year to look for returned birds. Try to deploy loggers mostly on males as they tend to be more faithful to their breeding site than females (better chances of coming back next year). In case of doubt about the bird's sex, you may mark both pairs on a nest. Avoid trapping in the first week of incubation to reduce risk of abandonment.

Geolocators - The loggers will be assembled, started, and calibrated in Rimouski (Québec, Canada). Keep the loggers away from heat and variation of light, which will negatively impact the battery life and the memory life. Keep the loggers in the dark until deployment. Also avoid exposing the loggers to vibration and static electricity, which could damage the logger or weaken the attachment to the band. At the end of the field season send any loggers that are not deployed to JF. They can be used to gather control data.

Banding - We want to be able to ID an individual with a combination of bands or engraved flags. We encourage each team to use distinct combinations specific to historic plover marking. It is essential to make sure that the combination used will still be unique once the geolocator is removed in a subsequent year.

Example of band combinations:

| Upper Left | Lower Left | Upper Right | Lower Right |
|---|---|---|---|
| geo,- | wf,dg | m,- | r,r |
| geo,- | wf,dg | m,- | r,y |
| geo,- | wf,dg | m,- | r,o |
| geo,- | wf,dg | m,- | r,dg |
| geo,- | wf,dg | m,- | r,db |
| geo,- | wf,dg | m,- | r,lg |

**AMGP Geolocator band location:** An American golden plover with a unique combination of bands. Note the spacer-ring bellow the geolocator. The logger flag is a previous model of attachment.

Example notation for banding records: geo,-/ wf,dg : m,-/ r,r
"/" separates above and below the 'knee', and ":" separates left and right leg. Always write left leg first, and top portion of leg first.
geo= geolocators and spacer band under it , wf= white flag-for Canada (would be dark green in USA)
other colors are: r=red, y=yellow, o=orange, db=dark blue, lg= light green, dg=dark green; m is for metal.

Deployment Procedures – **Before you begin, be certain to note the logger serial number on the banding form, which is written on the flag, so it can be linked later to the band number of the individual.** Check your band combination and select a combination with a geolocator. Mark it immediately as used and indicate the USFWS metal band number next to it (and any additional information). The logger should be installed early in the banding process, so that the glue has time to set before the bird is released. If using a spacer, place the spacer ring on the leg first so it will be below the logger flag (installation described below, see figure at left). Do this by unrolling the roll-up type band and gently reroll it on the bird's leg. Then you can roll it between your fingers in order to give back to proper diameter so it is the correct size for the birds. The band has a dark spot indicating from where you started. Roll the band until the edge of the roll touches the dot as it was before you unrolled it. Use the soldering pen to melt shut the roll-up band. Avoid making any sharp end that would hurt the bird's leg.


**AMGP Geolocator Modal:** Example of a band combination using an engraved flag. The geolocator flag shown is what the geolocators you receive will look like.

Place the logger flag on the left leg, with a spoon for plastic bands or with your fingers and slide the logger in place. Put the slanted edge downwards, such that when the band points forwards, the logger faces laterally. This positioning is key to proper exposure of the light sensor. If you have to put it on the right leg, the logger should also face out when pointing forward.
The logger flag should have sprung back in position (or almost). Using a small screwdriver or toothpick as a spatula, apply a small amount of crazy glue containing cyanoacrylate (gel type is less messy) between the flaps of the flag. Clamp the band shut with the modified clothes pin supplied. Be careful not to place any glue on the light sensor

(silver square) or torque the logger-band connection. Beware of putting any glue on your finger to avoid being glued to the bird.
If you have a very steady hand, you may choose to also solder the edges of flaps - just those that are furthest from the logger. Only do so if you're absolutely certain you will not touch the protective plastic jacket of the logger with the soldering tip, as doing so would breach the protective plastic casing and lead to failure of the logger.

If you can do so without adversely affecting the individual bird, make a special effort to collect health samples (e.g., avian malaria, fecal, and methyl mercury), blood and feathers from birds given loggers, as they have the potential to better understand differences among birds related to these health topics, as well as link geolocation, isotopic, and genetic analyses. This is particularly important in the year the loggers are recovered since we can link migration location to all of these factors.
Before release, double check that the banding records are complete. The logger serial number from the back of the flag should be recorded. The banding record is the critical link between the logger number and the USGS band number.

The banding record must contain the date and time the bird was released with the geolocator, so that the data preceding this time can be ignored when the data are processed. Use local time. At the end of the season include a note with your data submission that confirms the offset of your local time from GMT. Your GPS unit can provide local time and GMT. Data from the logger should be included as part of the banding file record submitted to the ASDN science coordinator (Rick Lanctot) or the logistical field coordinator (position not filled at time of writing).

## NEST MONITORING METHODS

### *Objective*

There are two approaches for locating and monitoring nests that are available to project leaders – please note clearly in your meta-database which approach you took. The Minimum Nest Search protocol includes searching a general area for nests, and then capturing adults at those nests and monitoring those nests for survival. The Intensive Nest Search protocol includes 1) designated intensive plots where nests are located in a standardized way but no adults are captured, and 2) a larger search area, typically surrounding these intensive plots, where nests are located and monitored, and adults are captured. The latter design will allow us to document nest density and species diversity on the plot, and also test whether banding of adults decreases nest survival. Study sites wishing to pursue the Minimum Nest Search protocol will have more flexibility in their nest searching efforts but will record the same information at nest sites and will visit nests at the same interval for monitoring nest survival.

Note: Much of the information below is specifically geared toward the intensive plot but many of the methods can be employed in the Minimum Nest Search protocol and on the larger search area portion of the Intensive Nest Search protocol.

### *Intensive plot protocol*

We will rely on a combination of intensive area search and rope-drag techniques to document birds breeding on intensive plots. Area searchers and rope-drag crews will record their data on individual plot maps each day, and then at the end of the season, data will be combined across all workers to create a

final nest and probable nest map (see more details below). The goal is to get the most accurate description of the species that nest on each plot (including nests found and probable nest locations) by documenting the presence of territorial birds (whose nests are not found) and finding nests during the field season. A second goal is to document nest survival. This will be accomplished by monitoring each nest on a regular basis until the nest hatches or fails. Attention to detail, communication with your co-workers, and accurate and timely recording of data is imperative to the success of each intensive plot. To be able to accurately compare how a site varies across years, and how sites vary among one another, **it is essential that standardized methods be used at each site.**

## *Intensive plot size and shape*

Plots should be located within a given study site in habitats that will likely produce the highest densities of the focal species. If possible, place the plots randomly within these high quality habitats (i.e., stratified random placement). If these random plots fall in poor nesting habitat, then move the plot to a location that does have good numbers of birds (i.e., it will no longer be randomly placed). How plots are ultimately located will affect the extent to which your data can be extrapolated – thus be sure to indicate how this was done in the meta-database that accompanies the study site. Plot size should be at least 10 ha in size but can be much larger in low density areas. Plots can be irregularly shaped (following contours of natural landscape features) or more geometrical in shape. Square plots are preferred as they have less edge per surface area and thus are less likely to have birds establishing territories along the borders of the plot. If density allows, we recommend establishing 16-ha (400-m x 400-m) plots, and marking the boundaries and interior portions with survey stakes placed at 50-m intervals, thereby subdividing the plot into 64 50-m x 50-m cells (Figure below). The 400 $m^2$ plot size was established to be consistent with Arctic PRISM intensive plots. The stakes will be labeled from A1 to A9 (west to east) and A1 to I1 (north to south). Researchers may alter the size or shape of the nest survival plots to conform to obvious geographic boundaries or other study objectives if necessary. The grid stakes are useful for ensuring complete coverage of the plot during nest searching, in addition to acting as landmarks for recording and relocating nest locations. All study plots must be marked with stakes that will be adequate to maintain and relocate the plot for 5 years. Sites where establishing a grid system is not helpful should at a minimum establish an outer plot boundary and midpoints (e.g. A1, A5, A9- west to east northern boundary; I1, I5, I9 -west to east southern boundary and north-south midpoints (Figure below). Stakes should be labeled in a systematic fashion and can be labeled with identifying numbers and letters; the GPS location should also be collected and saved on the plot description data sheet and entered into the meta-database. We recommend that stakes be inserted in the frozen tundra by first making a hole with a pointed metal frost spike (*US Customary*: 18” x 1.5” x 3/4” or *Metric*: 45cm x 3.8 cm x 2 cm) and mallet (*US Customary*: 3 lb or *Metric*: 6.5 kg.). A wooden surveyor’s stake (*US Customary:* 36” x 1.5” x 3/8” or *Metric*: 90 cm x 3.8 cm x 1 cm) can then be tapped into the hole. The stakes can be pushed into the tundra as the thaw depth increases during the summer and pushed in as deep as possible at the end of the season to last the winter. You may want to set up a minimum number of stakes when the ground is frozen, and complete the stake installation at the end of the first breeding season when the ground is thawed. We also recommend painting the top of the stakes with a light color house paint and then painting large letters and numbers (observable from at least 50 meters) with house paint (do not use markers – the letters wear off in the wind and must be remarked each year). Stakes can also be oriented in different directions (i.e., flat side of stake facing north and then east to maximize your ability to read the stake letter/numbers from different areas of the plot). Ideally, you should be able to read a stake from any location within the 50 x 50 m cell within the plot.

| | | | | | | | | |
|---|---|---|---|---|---|---|---|---|
| **A** 1 | A 2 | **A** 3 | A 4 | **A** 5 | A 6 | **A** 7 | A 8 | **A** 9 |
| **B** 1 | B 2 | **B** 3 | B 4 | **B** 5 | B 6 | **B** 7 | B 8 | **B** 9 |
| **C** 1 | C 2 | **C** 3 | C 4 | **C** 5 | C 6 | **C** 7 | C 8 | **C** 9 |
| **D** 1 | D 2 | **D** 3 | D 4 | **D** 5 | D 6 | **D** 7 | D 8 | **D** 9 |
| **E** 1 | E 2 | **E** 3 | E 4 | **E** 5 | E 6 | **E** 7 | E 8 | **E** 9 |
| **F** 1 | F 2 | **F** 3 | F 4 | **F** 5 | F 6 | **F** 7 | F 8 | **F** 9 |
| **G** 1 | G 2 | **G** 3 | G 4 | **G** 5 | G 6 | **G** 7 | G 8 | **G** 9 |
| **H** 1 | H 2 | **H** 3 | H 4 | **H** 5 | H 6 | **H** 7 | H 8 | **H** 9 |
| **I** 1 | I 2 | **I** 3 | I 4 | **I** 5 | I 6 | **I** 7 | I 8 | **I** 9 |

## *Nest searching methods and general techniques*

See data forms *nest record*

Although our primary focus is to find the nests of all shorebirds, it is important to document the presence of other nesting species, especially predators and other waterbirds. We do not plan to search for or monitor Lapland Longspur nests as part of the Network. Nests of all shorebirds, avian predators and waterbirds should be marked on plot maps (*see dataform-daily plot spot maps.doc*) and receive a nest number and filled out plot form. We do not plan to monitor nest survival of avian predators in an effort to minimize local disturbance.

Nest searching can be conducted in all types of weather, except perhaps in extreme cold (~25 °F or -4 °C) and rain which might cause egg cooling when incubating birds are disturbed. We don't recommend nest searching in winds >60km/h to avoid disturbing the birds and it is much less efficient due to changes in behaviors. On cold days, birds appear to flush closer to the observer, which increases the detectability of nests for species that typically flush at great distances (e.g. AMGP).

During the first visit to a plot, it is useful to walk through the plot finding as many nests as possible and noting the location of these nests and other territorial birds that likely have nests on a daily plot map. Since nests found in this way are typically the easiest to find, this is referred to as 'high-grading'. By the end of the first full-day equivalent, all territories will have been plotted and nest searchers will have a good estimate of the number of birds present (although more pairs may move into an area as the snow melts). On subsequent visits, the goal is to find nests for each territory holder on the plot, including those pairs on the edges of the plot; sort out the number of territory owners, especially in cases where there are numerous unmarked pairs of the same species nesting near each other; and then search intensively for territories and nests that were not part of the first estimate. It is particularly important to find the nests of birds holding territories near the edges of plots so they can be conclusively ruled in

or out of the plot. This information will be useful for estimating an accurate plot nest density that can be compared across years and study sites. In this process, nests found off plots will be useful for capturing adults to estimate adult survival. It is not a waste of time to spend hours looking for a nest that is eventually found off the plot.

It is advised that territorial birds thought to have probable nests be revisited on subsequent days, and that all portions of the plots are visited regularly (i.e., do not focus solely on the area nearest your approach location). Prior experience indicates it is better to visit each plot daily, rather than wait 3-4 days between nest visits; many nests can be depredated during this interval. One way to do this is to have two intensive plots be located relatively close to each other (i.e., have intensive plots be "paired") and then have one observer visit one plot in the morning and the second plot in the afternoon. The next day a different observer can visit these plots but in the opposite order. Because observers vary in nest searching skills, all observers should rotate between all plots. Search as much of the plot on each day as possible. If a nest search on a given plot cannot be completed in one visit, indicate where you stopped on your daily nest searching map, so that the 2$^{nd}$ nest searcher can focus on the area that was not searched.

It is common to find nests of other species within 1-2 m of a known nest, and Semipalmated Sandpipers sometimes nest within 1 m of a conspecific. Similarly, it may sometimes be necessary to flush a bird from a known nest to determine whether a nearby bird is associated with the known nest or is a member of a separate pair. It is also important to search for shorebird nests near the nests of charismatic species such as Tundra Swans, geese, and loons; the increased risk of predation to these species is an unfortunate necessity of obtaining an accurate estimate of shorebird density on a nest survival plot. Efforts should be made to avoid Spectacled Eider (*Somateria fischeri*) and Stellar's Eider (*Polysticta stelleri*) nests as they are protected species under the Endangered Species Act.

## *Intensive-area searches*

The intensive area search method involves a single person (e.g. nest searcher) who walks throughout the plot, using the presence and behavior of birds to determine the location of territories and nests. Because each nest searcher has different capabilities, we will have nest searchers rotate between plots to ensure consistency amongst plots in the number of nests found. To ensure complete coverage of each plot, it is recommended that nest searchers cover the plot by walking between stakes in a "W" pattern. This will reduce observer bias and increase the probability of all nests being found. However, nest searchers should not focus too hard on performing their "W" walk at the expense of missing birds that are flushing from nests in front of them. Unique detailed maps of each plot should be created during plot set up, noting any landforms (e.g. lakes and creeks), notable features or human debris. This grid structure can be copied onto daily search maps and notes can be gathered daily to create a plot master map at the end of the season. The territorial birds or nests discovered should be recorded on these maps. These maps should be shared among the nest searchers so that information on potential nest sites are shared daily and subsequent visits to a plot can be planned to enhance finding nests.

## *Plotting nests and territories during area searches*

The following approach is one way to maximize communication among nest searchers as they visit the various intensive plots to look for nests. Each camp may use a different technique as long as their nest searchers can reliably document both nests and territories on the intensive plots at the end of the field season. Each plot will have a **nest plot book** that has a plot map that illustrates known nests. Area

search and rope-drag crews will record nests in this book, but also record the location of territorial pairs on a separate map for each day they visit the plot (see Figure 10, top panel). These maps will be shared among crew members to maximize the chance of finding nests before they are depredated. In the windy and misty arctic environment, we find it best to place maps in a standard sheet protector, anchor it to a clip board with rubber bands, and write on the sheet protector with an ultra-fine tipped Sharpie (alcohol will remove ink for re-use of sheet protector). **After each day in the field, transfer new nests into the nest plot book for their respective plot.** Also make a field copy of the permanent map and use it within a sheet protector for the next visit to the plot. Update both the field and camp copies after each visit. We use only black Sharpie on the sheet protector because other colors tend to rub or wash off easily.

The term "territory" is used in a broad sense to mean the area of primary use. For example, the area of use of a Semipalmated Sandpiper pair that is staying close to a nest may be drawn as a very small circle. The area of use of a Black-bellied Plover pair whose nest has not been found and who travels widely on and off the plot, may be indicated by a series of arrows denoting movements rather than by a well-bounded circle.

Probable nests are defined as a location from which an individual flushes using a broken-wing, rodent-run or other distraction display but the nest was not located. The location must be precise enough for the observer to be certain that the actual nest is on the plot. No other definitions of probable nests are acceptable. Probable nests should be mapped on the daily plot map (not Nest Book) and mentioned to the subsequent person visiting the plot so they can look for the nest on the proceeding day. Probable nests ARE NOT given a nest number and will not have a nest form filled out until found.

At the end of the field season, time should be reserved so that nest searchers can make a final map for each plot depicting the nest sites and territories of birds whose nests were not found (Figure 10, bottom panel). The data sheets from the field visits can be useful here as it allows nest searchers to collate everyone's data together. For example, if a pair of plovers was consistently found in one part of the plot but a nest was never located, it is likely a pair initiated a nest but that nest failed before discovery.

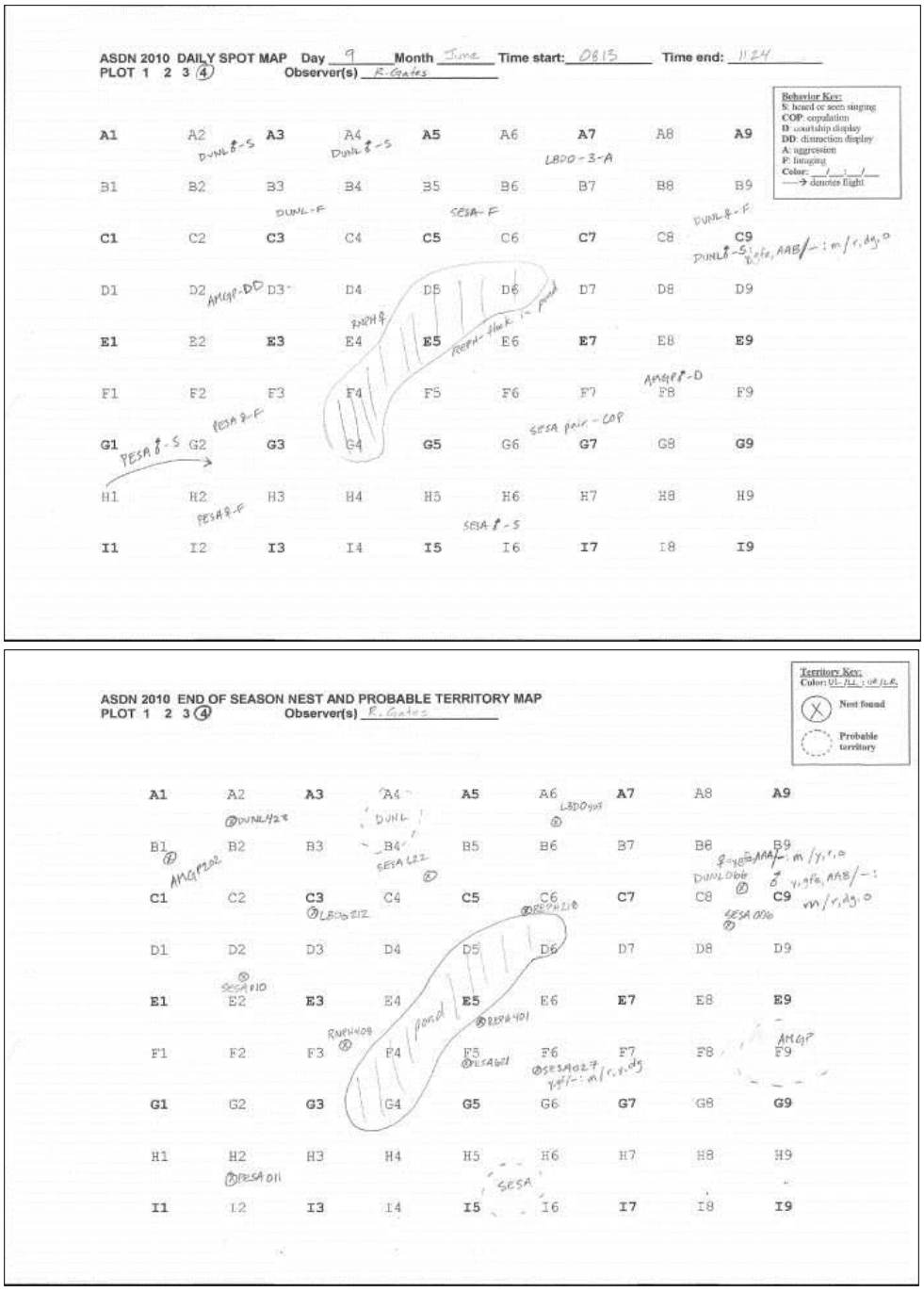



Figure 10: Example of a Daily Spot Map for taking behavioral notes while conducting intensive nest searches (top panel) and the End of the Season Nest and Probable Territory Map (bottom panel).

## *Rope-dragging*

Rope-dragging is a commonly used tool for locating the nests of ground-nesting birds. In this study, 2 person teams will rope-drag one to two times during mid to late-incubation on the intensive plots, and if time allows on the larger search areas. Rope-dragging should begin approximately 14-16 days after the first clutches are complete. Rope-drags on each plot are scheduled 4-6 days apart, but the decision to

rope drag a second time will depend on the species present in your area (i.e., if you have a lot of secretive species that only flush off a nest when nearly stepped on), the success in nest finding using the area search method, and the time available to do it.

Because dragging a simple rope across terrain with even mild topographic or vegetation height heterogeneity can be problematic (e.g., ropes gets caught on obstructions, freeing ropes from obstructions can result in some terrain not actually dragged, etc.), rope drags were designed to have hanging plastic strips or "dropper lines" placed every meter along the rope. These strips will contact the substrate, while the rope itself is suspended slightly off the ground by the rope-dragging crew. The drag consists of a main line of 1/4 - 3/8" rope, with dropper lines (1/4" rope) tied to it perpendicularly at 1.5m intervals (Figure 11). Improvised handles (and/or waist belts) should be tied to the rope so that observers can maintain tension on it and keep it from snagging on irregularities in the ground. In cases where the ground is very flat, a longer rope without dropper lines may be used. The time it takes to rope drag a plot varies dramatically with terrain and bird densities, and some plots may take much longer; it is advisable to begin rope-dragging in the morning when possible. Remember the goal is to find nests, not complete the rope-dragging of a plot. If you are finding nests, you are being successful.

Although rope-dragging will occasionally cause a bird to flush from its nests directly under the rope, some birds will flush well ahead of the rope and the approaching crew. If a quick inspection of the flushing area fails to locate the nest, the crew should continue past the nest a sufficient distance to allow the bird to return (this will vary by species, weather, individual, etc.), then stop and monitor the bird's behavior until it returns to the nest. If the rope-dragging crew fails to find the nest of individuals or pairs that are obviously "nesty", and the crew is confident that the nest is on the plot, they should record it as a probable nest and estimate the probable nest location as accurately as possible. The rope drag team can revisit this spot later in the day, or at the very least, other nest searchers can go back to this spot on a subsequent day.

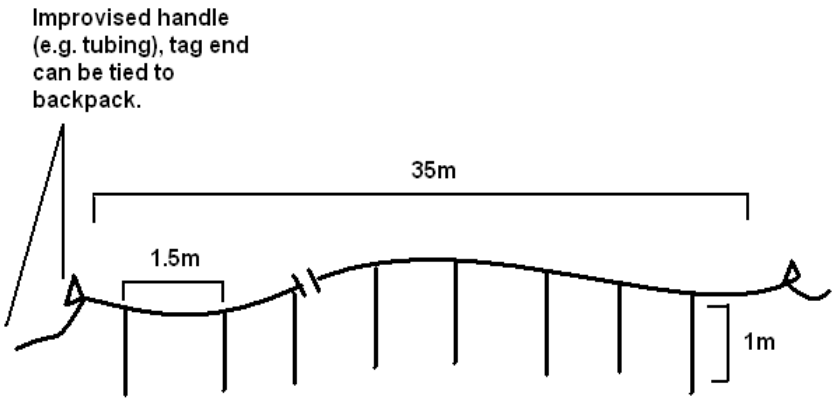



Figure 11. Construction of rope drag for intensive shorebird monitoring plots (Figure: Smith et al 2008)

## *Special techniques for finding nests*

While the area search and rope drag are general approaches to finding nests, these approaches can be enhanced by employing special techniques and using our general knowledge of the nest habitat preferences and general behavior of each species. Combinations of several nest-finding strategies are

necessary to find all nests on a plot. Below are suggestions for becoming proficient at different techniques.

## *Systematic searches*

During the beginning of the field season when snow covers most areas of the tundra, one-egg nests can sometimes be found by simply walking to all the available open spots and looking for nests (i.e., do not rely on bird behavior but simply search the tundra with your eyes). Occasionally 1 or 2 egg nests can be found this way, and these nests are generally the first laid nests of the year. Systematic searching can also be used later in the season when you have reduced the probable nest to a small area; the nest searcher can spend time thoroughly covering the area to find the nest. Nest searchers must be careful when using this technique to avoid stepping on the nest you are trying to find. Looking closely at each place you plan to step before taking a step will insure that you do not step on the nest you are trying to find.

## *Behavioral clues*

When watching adults, nests may be found by following incubating birds back to the nest or by pinpointing the location from which a bird flushes. The following is a summary of behavioral clues:

- Pick a territory or pair whose nest has not been found (look at daily plot maps), walk the known area of use until you find a nest or detect a bird which can be followed back to a nest.
- Any suspicious bird, or bird that does not appear to be associated with a known nest, is worthy of an extended observation. In dimorphic species in which only one sex incubates, like Pectoral Sandpipers where only the female incubates, it is easier to decide which birds are worth watching (i.e., follow the incubating sex, although the non-incubating sex may display near an incubating bird). In monomorphic species (e.g. Semipalmated Sandpipers) or those with dual incubation (e.g. Dunlin), it is more difficult to decide if a bird should be watched or not. Incubating birds sometimes appear disheveled and breast-preening is a classic tell-tale sign of a recently incubating bird. Birds that are incubating have various displacement behaviors that indicate that they are stressed and nervous and would like to return to the nest. When you happen upon a bird that is preening, nervous, or foraging really fast, it is likely that they are on an incubation break or you flushed them off a nest; back off the bird and allow it to return to the nest. In general, males will return to nests quicker than females – if you can determine the sex based on plumage, vocalizations, or behavior, this can help you determine whether a bird should be followed.
- Walk preferred nesting habitats of key species (e.g., pond edges or marshes for phalaropes; dryas benches for American Golden or Black-bellied Plovers).
- Nests of some species are more easily found by observing individuals at a distance before they are disturbed; this works well for Bar-tailed Godwits, American Golden, and Black-bellied Plovers. In these cases, try to take advantage of a higher elevated site where you can see the bird from a long distance. Using a spotting scope can also help (e.g., the black and white stripe pattern of an AMGP is very distinctive from a distance). Do not get too close or else the bird will not return to the nest.
- Use your binoculars creatively – from a bird's perspective they see a tall predator-like thing staring at them with huge eyes (e.g. your binoculars). Sometimes you can get down low, making the bird feel less watched and point your binoculars away from where you are looking, and then watch the bird return to the nest with unaided eyes. You can also cover your binoculars partially with your fingers, thereby breaking up the "huge eyes". Keeping your eyes on the incubating bird; bring your binoculars to your eyes to see exactly where the bird is. Remember to look for landmarks in the area to help you find the nest as you approach it from afar…keep your eye on the nest. Don't allow

yourself to get distracted by other birds in the area or the nesting bird as it tries to distract you away from the nest.

**Appendix B** has a species-specific guide to shorebird behavior that helps nest searchers interpret the behaviors they are observing and streamline their time to find nests. Shorebirds have diverse mating systems, incubation strategies and suites of sex-specific behaviors and vocalizations. Taking the time to understand the differences in species and sexes will save time in finding nests.

## *Nest monitoring*

Once nests are located, institute a **5-day visitation** schedule so we can obtain accurate estimates of nest survival. **Do not skip nest checks** – a delay in scheduled nest check reduces the precision of our daily nest survival estimates. Curiosity leads observers to visit nests more often than necessary; however, this likely has a negative impact on the nest's success. Nests should be visited 4 days prior to expected hatch (even if it doesn't fall on the 5$^{th}$ day of visiting). If there are no signs of starring, then the nest can be visited 2 days later and checked for hatch again. Continue this process until you see the first signs of chicks trying to exit the egg. Starred eggs typically hatch 2 days later (but can hatch the next day), and pipped eggs may hatch later that same day or early the next day (Figure 12). Continue to visit nests daily at this point until you have documented whether all four chicks have hatched. For nests where eggs do not hatch, collect the egg and determine whether an embryo was developing and died, or whether no embryo was present. These data on egg hatchability should be recorded on the nest record form too (frequently overlooked!).

*We recommend a 5 day visitation rate for all nests found, whether on the intensive plots or larger general search area.*

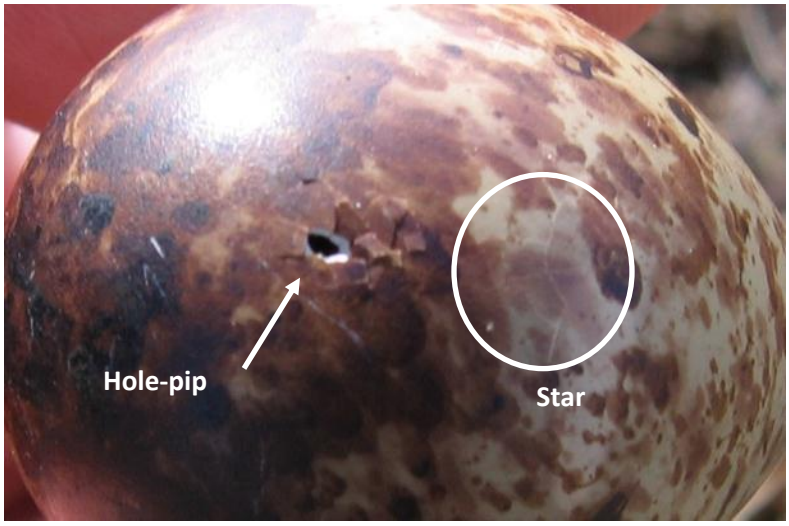



Figure 12. A Dunlin egg that has stars surrounding a "hole-pipped". Photo: D. Taylor/USFWS

**<u>Data recorded at each nest</u>**

The following data should be recorded at all nests when discovered (*see Nest Record data form*):

1. Nest identification number (Nest ID#):
2. Observer(s): (first name initial and last name e.g. loring, for Lewis Oring)
3. Plot and subunit if appropriate (e.g. unique subunit id, quadrat number or northwest stake)
4. Date and time of nest discovery (use military time)

5. GPS location of nest
6. Nest within plot boundaries (Yes or No, may need to measure if near border)
7. Estimated hatch/fledge date: calculated from age data
8. Species: (4-letter AOU code)
    a. 4 letter species code + number of nest found
9. Method of discovery (rope drag, area search, bander, other, explain observer first initial and last name)
10. Color band combos of pair, specific sex if known
11. Nest site map: a simple map of the nest site; include nearby distinguishing physical features (e.g., ponds, polygon rims, etc.).
12. Number of eggs/nestlings on discovery day
13. Flotation data: see **Appendix C** for specifics
14. Measure egg length and width to the nearest 0.01mm with dial calipers

Back at the office, estimate important nest dates such as initiation date, start of incubation, and estimated hatch date, and add to nest form.

**Nest monitoring data to record:**

15. Date (dd-month)
16. Time (military time)
17. Observer initials
18. Nest stage (laying [L], incubation [I], hatch [H], brood [B], predation [P], fail [F], abandon [A])
19. Nest contents/adult seen (Y/N)
20. Number of eggs[E] and/or number of chicks [C] if contents seen, otherwise N/A
21. Pip [P]/ star[star symbols with #of stars]/ crack [C]- dash for none observed
22. Done this visit: Flag, float, measure eggs =FFM, Nest check=N "with a check symbol" Hatch check H “with a check symbol” B=Band
23. Next visit date
24. MID-INCUBATION Measure: Percent nest concealment: use ocular tube and estimate to the nearest 10%

**After nest fate has occurred, record:**

25. Dominant vegetation and landform type. **Appendix G** has categories appropriate for the Western Alaska and the Arctic coastal plain. For other parts of the Arctic use the most reputable guide that lists dominant vegetation and landform.
26. Nest Fate determination (see procedures below):
    a. Date nest fate was determined (day and month)
    b. Was the nest scrape: Intact/scattered/Flattened and widened
    c. Fox urine smell (Yes or No), Fox scat present at nest site (Yes or No)
    d. Were egg fragments present (Yes or No), if yes describe number and location
    e. Were egg shells present (Yes or No), if yes, describe number and location
    f. Membranes: Attached/Separate/None
    g. Weather induced: (Y/N)
    h. Caribou trampling (Y/N)
    i. Adult present eliciting brooding behavior (Y/N)
    j. Was the brood seen (Y/N), if yes # of young, age, and distance from nest
    k. Nest fate (hatch/fledge, fail, unknown, undetermined), take note of dead chicks in chick database

l. For nests that fail, indicate how: predation, weather, trampling, human cause, other, etc.)
m. Write detailed notes on justification of nest fate determination (**be meticulous!**)

## *Marking nests*

Marking nests in a standard way allows all co-workers to find nests more quickly which minimizes disturbance and the potential attraction of predators. Markers (and marker placement) should minimize olfactory cues for nest predators. We recommend marking nests with two Popsicle sticks (i.e., large tongue depressor) and one pin flag. All nests should have one popsicle stick placed 1 meter north of the nest, a second popsicle 5 meters north of the nest and a green or blue (not red, orange or yellow) colored flag 10 meters to the north of the nest (see Fig.13). North can be determined by using the grid stakes (e.g., walking from I1 to A1 would be in a north direction) assuming the plot was laid out so A1 was in the northwest corner, or by using a compass. The flag and Popsicle sticks must create a direct line to the nest (i.e., so you can walk south from the flag and find the nest). A medium-point Sharpie can be used to write the year the nest was found, species, and nest number on the Popsicle stick closest to the nest and on the flag itself (e.g. SESA401). Nest locations MUST be plotted on the map in the nest booklet at the time they are found (see item 11 on nest record form). Use landmarks, number of paces, and cardinal directions to guide the observer from one landmark to another, then to the nest marker and then to the nest. Keep the distance between landmarks short, especially if landmarks are small, and use a compass (don't guess) to get directions from nearby stakes to the nest site. A crude drawing indicating the landmarks (e.g. ponds, creeks, hummocks), number of paces and direction between landmarks, the nest marker, and the nest is very useful.

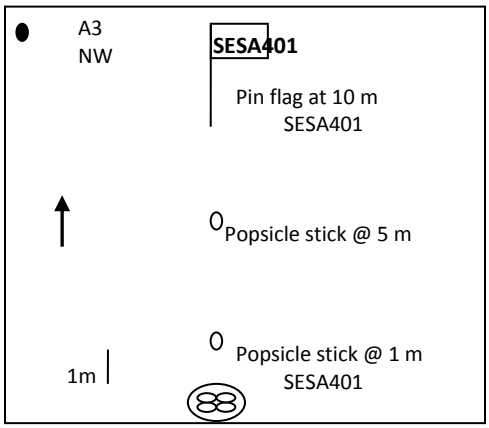



Figure 13: Schematic of marking nest to relocate for nest monitoring

## *Numbering nests*

Each observer numbers their nests consecutively regardless of species or plot. The most common error is to use a number more than once. Use a numbering system for identifying each nest that avoids such errors. It is advisable for each observer to keep a list in their field notebook of nest number, species, plot, and day on which it was found. On maps and in notes, refer to nests by species and number (e.g., SESA401). Field supervisors should assign each observer a series of numbers for use throughout the field season.

## *Recording locations of nests on maps*

Record the location of each nest in a GPS unit as well as on the nest record data form. Be sure the GPS location is recorded on the nest form prior to leaving the general location of the nest. Do not plan on copying GPS locations from your GPS unit to the nest form at night – this is frequently forgotten and the data are lost and the nest needs to be visited again to retrieve these data.

## *Nest initiation date determination*

Nest initiation date should be determined for every nest. Nest initiation day is defined as the day when the first egg was laid in the nest. We define the start of nest incubation as the day when the fourth egg is laid in the nest. This date is also commonly known as the clutch completion date. This day is defined as "day 0" when using nest incubation periods to determine estimated hatch date (i.e., the day when the 4$^{th}$ egg was laid is day 0, the next day is the 1$^{st}$ day of incubation, etc.). A reliable estimate of nest age is important for two main reasons: 1. It assists in accurately assessing nest fate by allowing a calculation of the estimated hatch date and, 2. It provides a way to correct estimates of density for non-detection associated with early nest loss.

**Use the following procedures to determine nest initiation and hatch dates:**

1. For nests found in the laying stage (incomplete clutch) count 1 day backward for each egg laid to estimate initiation date. You can forward calculate (using the estimated incubation/nestling stages for the respective species – see **Appendix D**) to estimate the hatch date. Shorebird nests found with less than four eggs, the average shorebird clutch size, should be revisited the next day, and every subsequent day until clutch size stays the same. Daily visits to an incomplete clutch will determine if the female is still laying eggs versus partial nest predation. This is particularly critical late in the season when 3-egg nests may be more common. You cannot assume a 3-egg nest will have a fourth egg – this has to be verified. Three egg nests found during late incubation should be floated upon discovery to determine nest initiation date.

2. Age of eggs for shorebirds nests found during incubation (i.e., with 4 eggs) should be estimated using the egg flotation technique. See **Appendix C** for complete instruction on egg flotation procedures. A simple float program and corresponding table (**Appendix J**) has been developed to aid in standardizing initiation dates based on egg floatation. Resources for estimating egg age and calculating estimated initiation, incubation and hatch dates can be found on the Manomet ASDN Webpage http://www.manomet.org/arctic-shorebird-demographics-network.

3. If nests are found with star cracks or pip holes in the eggs, or hatchlings (i.e., via nest monitoring near the expected hatch date), the initiation date of the nest can be determined by subtracting the number of days of a typical incubation period for that species from the calculated hatch date (see **Appendix D**; unless this nest was found during laying in which case the laying information should be used). For our purposes, hatch date will be the day the first chick is found in the nest (even though chicks may continue hatching for an additional 1 or 2 days sometimes). If for some reason you do not witness chicks hatching, use the following rules: 1) star-cracked eggs, assume hatch day is in two days (for shorebirds, one day for waterfowl), 2) pipped eggs, assume they will hatch the next day, 3) wet chicks, assume they hatched 4 hours prior to your visit – if your visit is at 4:00 AM, then

the chicks likely hatched the prior day. If at all possible, visit nests to confirm hatching of chicks (see chick banding section).

## *Nest fate determination*

Determination of nest fate will be most accurate when nests of known age are visited at more frequent intervals as expected hatching approaches. Put as much effort as possible into determining the fate of each nest. Generally, **2 pieces of evidence** are needed to classify a definitive nest fate. If nest fate is determined, record supporting evidence on the data form and how confident you are in this assessment. If there are doubts, the nest fate should be recorded as "unknown". Nests with unknown fate can still be used in the survival analysis (until the check prior to that where fate became unknown), but they contribute significantly less information than nests with known fate. See **Appendix E** for more information on finding eggshell remains and using them as evidence of nest fate for shorebirds. See **Appendix F** for fate determination definitions. Also see **Appendix R** for helpful scenarios describing nest and egg fates, and choice of categories to report in each case.

## *Nesting habitat classification protocol*

Basic habitat information should be collected at each nest that describes the macro and micro site conditions, or landform and vegetation type, respectively. **Nest concealment measures should be taken during mid-incubation for each nest that is still active**. **At the final visit to a nest (generally after hatch or when found depredated),** record the landform and vegetation types according to the habitat classification for your location (**Appendix G**).

1. Landform type: Record the dominant landform type within a 10 m diameter of where the nest occurs. Landform types for the North Slope of Alaska can be found in the Geobotanical Atlas of the Prudhoe Bay Region, Alaska (Walker et al. 1980). See **Appendix G** for landform descriptions. These landforms are large-scale, geophysical features that may contain a variety of vegetation types. For other areas, use the most reputable guide available.
2. Vegetation type: Record the dominant vegetation type within a 10 m diameter of where the nest occurs. See **Appendix G** for vegetation descriptions. For other areas, use the most reputable guide available. Develop standardized codes.
3. Nest concealment: Estimate the percent of the nest (nearest 10%) obscured by vegetation when viewed from 1m directly above the nest. **To reduce bias, use an "ocular tube" (piece of PVC approx. 1.5" inside diameter x 4.5" tube length (James and Shugart 1970) a cardboard toilet roll can also be used.** When estimating concealment, view the nest from 1m above, looking through the tube with one eye while keeping the tube centered on the nest. For species that do not have vegetation that falls over the rim of the nest (e.g. waterfowl, plover, loon, gull, and jaeger nests), the nest concealment value would be 0%. This measurement is taken during mid-incubation.
4. The popsicle stick with the nest ID will be placed in the ground at the edge of the nest once the nest is completed for a year. Leaving these markers allows us to document reuse of nest cups.

## *Recommendations to reduce anthropogenic effects on predation rate*

1. **Avoid leaving scent at the nest.** Common mistakes include the following: touching vegetation around the nest with hands (use a stick or nest marker if necessary), standing at the nest while making the nest card or marker (move away a few meters where you can still see the nest), or placing a backpack or notebook on the ground near the nest.

2. Conduct nest checks from a distance using binoculars if possible. Assume that the presence of an incubating adult indicates an active nest. However, if it is close to the hatch date, flush the bird and check the nest contents.

3. Avoid creating dead-end paths when checking nests. Approach the nest along one route and leave on another. This will make it more difficult for predators to locate nests by watching your activity or following your scent.

4. **Do not approach an active nest if predators are nearby or watching you.** Stop nest searching when predators are in the area. Do something else and return later.

5. Do not sit down or set down your pack or other belongings near a nest.

6. **Only touch nest contents if floating and measuring eggs and use surgical gloves if possible.**

7. Do not eat on study plots. Eat at least 50m outside of the plot boundary.

8. Do not urinate on the plots. When urinating off plot, do so in water to diffuse scent.

9. Cover unattended waterfowl nests with down, feathers, and vegetation to conceal them from avian predators.

10. Collect only necessary data at each nest site and leave as soon as possible.

11. Minimize time spent at nests, especially when taking pictures

# ECOLOGICAL MONITORING

## *Objective*

When establishing a long-term monitoring Network it is important to employ standard techniques for ecological monitoring components. Daily species lists, food resources collection, predator and alternative prey indices, and site and weather condition descriptions complement interpretation of avian monitoring studies.

## *Daily camp journal*

See data forms for *daily camp journal* and *daily species list*.
Adapted from Barrow Protocols and ArcticWOLVES (Smith et al. 2008).

Each camp will maintain a daily camp journal that details the activities at the field camp. This information can be hand written on the daily camp journal data form. Each day's entry should include a summary of the following daily information:

- Personnel on and off duty, including where they worked and what they were doing.
- Arrival/departure of personnel
- Denote ecological monitoring surveys, bug collection, snow surveys, etc.
- Summary of the day's weather
- Significant natural events
- Rare sightings
- Visitors (coming and going)

The journal should also note seasonally important natural history observations – such as how late the snow year is, when the first mosquitos came out, how many lemmings are present this year.

## *Daily vertebrate species list*

Each camp will maintain a list of daily incidental observations of vertebrate species, **including observer effort**, to understand the relative abundance of terrestrial vertebrates. This will be useful in comparisons between study sites or years within one study site. This protocol outlines a technique for providing a relative abundance estimate based on incidental observations.

The technique can be applied to virtually all vertebrate species, whether or not they are subjects of more accurate and labor-intensive population estimation protocols. There is value in obtaining a record of incidental observations for all of our focal prey and predator species, because these give an index of the relative levels of activity within the study area. However, the technique is also useful for providing insight into the abundance and timing of less frequently encountered species, such as certain birds (waterfowl, ptarmigan) and mammals (e.g., ungulates, larger wide-ranging carnivores, weasels).

The relative abundance of species is recorded as the number of individuals seen per hour spent in the field per observer. We will group these data for all observers over selected time periods to give a cumulative encounter rate index. Please take note of each observer's effort (if contributing to the daily species list) including the number of hours spent observing and mode of transportation on the Daily Camp Journal. The likelihood of making observations varies with the observer's activities and mode of

transport in the field. The observer should record whether they were on foot, using a motorized ground vehicle (boat, ATV, Snowmobile), or using aerial transport. If the observer(s) were doing focused ground work (e.g., vegetation sampling, or building exclosures) with little likelihood of making observations in the broader landscape, that time should be subtracted from the total time elapsed for observations.

Encounter rates should be calculated for time spent in the field, away from base camp. Observations made right from the camp should be recorded separately, and merely as a record of species observed, rather than an encounter rate. This is because it is difficult to estimate the amount of time spent in such observations at camp when much of the observers' attention is mostly on camp infrastructure.

Records should be kept daily in field notebooks, including start and end times for the period of the day spent in the field doing activities that have a reasonable chance of allowing observations, and a list of species with numbers of individuals, sex and age if possible, and any comments about unusual sightings. These should be written on the daily species list data form and transferred to a digital data file if possible.

## *Predator and alternative prey indices*

These data will be used to create an index of potential predators and lemmings at each study site, which will be incorporated into analyses as covariates when investigating adult and nest survival rates of species. In 2010, a point count method was used to document predator numbers (see Appendix K) and Network collaborators requested a protocol change to address the low encounter/detection rates at some sites. Accordingly, an index approach was developed to cover a wider geographic area within each study area and to allow predator abundances to be recorded throughout the day. Some camps will continue doing point counts along with this new method (detailed below) to preserve long-term continuity. We also added a lemming abundance count to this same protocol (i.e., lemmings [aka alternative prey] and predators will be counted concurrently). Lemming-only focused surveys will be conducted separately and are detailed below in the Lemming Surveys section.

**Count method**

Counts will be conducted daily or at least once per week depending on predator and lemming levels (high levels = less frequently) by at least one or multiple observers. Designated "predator-lemming counter (s)" will record observations of all predators and lemmings throughout the day regardless of their primary activity (e.g. banding, nest searching, and/or environmental monitoring).

Live counts will be conducted by the "designated counter" by keeping track of the number of predators and lemmings observed during the day's activity within the study area. **Predator and lemming counts will only occur when people are within the study area and should not include observations gathered during transportation to and from the study area nor incidental observations while at camp** (e.g. these observation should be recorded on the daily species list).

If the size of the study area is large enough to require multiple observers to count during the same time period, **observers will count independently** and make efforts to minimize double-counting. To make comparisons across Network sites, it is essential to not double count predators or lemmings (observers should not overlap in the geographic area surveyed), to only count individual predators or lemmings once, and to keep track of the time spent tabulating animals. For camps without plots, please define study areas surveyed using a map. This could be done at the beginning of the season to represent

"sections of the study area" visited over and over. This is important so we have an approximate area surveyed during a given day.

**Daily predator and lemming count data recorded**

1. Observer name(s) (first initial and full last name)
2. Location: Plot ID/section of study area.
3. Team effort (yes/no)
4. Number of observers
5. Date – dd-mm-yyyy
6. Time of arrival on plot/search area
7. Time of departure from plot/search area
8. Species observed
9. At end of the day (or when leaving a study plot/area), record the number of individuals for each species observed during your time within the study area. Indicate whether your number is an estimate or an exact number. This will make it easier to combine data across multiple observers later. See **Appendix H** for list of potential predator and lemmings, their scientific name and corresponding codes on the data card.
10. Notes
    a. Interactions among predators or with other species
    b. Observation of nest or adult predation on a prey item
11. Observation of nesting/denning predators should be recorded – if a breeding location is found be sure to collect a GPS location, note the species, record nest/den contents (# of eggs or young) and how many adults. Since it will be hard for anyone to know if these data have been recorded yet, we encourage you to record this information during each survey. This will allow us to also document how long a nest/den is active.

**REGARDLESS OF WHETHER A DESIGNATED PERSON IS DOING SURVEYS ON A GIVEN DAY, PLEASE HAVE ALL FIELD PERSONNEL RECORD ALL PREDATORS ON THE DAILY SPECIES LIST AT THE END OF THE DAY. BY FOLLOWING THIS APPROACH, THE DAILY SPECIES LIST WILL BE DONE THE SAME WAY ALL SUMMER.**

## *Lemming surveys*

Besides the daily lemming count mentioned above, we will employ three methods to keep track of lemming abundance within and between years. The first method will be a winter nest count that is done at the beginning of the spring season and indexes lemming abundance during the preceding winter. All camps need to conduct this survey and ***if you do not find winter nests OR see any lemmings at your site, you do not need to conduct live counts during the season***. The second method includes a variety of different ways in which to conduct live counts; these can be done either by directed personnel (see predator and alternative prey indices instructions) or by all people reported lemming sightings during daily species list. These focused lemming counts will be done daily throughout the summer. In the rare years and locations with lots of lemmings, a more directed lemming count will be done less frequently than the daily counts (see live transect count). The third method involves live trapping lemmings. This will only be done at a limited number of Network sites. Each of these methods is described below.

**Winter nest counts**

(adapted from Krebs et al 2008) See *Winter Nest Count* in data forms.

*Natural history*

Lemming abundance over the previous winter is relatively easy to measure indirectly by a survey for winter nests. Lemmings build winter nests of grasses and sedges under the snow and use them to keep warm. They appear to us like a ball of cut grass, (Figure 14) about 12 cm (5 inches) in diameter. Since they are abandoned in spring and not reused, they can be counted and picked up without harming the animals.

Figure 14. Lemming winter nest found during early summer snow surveys.

Both the brown lemming and the collared lemming build winter nests, as do voles like *Microtus* (tundra vole) and *Clethrionomys* (red-backed vole) in tundra habitats. It may be possible to tell what species constructed the nest from small amounts of hair left in with the grass, but this is relatively difficult and time consuming.

*Methods*

In most cases we simply record the nest without knowing what species constructed it. Count only fresh winter nests. Nests that are one year old are usually completely flattened and the grass has a grey color rather than a tan color. **All nests found should be ripped apart to avoid re-counting them the next year.** You may find larger winter nests 30 cm or more in diameter and lined with fur. These are weasel (ermine) nests. Weasels hunt lemmings and voles under the snow and convert lemming nests to their own use. Often you will find lemming stomachs left behind in weasel nests. We record weasel nests separate from lemming nests, since it gives a rough indication of the amount of weasel predation over the past winter.

Winter lemming nest surveys should be done as soon as possible after snow melt (early in summer), since high winds can blow the nests around after the snow melts. At high Arctic sites where vegetative growth is low, these searches can happen later in the summer. They cannot be done with great confidence in dense willow habitats or in tussock tundra where the winter nests may often be invisible under the tussocks and willows. Do not conduct the survey in these areas.

To obtain a density of winter nests, we will employ the line transect method as follows:

- The observer walks a straight line searching visually for lemming nests. Upon sighting a nest, he or she records the perpendicular distance of the nest from the line of travel. The data set consists of these perpendicular distances and the total length of survey line the observer walks. This information will be later used to estimate density using the Program DISTANCE.
- This approach has several key assumptions, as illustrated in Figure 15. First, this method assumes that all winter nests exactly on the line of travel are detected. Second, as the distance from a nest to the line travel increases, the likelihood of detecting it decreases. **Another key point, if you detect nests you were not aware of when pacing out towards a nest (to get perpendicular distance or to destroy a nest), you should not include it in your data.** Finally, the perpendicular distance to each nest seen is measured, no matter how far it is from the line of travel.

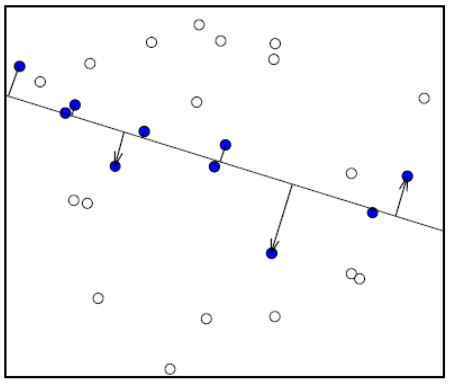


Figure 15. Schematic of winter nest count transect circles represent winter nests, and the line marks the survey line walked by the observer. Solid circles represent winter nests seen by the observer, and the lines mark the perpendicular distances measured to the center of the winter nest. Figure from Krebs et al 2008.

Line transect sampling should be done until at least 40 nests are seen and their perpendicular distances measured. In general, about one day of walking effort per site will be sufficient to generate an estimate of the number of winter nests per hectare. The larger the sample size, the more precise the estimate will be. The distance traveled can be determined from a GPS, or alternatively by following grid lines within your plot, and then summing up the distance from grid stake to grid stake. Record start and stop points for your line transect on the camp GPS coordinates page. On the tundra it is clearly impossible to walk a straight line, but this should not matter as long as one does not double back to cover the same ground.

**Data to record on lemming winter nest count**

1. Site (Network site name)
2. Date (dd-mm-yyyy)
3. Start time (24 -hr)
4. End time (24-hr)
5. Observer (e.g. first initial and last name)
6. Approximate distance (km): Estimate distance walk for transect count based on GPS data or grid stakes.
7. Start location: (Unique plot stake id or GPS location)
8. End location: (Unique plot stake id or GPS location)
9. Nest number (Used to help keep perpendicular distances unique and track sample size )
10. Perpendicular distance to nest (measure in decimeters)

**Live lemming counts**

Daily species list record of lemming numbers (do in low to mid lemming years)

All field observers record lemmings throughout the day and tally up the number of lemmings observed on the daily species list. Details on daily species list are above.

Live transect counts (do only on high lemming years! e.g., 100 lemmings/day/person)

In the rare years where lemmings are everywhere and it is impractical to count them during regular duties, lemmings will be indexed by conducting focused lemming transects. Here, the observer should focus entirely on lemmings and nothing else. It will likely be sufficient for one person to spend two hours doing such counts, and they should travel at least 2 kilometers. These counts should be done weekly (intensive effort) or three times per year (early, mid and late for less intensive effort).

**Data to record on lemming live transect counts**

1. Site (Network site code)
2. Date (dd-mm-year)
3. Start time (24 -hr)
4. End time (24-hr)
5. Observer (e.g. first initial and last name)
6. Start location: (Unique plot stake id or GPS location)
7. End location: (Unique plot stake id or GPS location)
8. Transect ID (Unique transect identification code that corresponds to meta database
9. Approximate distance (km): Estimate distance walk for transect count based on GPS data or grid stakes.
10. Species (4 letter species code, see **Appendix H**)
11. Total number of individuals observed / species.

**For camps conducting lemming trapping, please see Appendix M for instructions.**

## *Invertebrate food resources monitoring*

*Note: Only camps with independent funding for invertebrate sample analysis should collect samples in 2013. The Network-wide funding for invertebrate analysis has been exhausted, and there will not be any Network supported analysis of samples collected in 2013. However, some camps are still collecting data, and invertebrate data are very important to core aspects of the Network analyses, so the protocols are included here for reference by those camps still employing them. We encourage any sites that can handle their own sample analysis to continue invertebrate sampling.*

The distribution, phenology and abundance of invertebrate adults and larvae will be measured in terrestrial and aquatic habitats. We will identify and enumerate samples to acquire estimates of biomass, biodiversity, emergence and seasonal abundance. Trapping methods were designed to be comparable with on-going and historic shorebird food resources studies (e.g. ArcticWOLVES, MacLean 1969, MacLean and Pitelka 1971).

**Supplies required for food resources monitoring**

| ***Supplied by each camp leader*** | |
|---|---|
| **Item** | **Use (s)** |
| Propylene glycol (non-toxic anti-freeze) | To preserve DNA and inhibits trap solution from freezing |
| 20 rebar (1 ft long and ½ inch diameter) | Anchoring screen above pitfall trap |
| 20 10-inch nails | Anchoring trap to ground |
| Duct tape (big roll should be plenty) | Binding objects of all types together |
| 1 ½ gallons (~6 liters) 95% denatured ethanol | Sample preservation |
| Triton soap | To break surface tension in pitfall trap solution |
| One 4-inch fine mesh fish net (brine shrimp mesh) | For filtering insect from the solution samples |

| | |
|---|---|
| Whirl- Paks | For storing invertebrate samples |
| All materials for terrestrial pit fall traps (e.g. pvc pipes and glue, window screen netting, zipties) | Building terrestrial traps |
| One 5-inch fine mesh fish net | For collecting aquatic sweeps |

**Terrestrial invertebrate monitoring**

Terrestrial food resources will be collected from mesic and dry terrestrial habitats using modified Malaise pitfall traps. Terrestrial sampling will commence when the habitats are mostly snow and ice free (and likely before aquatic traps). The terrestrial pitfall trap design is similar to those used by ArcticWOLVES and includes modifications from Bylot Island in 2009 (J.F. Lamarre and E. Bolduc pers. comm.). It is recommended that each site permanently establish **2 trap line transects of 5 pitfall traps each.** The traps should be spaced 15 meters apart and the lines be put somewhere convenient for frequent sampling. One trap line will be established in a dry habitat-type (e.g. centers of high-centered polygons, non-patterned ground, or generally dry areas), and one transect line in more mesic (wet) habitats (e.g. pond edges and rims of low-centered polygons or strangmoor).

Pitfall trap solution

To effectively capture and preserve the terrestrial arthropods a solution for the pitfall traps must be created. The solution will contain 20-30% propylene glycol (non-toxic antifreeze) and 70-80% water with a drop or two of TRITON X-100 (a commercial-grade surfactant). Add this solution to the bottom of the pitfall trap to completely cover the bottom but so it does not pour out the overflow (about 1 inch or 2.5 cm deep). The solution should be changed periodically: weekly, or when the solution is dirty. Pitfall trap solution should be disposed of by flushing down a toilet, putting down a gray water sump, or at remote camps by dispersal.

Traps can be emptied by releasing the guy lines on the trap, lifting the screen off the rebar and placing it on ground. The pitfall container is accessible for removal and emptying of contents into Whirl-Pak (see Field collection procedures below).

Materials needed in the field for terrestrial sampling (see collection procedures below):

- 4 $inch^2$ or 12.5 $cm^2$ fine mesh dip net
- Squirt bottle with water
- Whirl-Paks (10 total per collection, 5 for mesic and 5 for dry)
- Pre-labeled Rite-in-the Rain labels
- Extra *fresh* water for wash bottle
- Meter stick
- Empty container for carrying used solution to camp
- Sawed-off juice or milk jug for catching solution as it goes through net
- Sponge

Pour contents (solution and captured insects) of pitfall container through the fine mesh fish net into an open container (e.g. big juice container) so solution can be returned to pit fall trap. Make sure the net's seam is on the outside so insects do not get stuck in the seam. The key is to first concentrate the collected material into the apex of the net by repeated dipping into a nearby wet area (but do not allow

edge of net to go below surface since you may collect other material this way), or flushing with the squirt bottle. Then let excess water drain, or even blot the captured material from the outside. A moist sponge is great for this if you plan ahead, but you can use any absorbent material that is not already saturated (e.g. damp clothing, moss). Transfer insects from net into a standard 6-inch diameter Nalgene powder funnel fit snugly into the mouth of a wide-mouth, 4oz (125ml) LDPE poly bottle whose bottom has been cut out (Figure 17). The cut off bottle serves as a funnel extension that can be inserted into the Whirl-Pak. This holds the Whirl-Pak open, and directs the sample into the bottom of the Whirl-Pak. Be sure not to leave material stuck to the inside of the bottle! One can hold the whole business in one hand, and use the other for the squirt bottle. Or if two hands are needed for the net and bottle, the unit can be propped up in the vegetation, in a pail or backpack, or held between the knees. It can be difficult to remove the net contents if the sides of the net bag stick together.

## *Aquatic monitoring*

*by Mac Butler – North Dakota State University*

Aquatic food resources will be measured using sweep-net sampling techniques at all sites. This method entails collecting aquatic invertebrates at the water surface along the downwind edge of ponds. In 2011, selected sites (Nome, Canning River, East Bay) maintained surface associated activity traps (i.e., pop-bottles) to assist in making conversions of the pop bottle traps to the sweep net method implemented in 2011. It is recommended that each site return to the same 5 ponds investigated in 2010 and 2011 to conduct aquatic invertebrate sampling unless these ponds proved not to be permanent. Ponds should have water present throughout the summer (i.e., permanent). It is advisable to look at remote imagery maps to look for ponds that appear very dark colored or use your personal experience at a site to locate permanent ponds. Permanent ponds are characterized by having rather sharp edges, edges lined with either sedges (*Carex* sp.) or pendant grass (*Arctophila sp*.), and mud bottoms in the middle.

During emergence of winged adults, all aquatic insect species become available to shorebirds that forage along pond margins. Collecting standardized sweep samples with a small dip net will permit monitoring of the seasonal timing of insect emergence. The bottle traps used in 2010 (and retained at selected sites in 2011) collect invertebrates swimming or crawling in the top 10 cm of water, but detection rates of emerging insects are low with this method. Many arctic insects remain on the water surface to swarm and mate, while other species take flight and leave the pond. In either case, emergence failures and spent adults will accumulate along the leeward shore, where they decompose after several days. This flotsam contains the cast exoskeletons (exuviae) that are shed when aquatic insect larvae or pupae emerge to their adult form. By collecting a regular series of semi-quantitative samples of this washed-up material throughout the nesting season, one can document the seasonality of insect emergence. This material will reflect the whole insect community in a pond, including species from microhabitats that may not be sampled well by the activity traps. Arctic insects typically show highly synchronous emergence within a species, but both seasonal timing and the total span of community emergence can vary considerably.

**Materials needed in the field for aquatic sampling**

Prepare ahead of time, you will need a standard set of Whirl-Paks or collection jars for each sampling event that can be used repeatedly throughout the season. Leave bulky supplies (net, funnel, bottle, etc.) within proximity of the transect, allowing sampling to occur en route from other projects.

- 5 inch$^2$ or 12.5 cm$^2$ fine mesh dip net for collecting sample
- Squirt bottle with water
- Whirl-Paks and sample pre-filled out Rite-in-the Rain labels. 5 per collection.
- Funnel with cut-off polybottle extension for sample transfer to Whirl-Pak.

**Sweep net technique**

Samples should be collected where there is windswept accumulation of material, so the exact sampling location may change between sampling dates. It's important to use the same sampling effort (i.e. five 1m sweeps) each time (e.g. across different days and ponds). Realize that less abundant species in one sample may be swamped by high numbers of another species, so a consistent effort is necessary for each species to have a similar detection probability. The most valuable data will come from counts of the pupal exuviae (cast skins), so be sure to follow through with consistent collection and sample processing, even if it appears that little or nothing is being caught.

Go to the downwind shore (or where wind has blown across the water surface for most of the past 24-hr (Figure 16). More material will accumulate during a period of time when there is higher wind to produce greater waves. Look for white foam along the shore, or a line where the emergent vegetation may serve as a filter to trap drifting material. Use the 5-inch brine shrimp mesh dip net to skim the water surface along this line and parallel to the lake shore **for a 1-meter long "sweep"**. The net should gather materials floating at the surface, thus the bottom of the net can be about 2 inches (5 cm) below the surface, with the top half of the net above surface. Also sweep the net along the edge of the vegetation so that you don't collect an undo amount of vegetation in your net. This may reduce the number of invertebrates collected but will greatly streamline picking of insects later, and if done consistently through time and across camps will still allow comparisons. Use a meter stick to estimate the distance of each sweep. Collect five (5) replicate 1m-long sweeps from the downwind edge of each pond; the 5 sweeps can be contiguous, but don't re-sweep the same 1m section.

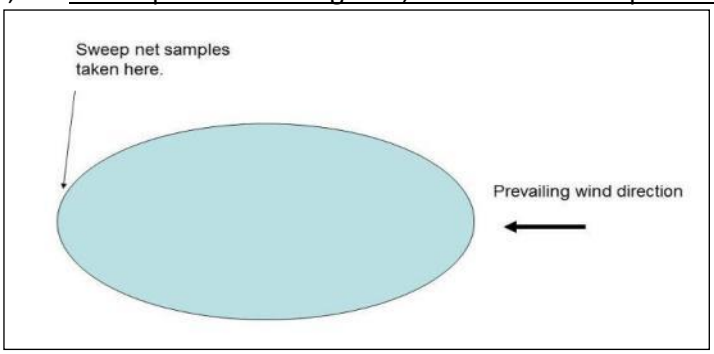



Figure 16. Schematic of location in pond where aquatic invertebrate emergence sweep net samples will be collected.

Deposit the material collected in all 5 replicate sweeps into a single Whirl-Pak. Any large pieces of vegetation can be picked out by hand and discarded, but should first be rinsed over the net with the squirt bottle (filled with preservative) to flush off clinging insects or exuviae. The key is to first concentrate the collected material into the apex of the net by repeated dipping into the pond, or flushing with the squirt or spray bottle. Let excess water drain, or even blot from the outside. A moist sponge is great for this if you plan ahead, but you can use any absorbent material that is not already saturated (e.g. damp clothing, moss). Then the net bag can be inverted so the bulk of collected material falls, and/or can be readily flushed, into the container (or funnel). Use the funnel with polybottle extender to flush the material from the net into the bottom of the Whirl-Pak (Figure 17) with preservative. It helps to pull on the metal corners of the Whirl-Pak to make it fit on the bottle, just

enough to still be tight on the bottle. This may take 3 hands: one to hold the container and funnel, and two to invert the net and rinse with the squirt bottle. One person may do this by holding the Whirl-Pak and funnel between feet or knees, thus freeing both hands. It is best to plan on doing this transfer in the field with the preservative (i.e. bring along extra preservative for the squirt and spray bottle for refills).

At times you may notice little in the net, but under magnification there will likely be evidence of insect emergence, in the form of cast larval and pupal skins. These insect exuviae are often small and nearly transparent, but for some species they may exceed 10 mm in length and be grey, brown, or gold in color. At times of heavy emergence the insect bodies and pupal skins may accumulate in great quantity. Some sites may wish to collect samples daily during peak emergence.


Figure 17. Powder funnel with polybottle extender (bottom removed), used for transferring aquatic samples into Whirl-Pak.

**Field collection procedures**

Samples will be stored separately for each trap (regardless of type) in Whirl-Paks filled with preservative solution (ethanol). Do not combine samples from different traps or ponds that were swept with nets. Each Whirl -Pak should have a cut (**do not tear paper**) Rite-in-the-Rain label inside that indicates the following information:

- collection date
- camp name
- sample type [for terrestrial (wet or dry) and for aquatic (sweep)]
    - (e.g. Terre Mesic 1, Terre dry 1, Aquatic pop 1, Aquatic sweep 1).
- Collector's name

This information should be written in **pencil** (not pen) on Rite–in-the-Rain paper so the data are not lost once placed in preservative. DO NOT USE REGULAR PAPER AS IT WILL DESINTEGRATE IN ALCOHOL. Ink from Sharpie pens will also be lost in alcohol. Cut the labels with scissors or a knife, because bugs can get caught in the rough edges of torn paper labels. The outside of the bags should also be labeled with an alcohol proof pen (see below). ***Do not crush the specimens during transport back to camp.***

## *Weather*

Daily weather conditions will be measured at each site using a locally established field camp weather station (HOBO U30 datalogger) or by retrieving data from established weather stations (e.g. Barrow, Prudhoe Bay, Churchill sites). The field camp weather stations will gather hourly measures of air temperature, relative humidity, wind speed and direction. Precipitation will be measured manually at each site with a rain/snow gauge. Weather stations should be checked daily to ensure they are functioning and data should be downloaded **weekly** using the data backup data shuttles. Be sure to download the data off the shuttles and view this on the computer to be sure data are being recorded correctly. In the event that the field station weather station is not recording it is important to collect daily minimum weather conditions (see below for procedures).

**HOBO U30 data logger weather stations specifications**

| **Measurement and location of sensor:** | **Instrument specification:** |
|---|---|
| Air temperature and relative humidity: 2.5 m above ground | Smart Sensor Temp/RH (Onset model S-THB-M002) |
| Wind speed and direction: 3 m above ground | Smart Sensor Wind speed and direction (Onset S-WCA-M003) |
| Precipitation (see below): ½ meter above ground | A separate rain gauge is employed for this. It is not part of the automated station but needs to be mounted at correct height. |

**Daily precipitation** – recorded manually (from Cadieux and Gauthier 2008)

We want the data to represent the daily accumulation of precipitations, it is recorded only once at the end of **every day** between 9PM and 10PM. If it does not rain, you can put zero without checking it. This can be adjusted depending on the latitude of each study sites. Precipitation is recorded in **millimeters**. Precipitations include rain, drizzle, freezing rain, freezing drizzle, hail and snow. All these types of precipitations, *except snow*, can be measured using a standard rain gauge (pluviometer) installed at 1.5 meters above the ground. It is important to make sure that it is installed **vertically** and that nothing will obstruct the arrival of the rain within a radius of a few meters from the funnel. Hence, care must be taken to locate the rain gauge above the highest point of the structure where it is installed (e.g. roof of a low building or a tent).

Snowfalls are not recorded using a pluviometer. Instead, it should be estimated by measuring the accumulation of snow (in cm) at a few locations on the ground using a ruler. The average of these measures can then be transformed into rain accumulation using the "ten-to-one" rule (1 cm of snow = 1 mm of rain). *See data forms for manual weather recording*.

**Manual weather recording**
(in the event the automatic weather recorder goes down)

**Daily air temperature** Minimum and maximum air temperatures are recorded twice daily (12 hrs apart) at consistent time period.

**Daily wind patterns** A description of the daily wind including speed and direction (e.g. N, W, S, E, NW, NE etc.) should be noted in the daily journal. Please include pattern of wind throughout the day, a range of wind speed for the day and the predominant direction.

**Snow and surface water**

The timing of snow melt is believed to be a key determinant of timing of breeding for shorebirds (e.g., Meltofte et al. 2007, Smith et al. 2010). Our intention is to monitor snowmelt with sufficient resolution to determine the date of 50% melt, which will allow us to detect differences among years at one site, and among sites in general. Throughout the season we will collect frequent visual measures of relative surface cover (e.g. snow [includes slush], water and land) to quantify the seasonal changes in hydrology. All sites will collect at least 10 replicate samples of relative cover at permanent locations (Record GPS locations of each survey location in the geographic meta-database) preferably **every other day** during the beginning of the season when the snow melts quickly, and **weekly after the snow has melted until the field season is completed.**

For sites with established plots, relative surface cover will be quantified by randomly or systematically selecting **at least ten 50- $m^2$ quadrats** within the study area and visually estimating absolute snow (which includes ice and slush), water and land (bare or vegetated ground) cover within the quadrat **to the nearest 5%, totaling to 100% for each station** (for plots, you might reference each station with the northwest stake number).

For sites without established plots, relative surface cover will be quantified by establishing 10 survey stations and estimating snow cover in **50-$m^2$ quadrat** surrounding the observation point. To increase the accuracy and consistency of this method, observers must pace 50 meters in at least 2 directions and pin flag the corners and the center. Efforts should be made to have surface cover sampling areas be spread throughout the study area as much as possible to give a representative sample of snow melt for the area. Camp leaders can reduce observer bias by training all observers in estimating cover at the beginning of each season or designating one observer to practice and conduct all snow surveys. Crew leaders can have multiple observers estimate cover and discuss ways to calibrate between observers. Cover estimates are more accurate if observers subdivide area into smaller units and estimate cover for each subunit and sum for the total estimate.

For example, the NW quadrat (Figure 18) contains 25% land, NE quadrat contains 5% land, 10% water and 10% snow, the SW quadrat contains 15 % land, 10 % water, and the SE quadrat contains 15% land, 5% water and 5% snow for a summed total of 25 % water, 60 % land and 15 % snow.

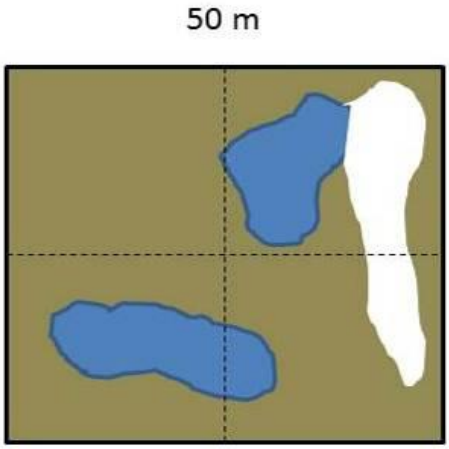



Figure 18. 50 $m^2$ quadrat for estimating relative surface cover. Surface cover values for each category illustrated here are as follows: 25 % water, 60 % land and 15 % snow.

## DATA ENTRY AND QUALITY, MANAGEMENT, AND SUBMISSION

There are 12 ASDN electronic databases and in these databases there are 18 worksheets for data entry (Table 6). The most recent version of this protocol, data forms, and databases can be found on the web at: http://www.manomet.org/arctic-shorebird-demographics-network. ASDN data forms were designed to be used as standard field data forms; they are distributed in an electronic format for camps to configure to their camps individual needs. Data should be entered throughout the season directly into the ASDN databases provided. Data entry during the field season allows errors in data collection to be identified and corrected, and results in the reduction of future data collection errors. Each database has a "Readme" worksheet that defines the database's fields, and lists standardized codes and details on how to correctly enter data. Follow the codes exactly as they are listed and if you must create a new code for any field, please highlight (e.g. turn row yellow) and define the code in the your data submission summary (see instructions below).

Table 6. File names of Arctic Shorebird Demographics Network data forms and electronic databases.

| **Data form (word files, .docx)** | **Electronic database (excel files, .xlsx)** |
|---|---|
| Field_camp_metaDB_information | Field_camp_info_metadata_ASDN_electronicDB_2013 |
| Field_camp_metaDB_study_area_map | None |
| Field_camp_metaDB_geo_information | Metadata_geo_ASDN_electronicDB_2013 |
| Daily_camp_journal | Observer effort data recorded in daily species_ASDN_electronicDB_2013 |
| Resight | resight_ASDN_electronicDB_2013 |
| Bird_band_record | band_ASDN_electronicDB_2013, (2 worksheets, one for adults and one for chicks) |
| Nest_record_sheet, includes chick band records | Nest_ASDN_electronicDB_2013, (2 worksheets, nest monitoring and Eggmm). *Chick data recorded in band database.* |
| Daily_species_list | daily species_ASDN_electronicDB_2013 (3 worksheets: daily records, first occurrence dates, observer effort*) |

| | |
|---|---|
| Snow_and_surface_water | snow_surface_water_ASDN_electronicDB_2013 |
| Manual_recording_and_daily_precip | manual_weather_ASDN_electronicDB_2013 (2 worksheets, snow fall and rain) |
| Manual_snow_fall | |
| Food_resources_collection_catalog | food_resources_catalog_ASDN_electronicDB_2013 |
| Lemming_winter_nest | lemming_data_ASDN_electronicDB_2013 (2 worksheets, winter counts and live counts) |
| Lemming_live_counts | |
| Lemming_intensive_trapping | No electronic database, developed by each site |
| Predator_lemming_index | daily_predator_lemming_index_ASDN_electronicDB_2013 |

## *Data quality*

Data quality management requires a concerted, cooperative effort by people throughout the Network. Data from each site is compiled into master databases and archived each year. It is ***extremely important*** to submit data that is accurate and complete. Data should be proofed against the raw data before submission by qualified personnel (someone who will be able to determine errors). It is essential to control data quality at each Network site to avoid GARBAGE IN and GARBAGE OUT for subsequent analyses. **Even though this step is labor intensive and tedious it is the most important contribution each individual can make.**

| DO'S | DO NOTS |
|---|---|
| Proof each data record for accuracy. Focus attention on each field.<br><br>Use lower case letters for all entries<br><br>Enter "." anytime data is missing, do not leave blank cells in fields. Blank cells are problematic because we cannot determine the difference between data not collected and data not entered<br><br>Use the codes defined in the "readme" worksheets. Any new field codes need to be documented in a separate summary document<br><br>Remove fields from the databases that are not part of the ASDN data structure before submission. (e.g. additional project-specific fields such as feather wear). | Add spaces or extra characters or numbers into fields<br><br>Don't create your own field codes unless absolutely necessary<br><br>Drag down dates or other fields automatically, without making sure they are correct. Avoid erroneous autofills. |

## *Data management and submission*

Prepare an annual summary of the camp's "data submission" that contains details on the following topics of interest: new field codes that were created (denote which database and field), missing data, deviations from the protocol, and/or anything of methodological interest. Databases must be proofed and errors corrected prior to submission to the ASDN data manager . Rename files to include your camps 4-letter code before submission by replacing the "*ASDN*" with your camp's four letter code. Attempt to send ALL your final databases by email at the same time to the data manager. **Submit below list of documents and materials and burn CD (s) of raw data electronic archive, final databases and**

**photos by 1 October 2012.** Report errors found in the databases after submission to the data manager by email (arctic.shorebird@gmail.com).

List of documents, naming conventions and materials to submit:

1. Camp data submission summary
    - Naming convention: "submission_camp4lettercode_year"
        - Submission_CAKR_2012.docx
2. All 12 ASDN electronic databases
    - Naming convention: "database name_Camp4lettercode_electronicDB_year"
        - banding_CAKR_electronicDB_2012.xlsx
3. Weather data (Hobo remote weather stations field camps only*)
    - Naming convention: "Camp 4 letter code_weather_startdate_enddate year"
        - CAKR_weather_28May_4July2012.dtf
4. Banding photos (usually 3-4 photos per bird captured)
    - Naming convention "bandnumber_sppalphacode_description"
        - 230155644_DUNL_data.jpg Photo of the datasheet
        - 230155644_DUNL_combo.jpg Photo of the color combo
        - 130155642_SESA_wing.jpg Photo of entire wing
        - 230155644_DUNL_innerwing.jpg Photo of the inner wing
        - 230155644_DUNL_outerwing.jpg Photo of the outer wing
5. Electronic back up of raw data (nest, band and encounter raw dataforms)
    - Naming convention: "ASDN site code_unique recordID_rawdata_year"
        - CAKR_SESA401_nest_rawdata_2012.jpg nest record sheet of SESA401.
        - CAKR_nest_rawdata_2012.jpg all nest record sheets from Cape K in 2012.

* The data manager will retrieve permanent weather station data from the National Weather Service. These sites include Barrow and Prudhoe Bay.

We archive raw data annually and request electronic copies of the three core databases (e.g. band, nest, and resight data forms). Archiving the raw data in one location (e.g. Anchorage) allows us to share the electronic raw data files with side-project collaborators that are working on data analysis. Collaborators can use the raw data to verify and correct data errors without needing to contact the project leaders. Standard naming conventions for the raw data archive somewhat depend on whether your data is lumped into only one file or recorded per data forms (e.g. scans of individual datasheets or photos). Please name the archives in the most logical format and include your camp code, year, and description of file.

Consider contributing your camp's data or summaries of your project to the following efforts:

- *International Breeding Conditions Survey on Arctic Birds* http://www.arcticbirds.net/
- *E-bird* http://ebird.org/content/ebird/
- *Alaska Shorebird Group* http://alaska.fws.gov/mbsp/mbm/shorebirds/working_group.htm

## *Sample labels, organization, and shipment*

Labeling blood, feather, and fecal samples is vitally important to ensure samples can be used and tracked back to the individuals they were collected from. If labels are illegible they will likely be destroyed and not used. The writing on the storage vials and envelopes must be legible and complete. Complete labeling of individual samples will allow accidental mistakes to be resolved (e.g., including

both species and nest # helps tremendously). All blood and fecal sample labels must be written with alcohol proof lab marking pens– do not use a Sharpie as the various preservatives and solutions will remove the ink. Alcohol proof pens and labels are also essential for invertebrate sample storage.

## Labeling blood and fecal samples

1. Use sticker labels provided and include the following information on each vial:
    - metal band number
    - 4-letter species code
    - Unique NEST ID
    - Collection date
    - 4-letter camp code

2. Write band number on top of vial with lab marker
3. Use tape around label to secure for long term storage
4. Organize blood and fecal samples into separate storage boxes by sample type (e.g. avian malaria, genetic, fecal) and species. If possible, organize samples by band number within a species. Clearly label the top of box with the following information:
    - Year
    - Camp code
    - What the box contains (e.g. whole blood in Longmire, species in the box)

## Labeling feather samples

1. Use stickers sent with feather envelopes and include the following information on each feather envelope:
    - metal band number
    - 4-letter species code
    - Unique NEST ID
    - 4-letter camp code
    - Collection date
    - Lat /long coordinates
    - Collection type: “methyl-mercury” or “ASDN archive”
2. Organize samples by project type (methyl mercury or ASDN archive), species, band number

## Labeling invertebrate samples (for sites conducting their own analyses)

1. Cut (do not rip) small pieces of Rite-in-the-Rain paper to create labels to place inside each Whirl-Pak and include the following information, **WRITTEN IN PENCIL:**

    - collection date
    - camp name
    - sample type
        - [for terrestrial (wet or dry) and for aquatic (sweep)]
            - (e.g. Terre Mesic 1, Terre dry 1, Aquatic sweep 1).
    - Collector’s name

2. Write on the outside of each Whirl-Pak the following information Make sure that the Whirl-Paks are sufficiently inflated to decrease the crushing of samples when stored in a box. **WRITTEN WITH AN ALCOHOL-PROOF LAB MARKER:**
    - collection date
    - camp name
    - sample type
        - [for terrestrial (wet or dry) and for aquatic (sweep )
            - (e.g. Terre Mesic 1, Terre dry 1, Aquatic sweep 1).
    - Collector's name

Pool all samples of one type in a quart-sized Ziplock bag and label with date of collection and contents and camp name (e.g. all terrestrial samples for one collection event, e.g. Terre Mesic 1-5). Pool all three samples types (Terre dry, terre wet, aquatic) from one collection event into a one gallon Ziplock bag. Double-bag the samples in a gallon sized Ziplock bag to minimize leakage. Store the entire collection of samples upright in a secure plastic box at field station or camp.

## Post field season shipment of Samples:

***ASDN genetic and feather samples***

Notify Rick Lanctot when you are planning to send samples to Anchorage and he will instruct on how to pay for shipment.

Attn: Rick Lanctot
US Fish and Wildlife Service
Migratory Bird Management-Shorebird Program
1011 East Tudor Rd MS 201
Anchorage, Alaska 99503
907 786- 3609 (office)
907 786 3641 (fax)
Richard_lanctot@fws.gov

***Methyl mercury samples:***

For camp that are using a cryoshipper: Ship cryoshipper with methyl mercury samples back to the USFWS Migratory Bird Management Office (Attn: Michelle St. Peters, address same as above). Instructions and payment for shipment will be included in the arriving shipment pre-field season)

For camps with freezers on site:
Send samples directly to: Jennifer Goyette
Biodiversity Research Institute
652 Main Street
Gorham, ME 04038 USA

Contact: Jennifer Goyette and Kevin Regan (BRI's Mercury Lab Manager) by e-mail: jennifer.goyette@briloon.org; kevin.regan@briloon.org or phone: 207-839-7600 ext.107 (Kevin; mercury lab) for instructions and assistance. To reach Jennifer Goyette by phone call 414-526-0808. *Please include a sample inventory with associated data to the email addresses provided when you ship samples. Also, for methyl mercury shipping see details in Appendix N.

***Avian malaria***

Avian malaria samples should be shipped back to Samantha Wisely and Claudia Ganser at the University of Florida at the following address. Please contact them prior to shipment to determine how to pay for shipment.

Dr. Samantha Wisely
Co-Director of the Molecular Ecology Laboratory,
Dept. of Wildlife Ecology and Conservation,
University of Florida,
110 Newins-Ziegler Hall, PO Box 110430,
Gainesville, Florida 32611
Phone: 352.846.0645, FAX: 352.392.6984,
Email: wisely@ufl.edu (Sam)
e-mail: gancla@ufl.edu (Claudia)

***Fecal samples***

Fecal samples should be shipped back to Kirsten Grond at Kansas State University at the following address. Please contact her prior to shipment to determine how to pay for shipment.

Kirsten Grond,
Division of Biology, 116 Ackert Hall,
Kansas State University, Manhattan, KS 66506
Ph: 785-477-6545
E-mail: kgrond@ksu.edu

***Moss feather samples***

Moss feather samples should be shipped back to Lily Lewis at the following address. Please contact her prior to shipment to determine how to pay for shipment.

Lily R. Lewis
Ecology and Evolutionary Biology
University of Connecticut
75 N Eagleville Road, U-3043
Storrs, CT 06269 USA
Phone: (860) 486-6306
Email: LilyRLewis@gmail.com or Lily.Lewis@uconn.edu

# APPENDICES

## *Appendix A. Project-specific sampling details for 2013 Network breeding season.*

| PROJECT | SESA FECAL | SESA BLOOD | SESA FEATHER | DUNL FECAL | DUNL BLOOD | DUNL FEATHER | All other species* FECAL | All other species* BLOOD | All other species* FEATHER |
|---|---|---|---|---|---|---|---|---|---|
| **Methyl Mercury**<br><br>*N= > 10 /site/species; if cannot get 10 then do not sample species* | n/a | Two, 1/3 filled 100µL of whole blood (freezer), or 1, 1/2 filled 100 µL (cryoshipper) HEPARANIZED capillary tube | One 10th secondary | n/a | Two, 1/3 filled 100µL of whole blood (freezer), or 1, 1/2 filled 100 µL (cryoshipper) HEPARANIZED capillary tube | 5 black breast feathers | n/a | Two, 1/3 filled 100µL of whole blood (freezer), or 1, 1/2 filled 100 µL (cryoshipper) HEPARANIZED capillary tube | One 10th secondary |
| **Avian Malaria**<br><br>*N*=all birds captured | n/a | 100 µL of whole blood in 2.0mL vial pre-filled with Longmire solution, PLAIN capillary tube | n/a | n/a | 100 µL of whole blood in 2.0mL vial pre-filled with Longmire solution, PLAIN capillary tube | n/a | n/a | 100 µL of whole blood in 2.0mL vial pre-filled with Longmire solution, PLAIN capillary tube | n/a |
| **ASDN genetic and stable isotope archive**<br><br>*N=all birds captured* | n/a | 25 µL of whole blood, in 1.5mL vial with 0.5 mL Longmire solution, PLAIN capillary tube | One 10th secondary; both 10th Sec. feathers once sample size for Hg study achieved | n/a | 25 µL of whole blood, in 1.5mL vial with 0.5 mL Longmire solution, PLAIN capillary tube | Birds in flight feather molt: one old and if possible one new primary from each side 5 black breast feathers Birds not in molt: one old 1st primary from each side and 5 black breast feathers | n/a | 25 µL of whole blood, in 1.5mL vial with 0.5 mL Longmire solution, PLAIN capillary tube | One 10th secondary; both 10th sec feathers once sample size for Hg study achieved |
| **Gut microbiota (i.e., fecal sample)**<br><br>*N=all recaptured individuals and additional new samples to total 10/species/site* | 1.5mL vial with 1mL of 100% ethanol | n/a | n/a | 1.5mL vial with 1mL of 100% ethanol | n/a | n/a | 1.5mL vial with 1mL of 100% ethanol | n/a | n/a |

Species-specific sample sizes (gut microbiota side project) need to be determined by each field camp based on side project participation and potential to capture target samples sizes (methyl mercury side project). We recommend not collecting blood for both methyl mercury and avian malaria from smaller birds (e.g., Semipalmated Sandpipers, Western Sandpipers, Red-necked Phalaropes, White-rumped Sandpipers), so do tasks on different birds and keep track of samples obtained.

## *Appendix B. Species-specific nest searching tips*

| Species | Mating system | Adult who Incubates | Nest location | Flushing distance | Flushing and Other Unique Behaviors | Return time |
|---|---|---|---|---|---|---|
| ***Single adult incubation*** | | | | | | |
| REPH | Polyandry – focus on males | Male | Wet | Close | Adult leaves area and monitors nest from a great distance; upon returning the male will typically fly from one little pond to the next until eventually reaching its nesting pond | moderate |
| RNPH | Polyandry – focus on males | Male | Wet | Close | Adult leaves area and monitors nest from a great distance | moderate |
| PESA | Polygynous – focus on females | Female | Wet | Moderate | Often does rodent run and alarm calls ; male can be watched – will sometimes fly and "boom" over female who may be either feeding or on a nest | moderate |
| BBSA | Lekking – single male display indicates nest | Female | Moderate | Close | May do rodent run and alarm call; single male occasionally displays near nest site; frantic feeding female likely on break -- can be followed to find nest. | quickly |
| WRSA | Monogamous to Polygynous | Both or female | Moderate | | | |
| ***Bi-parental incubation*** | | | | | | |
| DUNL | Monogamous – key on male display | Both | Moderate | Moderate | Both adults do rodent runs, but females more likely. Frequently flies off nest low and then lands and calls. | Quick to moderate |
| SESA | Monogamous – male display | Both | Mod to dry | Close | Both adults do rodent runs, but females more likely. | quickly |
| LBDO | Monogamous - pairs | Both | Mod to wet | Very Close | Tight sitter, does not flush easily; very secretive upon return to nest. | Slow |
| STSA | Monogamous – male display | Both | Mod to wet | Close | Tight sitter, does not flush easily | moderate |
| AMGP | Monogamous – look for pairs | Both | Dry | Far | Will stand up or leave nest when observer far away (>100 m) | Quickly if observer is out of sight |
| BASA | Monogamous | Both | Dry to mod | Close | Rodent run and alarm calls | quickly |
| WESA | Monogamous | Both | Mod to wet | Close | Rodent run and alarm calls | quickly |

## *Appendix C. Egg flotation - A method to determine egg age*

Materials

1. Float container (e.g. 3 inch plexiglass cube) with compass angles written on side
2. Water from the nearest natural water body or warm water carried in a thermos container
3. Protractor and ruler

Methods

If a shorebird nest contains a full clutch of eggs (typically 4) when it is discovered, float at least 2 eggs. If the 2 eggs differ in angle significantly float the 3rd egg (and 4th if necessary).

1. Place eggs on the bottom of the jar before releasing to prevent egg damage from dropping and to ensure they are not held by surface tension. Float each egg separately but keep track of the ones you've already floated.
2. With a protractor, measure the angle between the bottom of the cup and the center axis of the egg to the nearest 5° for each egg. Record the angle on the Nest discovery and fate data sheet.
3. If the egg floats at the surface, using a clear ruler, record the # of millimeters above the water surface that is exposed to the air and also use the protractor to measure the angle of the egg in the water column. Keep in mind the egg may float at the surface but not break the surface. Record these measurements while you are viewing the floating egg at eye level.

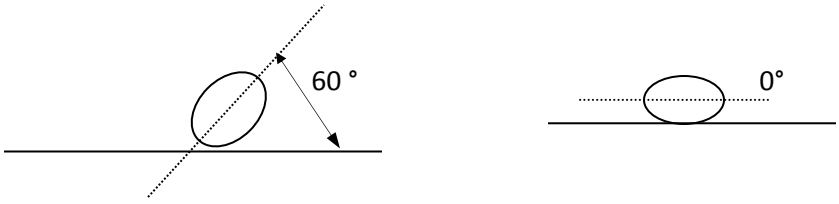
Carefully place eggs back in the nest. Remember to record the date you floated the egg.

Important

Float shorebird eggs **once** during the course of incubation unless you have issues with the original float data. **Float eggs on the discovery visit** because the nest may be depredated on your next visit and age estimates tend to be more accurate when eggs are floated early in incubation.

Use the float program in the excel file found at Manomet webpage (float_program.xls) to estimate the age of the eggs when calculating the estimated hatch date in the office. The program uses regression equations specific to species or use Float Tables in **Appendix J,** that were originally published in Liebezeit et al. (2007). Note the program is designed so that the lowest angle that can be inserted is 21° and the maximum angle is 89°. Since shorebird eggs are oblong, they will never lay flat on the bottom.

If the temperature is below freezing do not float the eggs. Do it on the next visit.



## *Appendix D. Nest stage periods for common breeding birds*

| Species | Laying (days)* | Incubation (days) | Species | Laying (days)* | Incubation (days) |
|---|---|---|---|---|---|
| *Shorebirds* | | | *Water birds* | | |
| Black-bellied Plover | 4 | 23-27 | Pacific Loon | 2 | 23-25 |
| American Golden-Plover | 4 | 26-27 | Red-throated Loon | 2 | 24-26 |
| Pacific Golden-Plover | 4 | 25 | Yellow-billed Loon | 2 | 27-29 |
| Semipalmated Plover | 4 | 24 | King Eider | 4-6 | 22-24 |
| Greater Yellowlegs | 4 | 23 | Common Eider | 3-5 | 24-26 |
| Lesser Yellowlegs | 4 | 22 - 23 | Long-tailed Duck | 5-7 | 26 |
| Whimbrel | 4 | 27-28 | Northern Pintail | 7-9 | 22-24 |
| Bristle-thighed Curlew | 4 | 26 | Tundra Swan | 3-4 | 31-32 |
| Hudsonian Godwit | 4 | 22 - 23 | Brant | 3-5 | 22-25 |
| Marbled Godwit | 4 | 24 -26 | Greater White-fronted Goose | 4-5 | 25 |
| Bar-tailed Godwit | 4 | 20-21 | Snow Goose | 2-6 | 23-24 |
| Ruddy Turnstone | 4 | 22-24 | Cackling Canada Goose | 4-7 | 25-30 |
| Black Turnstone | 4 | 22 -24 | *Passerines* | | |
| Surfbird | 4 | 22 – 24 | Lapland Longspur** | 4-6 | 11-13 |
| Red Knot | 4 | 21 -23 | *Other birds* | | |
| Sanderling | 4 | 23 -27 | Willow Ptarmigan | 4-14 | 21-23 |
| Semipalmated Sandpiper | 4 | 20-22 | Rock Ptarmigan | 6-13 | 20-26 |
| Western Sandpiper | 4 | 20-22 | Pomarine Jaeger | 2 -3 | 26 - 28 |
| Least Sandpiper | 4 | 19 - 23 | Parasitic Jaeger | 2 | 25-28 |
| White-rumped Sandpiper | 4 | 21-22 | Long-tailed Jaeger | 2 | 23-25 |
| Baird's Sandpiper | 4 | 21 | | | |
| Pectoral Sandpiper | 4 | 21-23 | | | |
| Rock Sandpiper | 4 | 20 - 21 | | | |
| Dunlin | 4 | 21-22 | | | |
| Stilt Sandpiper | 4 | 20 | | | |
| Buff-breasted Sandpiper | 4 | 23-25 | | | |
| Long-billed Dowitcher | 4 | 21-22 | | | |
| Short-billed Dowitcher | 4 | 21 -22 | | | |
| Red Phalarope | 4 | 18-20 | | | |
| Red-necked Phalarope | 4 | 19-21 | | | |

* Birds typically lay one egg / day. ** Nestling stage = 8-11 days

Note: The information in this Appendix was obtained from various sources in the literature (mostly BNA accounts and the Birder's Handbook – Ehrlich et al. 1988). Nest stage lengths may vary somewhat across sites. Note that the float equations use a fixed length of incubation time for each species (not range as reported here).

## *Appendix E. Using eggshell remains to determine nest fate in shorebirds*

The presence of pip fragments and eggshell "tops" and "bottoms" at Piping Plover, Snowy Plover, and Killdeer nests typically indicate a successful hatch (Mabee 1997). This evidence is probably reliable for assessing the fate of other precisian species. The following is a brief description of a methodology for finding eggshell remains and assessing fate of the nests they are associated with.

Egg shell top and bottoms: those parts of an eggshell that exhibit a nearly equidistant length from the center of the top or bottom eggshell to the broken edge of the shell. Many hatching chicks pip through an eggshell at a fairly uniform level around the top of the egg and produce well-defined tops and bottoms.
Eggshell fragment: pieces that range from ~ 1 to 5 mm. Fragments within this size range are expected in successful nests because chicks break through the eggshell and produce small pipping fragments when hatching.
Eggshell pieces: any piece larger than 5 mm in length. Fragments > 5 mm may be found in both successful and depredated nests and are less reliable in classifying nest fate.

For finding eggshell tops and bottoms:
When a nest is no longer active and you are assessing its fate, search within a **5m radius** of the nest for eggshell tops and bottoms. Most shorebirds remove eggshells from the nest immediately after the chicks hatch. They often deposit the eggshells not far from the nest. Remember to check to see if the membrane adheres to or easily pulls away from the eggshell "tops" and "bottoms". This may be difficult to determine for the smaller shorebird species.

For finding pip fragments
With the tip of a mechanical pencil or tongue depressor carefully pull away the top layers of the nest. Continue all the way down to the soil. Pip fragments often sink into the nest lining.

1. Some birds may re-use a nest scrape from a previous year and may contain pip fragments from a previously successful nest. Only count pip fragments that look new. New fragments typically are bi-colored (i.e. different colors on the 2 sides – mottled on outside of egg, white on inside).
2. If you do not find fragments after reaching the bottom of the nest, place the nest contents on a sheet of white paper (or on the center-fold of your rite-in-the-rain field notebook). Carefully go through the nest contents as you place it back in the bowl.
3. Inexperienced researchers may collect the nest, place it in a plastic bag or Tupperware and bring it back to the office for further scrutiny.

- Be careful not to confuse egg fragments with pieces of lichen
- Sometimes you will just find eggshell membrane pieces (and no actual shell fragments)

Phalaropes and dowitchers often leave a large number of eggshell fragments in the nest, often pip fragments are > 5mm for these species.

## *Appendix F. Nest fate determination and causes of nest failure*

(Adapted from WCS Alaska tundra predator study)
Also See Appendix R for helpful nest fate scenario examples.

| **SUCCESSFUL** | |
|---|---|
| At least one egg hatched.<br><br>This is confirmed if chicks are located in the nest. If no chicks are in the nest, then at least one of the two conditions must be met:<br>1) Hatchlings are observed within 50m of the nest within 2 days of the expected hatch date<br>2) The nest contained fragments of pipped eggs (typically 1-5 mm in size) and/or eggshell tops/bottoms are observed within 5m of the nest AND it is within 4 days of the estimated hatch date based on:<br>• Starring or pipping of eggs on previous visit.<br>• Dating from known laying dates or laying dates estimated from float tables.<br><br>The following lines of evidence suggesting a successful nest are considered much less reliable. They should be recorded as supporting evidence but not used as one of the two conclusive lines of evidence:<br>• Parent bird(s) defending nest territory<br>• Presence of infertile eggs remaining in an inactive nest | |
| **FAILURE** | |
| Assume a nest has failed if the nest contents are gone prior to 4 days before the calculated hatch/fledge date OR if it is within 4 days of the calculated hatch date and there is not adequate evidence to classify as successful (see above). Only classify a nest as "failure" if you are sure it failed but cannot determine the cause of failure (See below for causes of failure) | |
| **UNKNOWN** | |
| Put a nest's fate as unknown If you cannot determine the fate of the nest or there is conflicting evidence of hatching, fledging or failure (e.g., shorebird eggs gone > 4 days before hatch date yet egg fragments are found). Nests within 4 days of calculated hatch/fledge dates that do not have adequate evidence to classify as successful and do not have evidence of cause of failure (i.e., signs of predation, weather, trampling, etc.). | |
| **UNDETERMINED** | |
| Nest occupied but monitoring discontinued (e.g. nest never actively monitored or the nest was still active when field work was concluded). | |
| **CAUSES of FAILURE** (If there is evidence for cause of failure, classify as follows: | |
| **PREDATION** | |

Assume a nest has been depredated if predation was observed or there is at least one line of evidence strongly indicating predation. The following evidence is suggestive of a predation event and should be recorded if observed:

- Eggs are missing from nest > 4 days prior to the estimated hatch/fledge date
- Attached membranes with blood or yolk
- Destroyed nest; egg fragments (larger than 5mm for shorebirds) outside of nest
- Remains of adult or chicks within 10 m of the nest
- Fox urine or scat at empty nest is confirmatory evidence but must occur with other signs above to indicate predation.

**ABANDONMENT**

If eggs are present in a nest longer than the expected hatch/fledge date (>7d) and/or no attendant adults are present on repeated visits (eggs are typically cold or wet). Causes of abandonment may include:

Infertile eggs or dead embryos: when an attendant adult is observed incubating eggs long after (at least 7d) the calculated hatch date.

Adult mortality: a strong possibility when the nest is abandoned for no apparent reason in the late incubation or nestling stage.

Abandoned prior to egg-laying because of nest site disturbance
If a nest is suspected of abandonment, reorient eggs (e.g. all large ends toward the center) so you can reconfirm abandonment on the next visit. If the adult is still present, the eggs should be reoriented so that the large ends are now facing out. **But see special note at end of Appendix R for Phalaropes**.

**WEATHER**

A nest that is active up until a severe storm occurs. The nest may be inundated with water or covered with snow. Be sure to check these nests multiple times after a weather event because they often are quite resilient and survive.

**TRAMPLING**

A nest (and its contents) that is crushed by caribou or other large animal (e.g. Musk oxen, caribou). Supporting evidence would include animal presence in the area, tracks, droppings, and the presence of smashed eggs with contents still present.

**OBSERVER**

Any nest failure directly attributable to research work. Examples include: 1. The presence of researchers cue predator(s) to nest location (only use this if you witness predation due to your visit – do not assume a nest was depredated due to your visit if you find it empty on the next visit). 2. A nest that is inadvertently destroyed or damaged by a researcher.

## *Appendix G. Habitat classification schemes for Network sites*

**Western Alaska (Nome and Cape Krusenstern) and Eastern Arctic (Churchill)**

Adapted from Petterson 1991, with modifications from Nome site.

Terrestrial habitat near Krusenstern Lagoon was stratified according to structural components for the purpose of examining shorebird nesting ecology and determining shorebird nesting densities. Habitats were classified as follows:

**Mixed Sedge-Dwarf Ericaceous Shrub Tundra (DEST)** - Mesic lowland tundra composed of 25-35 percent dwarf (<10 cm.) and low shrub (<20 7 cm.), 30-35 percent non-tussock forming sedges, 10-20 percent tussock- forming sedges, 10-15 percent moss, 5-10 percent lichen, and 10 percent non-sedge graminoid on some plots. These islands and peninsulas are elevated about 1 meter above the slough channels, lagoons, brackish lakes, unvegetated mudflats, and margins of wet sedge meadow and other wet graminoid types that fringe them.

**Mixed Shrub~Sedge Tussock Tundra (MST)** - Upland tundra with at least 25% shrub cover and co-dominated by tussock-forming sedges. Similar to tussock tundra but with a greater shrub component. Structural composition is 25 percent Salix sp., 30 percent *Eriophorum vaginatum*, 15 percent *Carex* sp., 15 percent *Dryas* sp., and 15 percent moss and lichen. Frost heave and frost boil account for the hummocky character of this terrain.

**Willow-Sedge Shrub Tundra (WST)** - Upland tundra structurally similar to mixed shrub-sedge tussock tundra, but tufted grasses and sedges more important than tussocks. The shrub component is 25-30%Salix sp. commonly occurring in stringers along frost heaves. Tufted grasses and sedges compose 45 percent. *Eriophorum vaginatum* composes 25 percent and moss compose 5 % willow stringers and hummocks, oriented in long rows in the direction of the slope are a prominent feature in this habitat and are probably indicative of subsurface drainage patterns. Small triangular-shaped thaw puddles line occur in pockets along the willow stringers. Aerial photographs exhibit characteristics of fluvial action perhaps originating with solifluction lobes high on adjacent Dryas slopes. Soil creep retards the development of tussocks in this habitat.

**Dryas-Dwarf Shrub Tundra (DT)** - *Dryas* sp. is dominant but sparse (10-50 percent) on stony well-drained soils at windswept alpine sites. Graminoids and herbaceous perennials (e.g. Anemone, Carex, Pedicularis, Poa, Potentilla) are common but not co-dominant. Solifluction and wind erosion are the primary landscape processes in this habitat.

**Tussock Tundra (TT)** - Dominated by *Eriophorum vaginatum* (60percent), a tussock-forming cottongrass, and *Carex bigelowii* (10percent). Low birch and ericaceous shrubs constitute up to 25 percent of the cover, growing up between tussocks but usually lower than the tops of the sedges. *Ledum palustre* comprises 20 percent while lichens and mosses make up 10 percent of cover.

**Wet Sedge Meadow Tundra (WSM)** - Dominated by *Carex aguatilis* and *Eriophorum angustifolium* in standing water with occasional hummocks of moss, graminoid and dwarf woody vegetation. These wetlands associated hydrologically with lake lakes. Frost wedging is an important process in this habitat.

**Birch Ledium Empetrum (BLES)**- Dominated by birch, Ledum, Empetrum, non-tussock-forming sedges; may contain willow (BLES w/ willow) or moss patches (BLES w/ moss)
**Lowland mesic (LOM**)- area is 50-80% *Carex lyngbyei* sedge, 10-50% dwarf willow, 0-5% *Empetrum*
**Lawn mesic (LAM)** - Dominated by lawn-like grass or sedge, some Sedum and willow, moss patches
**Marsh mesic (MM)** - area is 80-100% *Carex lyngbyei* sedge, 5-20% dwarf willow, pools of water

**Arctic Coastal Plain (Barrow, Ikpikpuk, Prudhoe Bay, Canning, Colville and Mackenzie Delta) and Northeastern Canadian Arctic (East Bay and Bylot)**
From Walker et al. 1980. Geobotanical Atlas of the Prudhoe Bay Region, Alaska

Abbreviated list of Landforms Units
**HCP>0.5m**: High-centered polygons, Center-Trough Relief >0.5m
**HCP<0.5m**: HCP, C/T Relief ≤ 0.5m
**LCP>0.5m**: Low-centered Polygons, Rim-Center Relief >0.5m
**LCP<0.5m**: LCP, R/C Relief ≤ 0.5 m
**Mixed HCP/LCP**: Mixed H & LCP
**Frost boil**: Frost boil Tundra
**Strangmoor**: Strangmoor and/or Disjunct Polygon Rims
**Hummocky**: Hummocky Terrain
**Reticulate hummocks**: Reticulate-Patterned Ground
**Non-patterned**: Non-patterned Ground
**Alluvial**: Alluvial Floodplain
**Pingo**: Pingo
**Unveg Dune**: Unvegetated dunes
**Veg Dune**: Vegetated dunes
**Upland Bluff**: Upland Bluff

HCP > 0.5m: High-centered polygons, Center-Trough Relief >0.5m
High-centered polygons in the Prudhoe Bay region occur most commonly in a narrow band extending only a few tens of meters inland along streams and the shorelines of former thaw lakes. They are the product of thermokarst and/or thermal erosion in the troughs of former low-centered polygons. These processes become active when drainage of the thaw lakes or change in stream gradient permits better surface and subsurface drainage, resulting in melting of the ice and subsequent deepening of the troughs. The over-deepened (greater than 0.5 m deep and commonly 1.0 m or more) troughs permit slumping of the rim elements and a gradual topographic reversal of the polygon center. This is accompanied by a reduction in surface area of the center. HCP > 0.5m commonly has no other units included with it, although in some circumstances Mixed HCP/LCP (mixed high- and low-centered polygons) may be associated.

HCP < 0.5m: High-centered Polygons, Center-Trough Relief < 0.5m
Certain upland areas, or broadly convex interfluves, have large-diameter (5 to 10 m) polygons whose centers are slightly convex or raised with respect to the adjoining contraction crack or trough. Although the difference in height between the center and the trough may reach 0.5 m it is commonly on the order of 10 to 20 cm and sometimes much less. The central portions of these polygons may be patterned with small (25-50 cm) polygons suggestive of desiccation. The unit may include Reticulate in areas where the desiccation cracks (polygons) are the dominant landform.

LCP > 0.5m: Low-centered Polygons, Rim-Center Relief >0.5m

Low-centered polygons predominate in the unit. In plan the landform consists of polygonal cells with diameters ranging between 5 and 12 m. Each polygon is composed of three elements. The central portion, circular or weakly polygonal in shape and commonly 8 to 10 m in diameter, is surrounded by a rim 0.5 m or slightly more high and up to 1.0 m wide. Centers may contain up to 10 cm of standing water early in the summer but commonly become only moist as the thaw season progresses. The rim of one polygon is separated from that of the adjacent one by a trough whose depth ranges to 50 cm below the rim crest. The troughs mark the position of contraction cracks and ice wedges that extend to depths of 3 to 5 m. Associated landform units that in aggregate compose less than 20% of LCP > 0.5m include LCP < 0.5m, Mixed HCP/LCP, and Frost boil.

LCP < 0.5m: Low-centered Polygons, Rim-Center Relief < 0.5 m
In this extensive unit the polygons tend to be more orthogonal than those of LCP > 0.5m and relief contrast is commonly less than 0.5 m. Basin areas of these polygons are quite wet, with water at or near the surface throughout the thaw period. Landform elements commonly associated with LCP < 0.5m are Stangmoor, LCP > 0.5 m.

Mixed HCP/LCP: Mixed High- and Low-centered Polygons
This landform unit contains high-centered polygons similar to those of HCP > 0.5m and low-centered polygons undergoing topographic reversal and conversion to high-centered polygons. The unit is restricted in area and represents incomplete topographic adjustment to recently decreased base level, for example the drainage of a thaw lake or the relatively recent head ward extension of a tributary drainage.

Frost boil: Frost-Boil Tundra
Frost boil tundra reaches its maximum development in the Prudhoe Bay region along the Putuligayuk River. The landform consists of two elements; the frost boils proper and the vegetated areas between them. The boils consist of active, frost-susceptible mineral materials exposed at the surface or apparently inactive beneath a thin organic mat. The center spacing of individual boils is on the order of 2.5 m; however, areas with much closer spacing are common. Ordinarily the other landform units do not occur within Frostboil, although Reticulate may border it adjacent to the Putuligayuk River.

Strangmoor: Strangmoor and/or Disjunct Polygon Rims
This very wet landscape unit consists of string bogs (strangmoor) in which the hummock ridges (strangs) are less than 0.5 m high and are commonly discontinuous. In extreme cases they are merely an aligned series of hummocks. In some instances the strangs appear oriented normal to the hydrologic gradient and thus serve as a clue to the direction of surface and subsurface water movement. Commonly, however, they grade to low, discontinuous rims of poorly defined, large diameter polygonal cells. The landscape unit is a young terrain feature. The principal associated landscape units are Non-patterned ground and LCP < 0.5m.

Hummock: Hummocky Terrain
This unit is common on slopes greater than 6% on the sides of Pingoes and along stream bluffs. It consists of hummocks whose surface areas range between 25 and 50 cm and which extend to 20 cm or more above the adjacent inter-hummock areas. The unit commonly grades into Unit 9 as slope and angle decreases at the top of the bluff or slope. Thus the hummocks may represent the Reticulate-patterned ground landform rounded and accentuated by erosion, partly thermal and partly related to runoff from the snow banks, which form in these areas.

Reticulate: Reticulate-Patterned Ground
The reticulate landform occurs on the uplands immediately adjacent to active drainage ways and on low linear interfluves or hydrostatic forms underlain by sandy-textured mineral materials. The pattern is an intricate arrangement of slightly convex, small diameter polygons (less than 1.0 m), commonly with a hummocky micro relief (less than 15 cm). As topographic slope steepens toward an adjacent drainage the reticulate landform grades in to the large hummocks of Hummocky Terrain. Away from the drainage and marginal to the wetter tundra elements, especially along the Putuligayuk River, Reticulate-Patterned may include small amounts of Frost boil and HCP < 0.5m.

Non-patterned: Non-patterned Ground
Areas designated as non-patterned ground occur within the basins of recently drained thaw lakes and surrounding shallow, active thaw lakes. Such areas are wet, commonly with standing water throughout the thaw period. They are considered to represent some of the youngest areas in the landscape. Randomly distributed hummocks or short non-aligned hummock ridges, a few tens of centimeters in height, may characterize the surface in some localities. Low relief, low-centered polygons of LCP < 0.5m may compose up to 20% of this unit.

Alluvial: Alluvial Floodplain
This unit contains the river floodplains. Micro topographic expression is commonly lacking or consists of undulating scour pits and abandoned stream channels and bars or the beds of intermittently flowing streams.

Pingo: Pingo
Pingoes are probably the most distinctive and least extensive of the landform units recognized at Prudhoe Bay. In the area covered by this atlas the features are conical to slightly elliptical in form, with basal diameters of several tens to several hundreds of meters. They extend up to 15 m above the surrounding tundra. Their summits may be cracked or may have a central depression due to collapse as the ice core melts. Although the upper portions of the steep side slopes may be severely wind-eroded, the lower portions display the hummock forms of Hummocky Terrain. Pingoes are common features in drained lake basins.

Unveg Dune: Unvegetated (Sand) Dunes
Although they do not appear on the main mapped area in this atlas, sand dunes form a unique landform element in the area just west of the delta of the Sagavanirktok River. Dunes consist of sand ridges 1 to 2 m high extending leeward from stabilized or partly stabilized coppice-like dunes or dune remnants. Sandy areas between ridges are mostly devoid of vegetation and commonly moist. In some areas polygon terrain similar to LCP < 0.5m can be seen underlying areas recently or thinly covered by the sands. .Note: these are active sand dunes too unstable for vegetation establishment with <30% vegetation cover.

Upland: Upland Bluff
As described from the Canning Delta site; these areas consisted of ridges or low-sloping bluffs that extended 1-2 m above the surrounding tundra. Sites were typically well-drained and consisted mainly of vegetation types (from Walker et al. 1980). U3 (moist Eriophorum vaginatum, Dryas integrifolia, Tomenthypnum nitens, Thamnolia vermicularis graminoid meadow) and U4 (moist Carex aquatilis, Dryas integrifolia, Tomenthypnum nitens, Salix arctica graminoid meadow).

Veg Dunes: Vegetated Dunes

Inactive and stabilized dunes with ≥30% vegetation cover. Generally dominated by dwarf and upland shrubs such as Dryas or Cassiope with associated forbs and low-growing Salix. Dwarf scrub tundra on upland ridges, stabilized sand dunes and river terraces dominated by Dryas integrifolia or Cassiope tetragona. Upland Dryas sites typically are dry and sandy with deep thaw depths (>1.0 m), common associated species include Salix glauca, S. reticulata, Arctostaphylos alpina, Arctagrostis latifolia, Thamnolia vermicularis, and Cetraria cuculata. Riverine Dryas sites occur on well-drained, sandy river terraces, co-dominant species often include Equisetum variegatum and Salix reticulata, with S. lanata richardsonii, Arctostaphylos rubra, Oxytropis deflexa, Tomentypnum nitens, and Thamnolia vermicularis as associated species. Cassiope tetragona is found on slightly moister sites such as banks of thaw basins, riverbanks, and banks of older, well-stabilized dunes. On intermediate soils Dryas integrifolia may be co-dominant. Species found in association with Cassiope include S. phlebophylla, Salix reticulata, Vaccinium vitis-idaea, Carex bigelowii, Hierochloe alpina, and Arctagrostis latifolia. Cryptogams present include crustose lichens, Hylocomium splendens, Dicranum sp., Tomentypnum nitens, and Rhytidium rugosum. All sites have a wide variety of forbs.

Abbreviated list of Vegetation Units

*Generally B = DRIEST U = MOISTER M = WET E = EMERGENT*

| **Codes** | **Description** |
|---|---|
| **B1** | driest, most exposed: sides of pingos, centers of high-centered polygons |
| **B2** | less exposed to wind than B1; otherwise similar |
| **B3** | tops of frost boils |
| **B14** | dry, early-thawing snowbanks with hummocky terrain |
| | |
| **U1** | polygons rims and aligned strangmoor in acidic tundra; LICHEN |
| **U2** | well-drained upland sites; tussocks < 20 cm & dense sedge cover; LICHEN |
| **U3** | well-drained upland sites with slightly high-centered polygons; LICHEN |
| **U4** | moister upland sites, centers of low polygons or poly rims; NO LICHEN |
| **U6** | well-drained snowbanks with *Cassiope tetragona* |
| **U7** | late-thawing snowbanks with *Salix* present |
| **U8** | stream banks or lake margins with *Salix* and *Carex* present |
| **U9** | upland stream banks swept by spring flood |
| **U10** | pingo tops, bird mounds, animal dens – graminoid meadow |
| | |
| **M1** | wet micro sites in acidic tundra with aligned strangmoor; NO LICHEN/*Salix* |
| **M2** | wet polygon center and troughs, lake margins; NO LICHEN |
| **M4** | very wet polygon centers, drained lakes, lake margins; NO LICHEN |
| **M5** | moist stream banks; *Carex* and *Salix* present |
| | |
| **E1** | very wet: water to about 30 cm; *Carex* present |
| **E2** | very wet: water to about 100 cm: *Arctophila* present |

More complete descriptions of these vegetation units are available in Walker et al. (1980) but the traits listed above focus on the most relevant features of each unit.

## *Appendix H. Potential adult and egg predators in the Arctic and lemmings.*

| ***Potential nest predator**** | ***Species code*** |
|---|---|
| ***AVIAN*** | |
| Glaucous Gull *(Larus hyperboreus)* | GLGU |
| Pomarine Jaeger *(Stercorarius pomarinus)* | POJA |
| Parasitic Jaeger *(Stercorarius parasiticus)* | PAJA |
| Long-tailed Jaeger *(Stercorarius longicaudus)* | LTJA |
| Common Raven *(Corvus corax)* | CORA |
| Ruddy Turnstone *(Arenaria interpres)* | RUTU |
| Sandhill Crane *(Grus canadensis)* | SACR |
| Golden Eagle *(Aquila chrysaetos)* | GOEA |
| Snowy Owl *(Nyctea scandiaca)* | SNOW |
| Peregrine Falcon (*Falco peregrinus*) | PEFA |
| Northern Harrier (*Circus cyaneus*) | NOHA |
| Rough-legged Hawk (*Buteo lagopus*) | RLHA |
| Short-eared Owl (*Asio flammeus*) | SEOW |
| | |
| ***MAMMALIAN*** | |
| Arctic fox (*Alopex lagopus*) | ARFO |
| Red fox (*Vulpes vulpes*) | REFO |
| Brown (Grizzly) bear *(Ursus arctos)* | BRBE |
| Wolverine (*Gulo gulo*) | WOLV |
| Polar bear *(Ursus maritimus)* | POBE |
| Short-tailed weasel *(Mustela erminea)* | STWE |
| Least weasel *(Mustela nivalis)* | LEWE |
| Arctic ground squirrel (*Spermophilus parryii*) | AGSQ |
| | |
| Brown lemming (*Lemmus trinucronatus*) | BRLE |
| Collared lemming (*Dicrostonyx groenlandicus*) | COLE |

* Species that have been observed or suspected of depredating nesting birds, eggs, or young at tundra-nesting bird nests. We do not record Sabine's Gulls and Arctic Terns as potential nest predators because they are only believed to very rarely depredate nests.

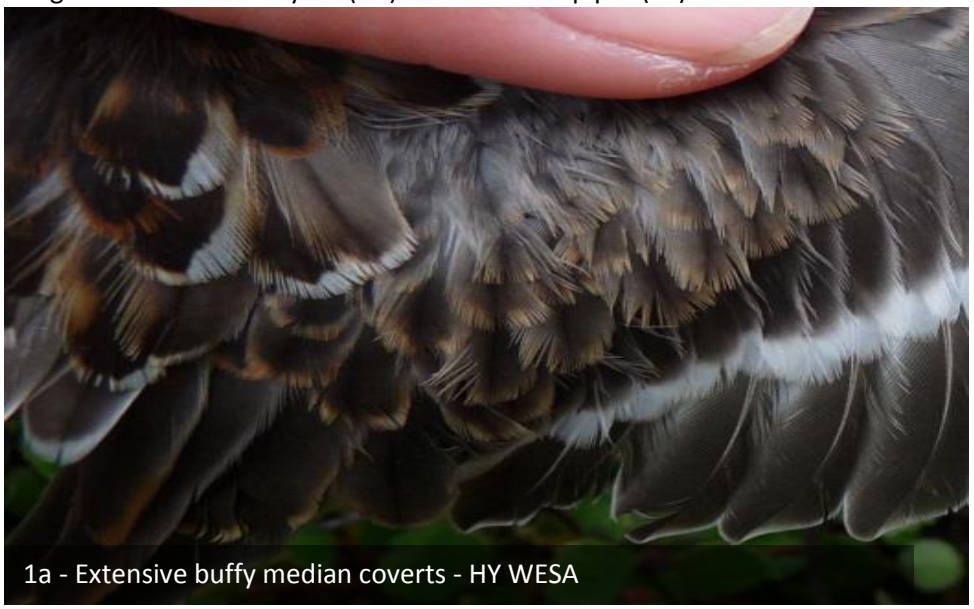
## *Appendix I. Focal species ageing guide by species*

The information presented below is a reference guide for ageing Calidris sandpipers. For more extensive resources about molt and age classification see list of additional resources at the end of this guide. We advise using this reference, in addition to training with someone who has extensive banding experience and ageing. Please take pictures of **captured bird wings according to instructions in banding section. This creates a permanent record of the banded bird and will be used to verify and assess age in a consistent manner**. This guide was developed by Samantha Franks and River Gates.

**Plumage criteria that can be used to age Calidris sandpipers: Semipalmated and Western Sandpipers and Dunlin.**

| | **Age** | | |
|---|---|---|---|
| **Plumage criteria** | **HY-juvenile** | **SY-yearling** | **ASY-adult >1 year old** |
| **Inner median coverts** | Extensive buffy or reddish edging on all coverts, dark centers (WESA: Fig 1a, DUNL: Fig 1b). | All heavily worn, a few with slight buffy or reddish edges (SESA: Fig 2a, WESA: 3a & b, DUNL: 4a). | Slightly worn, No distinct buffy or reddish-edged coverts apparent, coverts are white-edged and (SESA: Fig 5a, WESA: Fig 5c, DUNL: Fig 6a) |
| | Calidris HY plumages are all very similar in appearance. | Inner coverts may have darker centers with less distinction in color between shaft and feather center | Inner coverts may have lighter centers with more distinction in color between shaft and feather center |
| **Primary and greater coverts** | Fresh white tips, smooth edges | Extremely worn white tips, frayed edges | Moderate wear of white tips, more smoothly edged |
| **Flight feather wear** | Very fresh, dark grey feathers with no wear at tips and little fraying along edges (WESA: Fig 1c) | Lots of wear at tips of outer primaries, frayed primary edges (SESA: Fig 2b, WESA: Fig 3c) | Minimal to moderate wear at tips of outer primaries, more smoothly edged primaries (SESA: Fig 5b, WESA: Fig 5d) |
| **Primary molt** | n/a | May exhibit partial primary molt, where outer primaries (~p6-10) have been replaced (SESA: Fig 2c) | n/a |

Figures 1a – c. Hatch year (HY) Western Sandpiper (1a) and Dunlin both ~14 days old (1b). Photos by S. Franks and J. Choi



1a - Extensive buffy median coverts - HY WESA

1b - Extensive buffy median coverts – HY DUNL

1c - Fresh primary tips – HY WESA

Figure 2a-c. A second year (SY) Semipalmated Sandpiper.

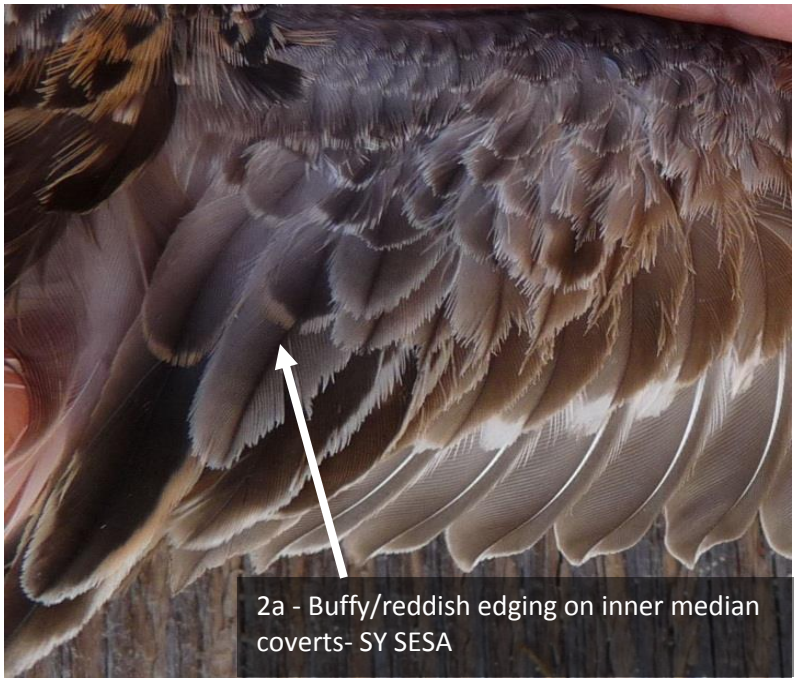
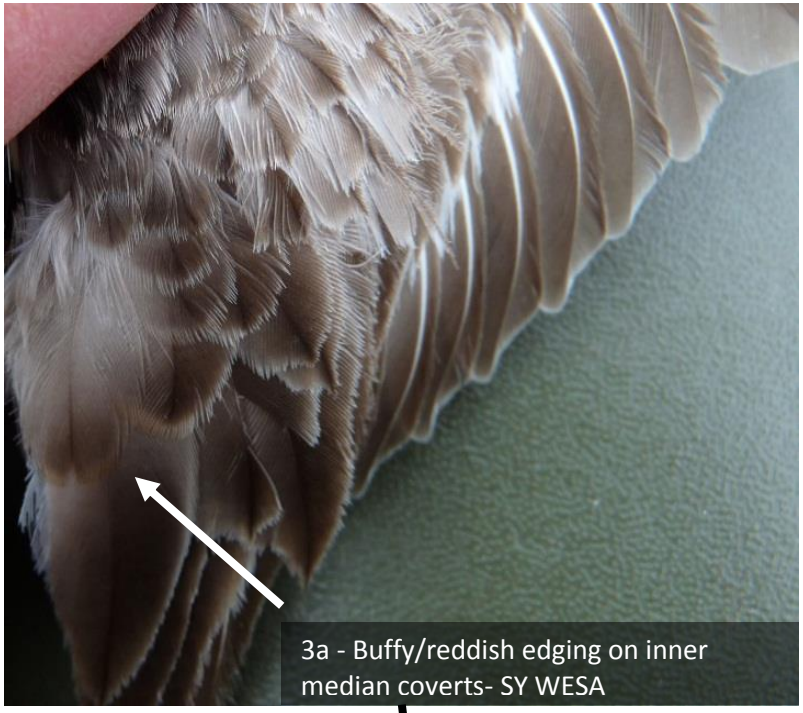
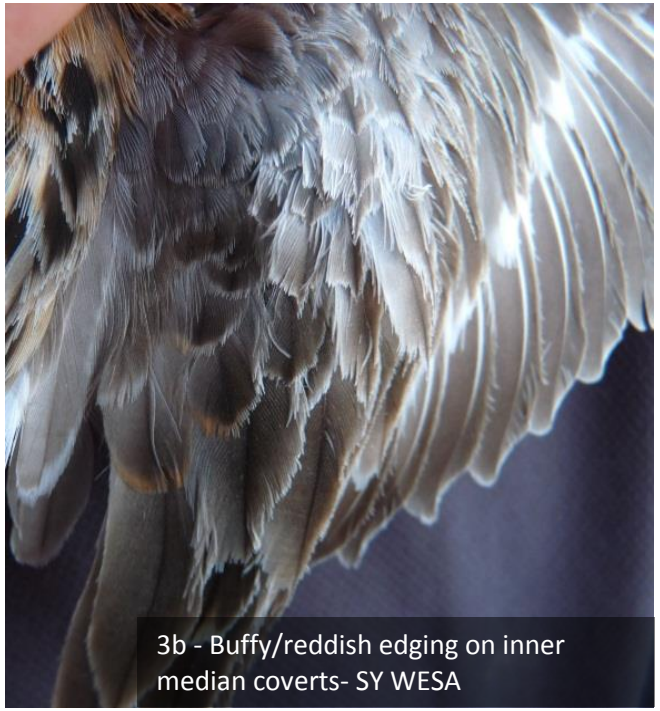




Figure 3a-b. A second year (SY) Western Sandpiper.



3a - Buffy/reddish edging on inner median coverts- SY WESA



3b - Buffy/reddish edging on inner median coverts- SY WESA

3c - Extremely worn primary tips- SY WESA

Figure 4a: A second year (SY) Dunlin. Photos by J. Choi/USFWS.

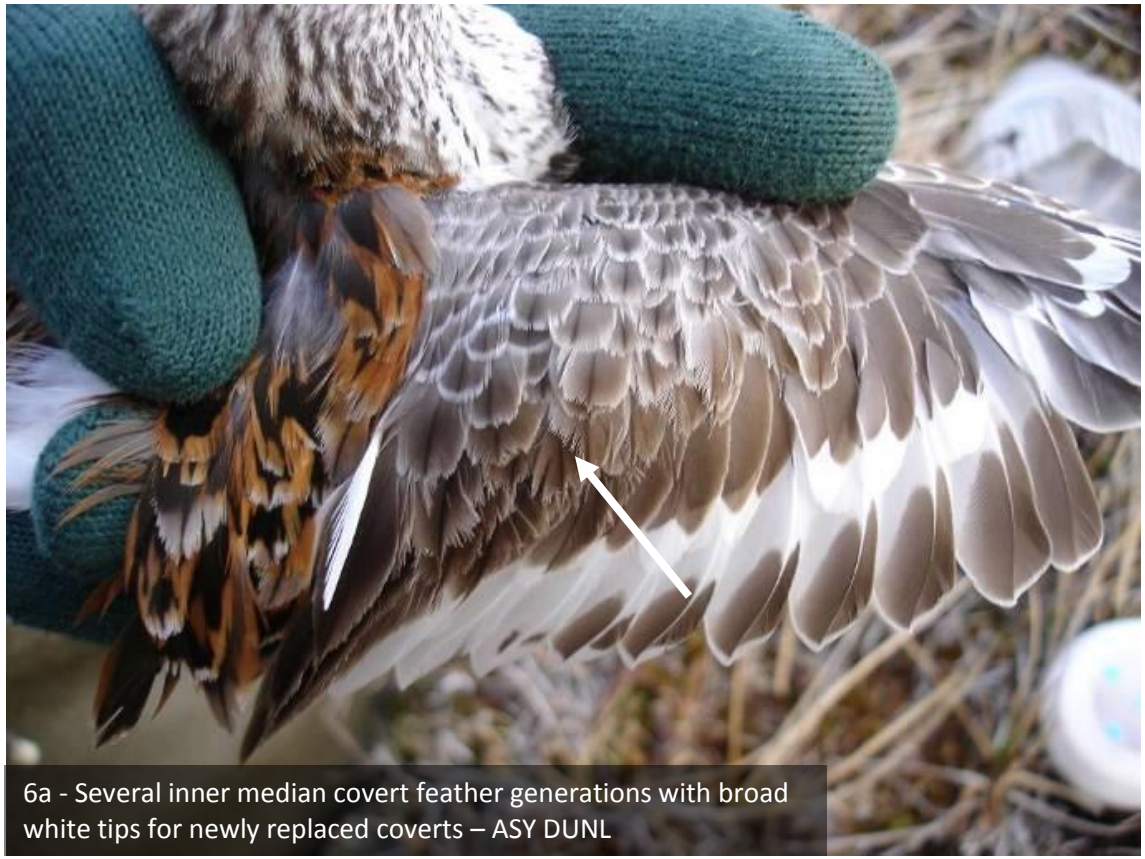
Figure 5a-b. An after second year (ASY) Semipalmated Sandpiper, and c – d an after second year (ASY) Western Sandpiper

5a - White-edged inner median coverts- ASY SESA

5b - Minimally to moderately worn tips - ASY SESA

5c - White-edged inner median coverts-ASY WESA

5d - Minimally to moderately worn tips- ASY WESA



6a - Several inner median covert feather generations with broad white tips for newly replaced coverts – ASY DUNL

*Additional resources:*

Pyle, P. 2008. Identification Guide to North American Birds. Part II. Slate Creek Press, Point Reyes Station, CA

Prater, A.J., Marchant, J.H. and J. Vuorinen 1977. Guide to identification and ageing of Holarctic waders. British Trust For Ornithology Guide 17.

Gratto-Trevor, C. L. 2004. The North American Bander's Manual for banding shorebirds. North American Banding Council

## *Appendix J. Sinking and floating egg float tables for select shorebird species*

There is a spreadsheet within the banding excel electronic file that can be used to estimate days to hatch that incorporates equations provided in Liebezeit et al. (2007).

**SINKING EGGS** DTH = "days to hatch" | *From Liebezeit et al. 2007*

| **AMGP** | | **BBPL** | | **BBSA** | | **DUNL** | | **LBDO** | |
|---|---|---|---|---|---|---|---|---|---|
| ***Angle*** | ***DTH*** | ***Angle*** | ***DTH*** | ***Angle*** | ***DTH*** | ***Angle*** | ***DTH*** | ***Angle*** | ***DTH*** |
| 21 | 25.8 | 21 | 25.3 | 21 | 27.8 | 21 | 21.3 | 21 | 22.4 |
| 25 | 23.4 | 25 | 23.1 | 25 | 24.2 | 25 | 19.8 | 25 | 20.0 |
| 30 | 22.2 | 30 | 22.1 | 30 | 22.6 | 30 | 19.1 | 30 | 18.9 |
| 35 | 21.5 | 35 | 21.5 | 35 | 21.5 | 35 | 18.6 | 35 | 18.2 |
| 40 | 20.9 | 40 | 21.0 | 40 | 20.7 | 40 | 18.3 | 40 | 17.6 |
| 45 | 20.4 | 45 | 20.5 | 45 | 20.0 | 45 | 18.0 | 45 | 17.2 |
| 50 | 20.0 | 50 | 20.1 | 50 | 19.3 | 50 | 17.7 | 50 | 16.7 |
| 55 | 19.6 | 55 | 19.8 | 55 | 18.7 | 55 | 17.5 | 55 | 16.3 |
| 60 | 19.1 | 60 | 19.4 | 60 | 18.1 | 60 | 17.2 | 60 | 15.9 |
| 65 | 18.7 | 65 | 19.0 | 65 | 17.5 | 65 | 16.9 | 65 | 15.5 |
| 70 | 18.2 | 70 | 18.6 | 70 | 16.8 | 70 | 16.6 | 70 | 15.0 |
| 75 | 17.6 | 75 | 18.1 | 75 | 16.0 | 75 | 16.3 | 75 | 14.4 |
| 80 | 16.9 | 80 | 17.4 | 80 | 14.9 | 80 | 15.8 | 80 | 13.7 |
| 85 | 15.8 | 85 | 16.4 | 85 | 13.2 | 85 | 15.1 | 85 | 12.6 |
| 89 | 13.3 | 89 | 14.2 | 89 | 9.7 | 89 | 13.6 | 89 | 10.2 |
| **PESA** | | **RNPH** | | **REPH** | | **RUTU** | | **SESA** | |
| ***Angle*** | ***DTH*** | ***Angle*** | ***DTH*** | ***Angle*** | ***DTH*** | ***Angle*** | ***DTH*** | ***Angle*** | ***DTH*** |
| 21 | 20.9 | 21 | 21.2 | 21 | 18.1 | 21 | 21.5 | 21 | 20.2 |
| 25 | 19.6 | 25 | 19.4 | 25 | 17.0 | 25 | 19.4 | 25 | 18.6 |
| 30 | 18.9 | 30 | 18.5 | 30 | 16.5 | 30 | 18.4 | 30 | 17.9 |
| 35 | 18.5 | 35 | 18.0 | 35 | 16.1 | 35 | 17.8 | 35 | 17.5 |
| 40 | 18.2 | 40 | 17.6 | 40 | 15.9 | 40 | 17.3 | 40 | 17.1 |
| 45 | 17.9 | 45 | 17.2 | 45 | 15.6 | 45 | 16.9 | 45 | 16.8 |
| 50 | 17.7 | 50 | 16.9 | 50 | 15.4 | 50 | 16.5 | 50 | 16.5 |
| 55 | 17.5 | 55 | 16.6 | 55 | 15.2 | 55 | 16.2 | 55 | 16.2 |
| 60 | 17.2 | 60 | 16.3 | 60 | 15.0 | 60 | 15.8 | 60 | 16.0 |
| 65 | 17.0 | 65 | 16.0 | 65 | 14.8 | 65 | 15.4 | 65 | 15.7 |
| 70 | 16.7 | 70 | 15.6 | 70 | 14.6 | 70 | 15.0 | 70 | 15.4 |
| 75 | 16.4 | 75 | 15.2 | 75 | 14.3 | 75 | 14.6 | 75 | 15.0 |
| 80 | 16.0 | 80 | 14.7 | 80 | 14.0 | 80 | 13.9 | 80 | 14.6 |
| 85 | 15.4 | 85 | 13.8 | 85 | 13.5 | 85 | 13.0 | 85 | 13.9 |
| 89 | 14.0 | 89 | 12.0 | 89 | 12.3 | 89 | 10.9 | 89 | 12.3 |

| **STSA** | | **Other shorebirds** | |
|---|---|---|---|
| ***Angle*** | ***DTH*** | ***Angle*** | ***% of incubation complete*** |
| 21 | 17.6 | 21 | 0.016 |
| 25 | 16.9 | 25 | 0.075 |
| 30 | 16.5 | 30 | 0.118 |
| 35 | 16.3 | 35 | 0.145 |
| 40 | 16.2 | 40 | 0.166 |
| 45 | 16.0 | 45 | 0.184 |
| 50 | 15.9 | 50 | 0.200 |
| 55 | 15.8 | 55 | 0.216 |
| 60 | 15.7 | 60 | 0.232 |
| 65 | 15.5 | 65 | 0.248 |
| 70 | 15.4 | 70 | 0.266 |
| 75 | 15.2 | 75 | 0.287 |
| 80 | 15.0 | 80 | 0.314 |
| 85 | 14.7 | 85 | 0.356 |
| 89 | 14.0 | 89 | 0.448 |

**FLOATING EGGS DTH = "days to hatch"**

| AMGP | | | BBPL | | | BBSA | | | DUNL | | |
|---|---|---|---|---|---|---|---|---|---|---|---|
| *Angle* | *height* | *DTH* | *Angle* | *height* | *DTH* | *Angle* | *height* | *DTH* | *Angle* | *height* | *DTH* |
| 90 | 0 | 14.0 | 90 | 0 | 15.8 | 90 | 0 | 15.9 | 90 | 0 | 12.8 |
| 90 | 1 | 12.6 | 90 | 1 | 12.8 | 90 | 1 | 13.7 | 90 | 1 | 11.2 |
| 90 | 2 | 11.2 | 90 | 2 | 9.9 | 90 | 2 | 11.5 | 90 | 2 | 9.5 |
| 90 | 3 | 9.8 | 90 | 3 | 7.0 | 90 | 3 | 9.4 | 90 | 3 | 7.9 |
| 90 | 4 | 8.4 | 90 | 4 | 4.0 | 90 | 4 | 7.2 | 90 | 4 | 6.3 |
| 90 | 5 | 7.1 | 90 | 5 | 1.1 | 90 | 5 | 5.0 | 90 | 5 | 4.7 |
| 90 | 6 | 5.7 | 90 | 6 | 1.9 | 90 | 6 | 2.8 | 90 | 6 | 3.1 |
| 90 | 7 | 4.3 | | | | 90 | 7 | 0.6 | 90 | 7 | 1.5 |
| 90 | 8 | 2.9 | 80 | 0 | 16.0 | | | | 90 | 8 | 0.1 |
| | | | 80 | 1 | 13.1 | 80 | 0 | 15.1 | | | |
| 80 | 0 | 13.6 | 80 | 2 | 10.1 | 80 | 1 | 12.9 | 80 | 0 | 11.7 |
| 80 | 1 | 12.2 | 80 | 3 | 7.2 | 80 | 2 | 10.7 | 80 | 1 | 10.1 |
| 80 | 2 | 10.8 | 80 | 4 | 4.2 | 80 | 3 | 8.5 | 80 | 2 | 8.5 |
| 80 | 3 | 9.4 | 80 | 5 | 1.3 | 80 | 4 | 6.3 | 80 | 3 | 6.9 |
| 80 | 4 | 8.0 | 80 | 6 | 1.6 | 80 | 5 | 4.1 | 80 | 4 | 5.3 |
| 80 | 5 | 6.6 | | | | 80 | 6 | 1.9 | 80 | 5 | 3.7 |
| 80 | 6 | 5.2 | 70 | 0 | 16.2 | 80 | 7 | 0.3 | 80 | 6 | 2.1 |
| 80 | 7 | 3.9 | 70 | 1 | 13.3 | | | | 80 | 7 | 0.5 |
| 80 | 8 | 2.5 | 70 | 2 | 10.3 | 70 | 0 | 14.2 | | | |
| 80 | 9 | 1.1 | 70 | 3 | 7.4 | 70 | 1 | 12.0 | 70 | 0 | 10.7 |
| 80 | 10 | 0.3 | 70 | 4 | 4.5 | 70 | 2 | 9.8 | 70 | 1 | 9.1 |
| | | | 70 | 5 | 1.5 | 70 | 3 | 7.7 | 70 | 2 | 7.5 |
| 70 | 0 | 13.2 | 70 | 6 | 1.4 | 70 | 4 | 5.5 | 70 | 3 | 5.9 |
| 70 | 1 | 11.8 | | | | 70 | 5 | 3.3 | 70 | 4 | 4.3 |
| 70 | 2 | 10.4 | | | | 70 | 6 | 1.1 | 70 | 5 | 2.7 |
| 70 | 3 | 9.0 | | | | | | | 70 | 6 | 1.1 |
| 70 | 4 | 7.6 | | | | | | | 70 | 7 | 0.6 |
| 70 | 5 | 6.2 | | | | | | | | | |
| 70 | 6 | 4.8 | | | | | | | | | |
| 70 | 7 | 3.4 | | | | | | | | | |
| 70 | 8 | 2.1 | | | | | | | | | |
| 70 | 9 | 0.7 | | | | | | | | | |

| **LBDO** | | | **PESA** | | | **RNPH** | | | **REPH** | | |
|---|---|---|---|---|---|---|---|---|---|---|---|
| ***Angle*** | ***height*** | ***DTH*** | ***Angle*** | ***height*** | ***DTH*** | ***Angle*** | ***height*** | ***DTH*** | ***Angle*** | ***height*** | ***DTH*** |
| 90 | 0 | 15.4 | 90 | 0 | 13.0 | 90 | 0 | 11.6 | 90 | 0 | 11.1 |
| 90 | 1 | 14.4 | 90 | 1 | 11.8 | 90 | 1 | 9.1 | 90 | 1 | 9.8 |
| 90 | 2 | 13.4 | 90 | 2 | 10.5 | 90 | 2 | 6.6 | 90 | 2 | 8.6 |
| 90 | 3 | 12.5 | 90 | 3 | 9.3 | 90 | 3 | 4.0 | 90 | 3 | 7.3 |
| 90 | 4 | 11.5 | 90 | 4 | 8.1 | 90 | 4 | 1.5 | 90 | 4 | 6.1 |
| 90 | 5 | 10.5 | 90 | 5 | 6.8 | | | | 90 | 5 | 4.8 |
| | | | | | | 80 | 0 | 11.7 | | | |
| 80 | 0 | 12.0 | 80 | 0 | 12.4 | 80 | 1 | 9.2 | 80 | 0 | 10.1 |
| 80 | 1 | 11.0 | 80 | 1 | 11.1 | 80 | 2 | 6.7 | 80 | 1 | 8.9 |
| 80 | 2 | 10.0 | 80 | 2 | 9.9 | 80 | 3 | 4.2 | 80 | 2 | 7.6 |
| 80 | 3 | 9.0 | 80 | 3 | 8.7 | 80 | 4 | 1.7 | 80 | 3 | 6.4 |
| 80 | 4 | 8.0 | 80 | 4 | 7.4 | 80 | 5 | 0.8 | 80 | 4 | 5.1 |
| 80 | 5 | 7.0 | 80 | 5 | 6.2 | | | | 80 | 5 | 3.8 |
| 80 | 6 | 6.0 | 80 | 6 | 5.0 | 70 | 0 | 11.9 | 80 | 6 | 2.6 |
| 80 | 7 | 5.0 | 80 | 7 | 3.7 | 70 | 1 | 9.4 | 80 | 7 | 1.3 |
| 80 | 8 | 4.0 | 80 | 8 | 2.5 | 70 | 2 | 6.9 | 80 | 8 | 0.1 |
| 80 | 9 | 3.0 | 80 | 9 | 1.3 | 70 | 3 | 4.4 | | | |
| 80 | 10 | 2.0 | 80 | 10 | 0.0 | 70 | 4 | 1.9 | 70 | 0 | 9.2 |
| | | | | | | 70 | 5 | 0.7 | 70 | 1 | 7.9 |
| 70 | 0 | 8.6 | 70 | 0 | 11.7 | | | | 70 | 2 | 6.7 |
| 70 | 1 | 7.6 | 70 | 1 | 10.5 | | | | 70 | 3 | 5.4 |
| 70 | 2 | 6.6 | 70 | 2 | 9.3 | | | | 70 | 4 | 4.2 |
| 70 | 3 | 5.6 | 70 | 3 | 8.0 | | | | 70 | 5 | 2.9 |
| 70 | 4 | 4.6 | 70 | 4 | 6.8 | | | | 70 | 6 | 1.6 |
| 70 | 5 | 3.6 | 70 | 5 | 5.6 | | | | 70 | 7 | 0.4 |
| 70 | 6 | 2.6 | 70 | 6 | 4.3 | | | | 70 | 8 | 0.9 |
| 70 | 7 | 1.6 | 70 | 7 | 3.1 | | | | | | |
| 70 | 8 | 0.6 | 70 | 8 | 1.9 | | | | | | |

**FLOATING EGGS** DTH = "days to hatch"

| **RUTU** | | | **SESA** | | | **STSA** | | | **Other Shorebird*** | | |
|---|---|---|---|---|---|---|---|---|---|---|---|
| ***Angle*** | ***height*** | ***DTH*** | ***Angle*** | ***height*** | ***DTH*** | ***Angle*** | ***height*** | ***DTH*** | ***Angle*** | ***height*** | ***% of incubation complete*** |
| 90 | 0 | 13.4 | 90 | 0 | 11.5 | 90 | 0 | 11.2 | 90 | 0 | 0.42 |
| 90 | 1 | 10.0 | 90 | 1 | 10.2 | 90 | 1 | 10.4 | 90 | 1 | 0.48 |
| 90 | 2 | 6.7 | 90 | 2 | 8.9 | 90 | 2 | 9.5 | 90 | 2 | 0.55 |
| 90 | 3 | 3.3 | 90 | 3 | 7.5 | 90 | 3 | 8.7 | 90 | 3 | 0.62 |
| 90 | 4 | 0.0 | 90 | 4 | 6.2 | 90 | 4 | 7.8 | 90 | 4 | 0.68 |
| | | | 90 | 5 | 4.9 | 90 | 5 | 7.0 | 90 | 5 | 0.75 |
| 80 | 0 | 13.8 | 90 | 6 | 3.5 | | | | 90 | 6 | 0.82 |
| 80 | 1 | 10.4 | 90 | 7 | 2.2 | 80 | 0 | 10.5 | 90 | 7 | 0.89 |
| 80 | 2 | 7.0 | 90 | 8 | 0.9 | 80 | 1 | 9.7 | 90 | 8 | 0.95 |
| 80 | 3 | 3.7 | | | | 80 | 2 | 8.8 | | | |
| 80 | 4 | 0.3 | 80 | 0 | 10.8 | 80 | 3 | 8.0 | 80 | 0 | 0.46 |
| | | | 80 | 1 | 9.5 | 80 | 4 | 7.1 | 80 | 1 | 0.53 |
| 70 | 0 | 14.1 | 80 | 2 | 8.1 | 80 | 5 | 6.3 | 80 | 2 | 0.59 |
| 70 | 1 | 10.7 | 80 | 3 | 6.8 | 80 | 6 | 5.4 | 80 | 3 | 0.66 |
| 70 | 2 | 7.4 | 80 | 4 | 5.5 | 80 | 7 | 4.6 | 80 | 4 | 0.73 |
| 70 | 3 | 4.0 | 80 | 5 | 4.1 | 80 | 8 | 3.7 | 80 | 5 | 0.79 |
| 70 | 4 | 0.6 | 80 | 6 | 2.8 | 80 | 9 | 2.9 | 80 | 6 | 0.86 |
| | | | 80 | 7 | 1.5 | 80 | 10 | 2.0 | 80 | 7 | 0.93 |
| | | | 80 | 8 | 0.1 | | | | 80 | 8 | 0.99 |
| | | | | | | 70 | 0 | 9.8 | | | |
| | | | 70 | 0 | 10.0 | 70 | 1 | 9.0 | 70 | 0 | 0.50 |
| | | | 70 | 1 | 8.7 | 70 | 2 | 8.1 | 70 | 1 | 0.57 |
| | | | 70 | 2 | 7.4 | 70 | 3 | 7.3 | 70 | 2 | 0.63 |
| | | | 70 | 3 | 6.1 | 70 | 4 | 6.4 | 70 | 3 | 0.70 |
| | | | 70 | 4 | 4.7 | 70 | 5 | 5.6 | 70 | 4 | 0.77 |
| | | | 70 | 5 | 3.4 | 70 | 6 | 4.7 | 70 | 5 | 0.84 |
| | | | 70 | 6 | 2.1 | 70 | 7 | 3.8 | 70 | 6 | 0.90 |
| | | | 70 | 7 | 0.7 | 70 | 8 | 3.0 | 70 | 7 | 0.97 |
| | | | | | | 70 | 9 | 2.1 | | | |
| | | | | | | 70 | 10 | 1.3 | | | |

* To calculate the "% of incubation complete" for species for which we do not have species-specific float tables, use the "other shorebird" float table.

For example: You discover a Bar-tailed Godwit nest and float the eggs. The eggs are floating at the water surface at an angle of 80° and the egg is exposed 2 mm above the water line.

<u>% of incubation complete</u> = 0.59 (from "other shorebird" table) x 21 (mean incubation length for BTGO) = 12.4 days old. The eggs will hatch in approximately (21 – 12.4) = 8.6 days

## *Appendix K. Predator point counts 2010*

***For sites continuing to conduct independent predator point counts.*** **Not currently part of ASDN methods.** These data will be used to index the abundance and activity level of potential predators at each study site and will be used in analyses to determine the relationship between predator numbers, and adult and nest survivorship. We will conduct predator surveys once a week for intensive sites or a minimum of three times per season (early, mid and late) for minimum sites from the beginning of nesting to the end of hatch (or through brood-rearing for situations where brood survival is being monitored). The following protocol will be used during each survey day (adapted from Liebezeit 2009).

- Conduct a 10-minute "point count" at a minimum of 10 different locations within the study area. If you have plots, you can conduct 10-minute counts within each plot (spaced at least 200m apart). The goal is to collect enough observations of dominant predators to be able to use the DISTANCE program (ideally 50 observations of each species across the entire season). Record GPS locations for these points on the meta database for the camp.
- To identify predators during point counts use binoculars (8 x 42 or 10 x 40).
- Timed surveys will be conducted weekly (intensive approach) or 3 times (less intensive) during the season: 1st: during early nesting (5 June to 20 June), 2nd: mid-incubation: (21 June to 5 July), 3rd: late-nesting (6 July to 25 July).
- Wait at least 10 minutes (longer if possible) between individual point counts and conduct the consecutive counts at a stake at least 200m from the previous one.
- During the "point count" the observer should scan the surrounding terrain for any visual or auditory detection of potential nest predators and record any predators seen within a 300m radius of the point count stake you are at.
- Record the predator species and the distance to the predator upon the initial sighting.
- Estimate the distance to the predator within the nearest 5-10m. If the predator is within 50-100m of you, try to estimate its distance to the nearest meter.
- To estimate distances use rangefinders, pace the distance on foot, or use adjacent centerline plot markers as a guide (they are 50m from each other). You will not be able to get a distance on a moving predator with the rangefinder, instead, obtain a distance on a patch of tundra that is below a flying bird or where a moving fox was seen.

Additional guidelines:

1. Do **not** perform predator counts during rope drag visits or at times when more than one observer is on a study plot.
2. Take effort not to re-count the same individual predators.
3. Do not wait at a point for a "settling period" before starting a count. Start the count right away.
4. Fill out a nest record form for all avian and mammalian predator nests within the study area, especially Snowy Owl and Pomarine Jaegers. Note contents of nest (e.g., number of eggs or chicks) and adult behavior.

Predator Point Count Data recorded
(see ASDN V1 protocol)

1. Observer name (first initial and full last name)
2. Plot ID
3. Date – dd-mm-yr
4. Time of arrival on plot/search area
5. Time of departure from plot/search area

6. Record start and end time for each timed census count at each survey point; record GPS location of these predator point counts.
7. Write species (including number of individuals) detected within 300m of the survey point. See **Appendix H** for list of potential predators and their codes
8. Record perpendicular distance (meter) to predator.
9. Comment on any important observations:
    a. Hunting behavior observed
    b. Interactions among predators or with other species
    c. Observation of nest predation
    d. Possibility you already recorded this same predator during this survey.
    e. Plumage variations that may aid in identifying individuals (e.g. dark-morph versus light-morph jaegers)
10. Observation of nesting predators should be recorded – if a new nest is detected be sure to fill out a nest card so that the presence of this nest (and its GPS location) is present for later predator abundance estimates.

## *Appendix L. Building and deploying terrestrial invertebrate traps*

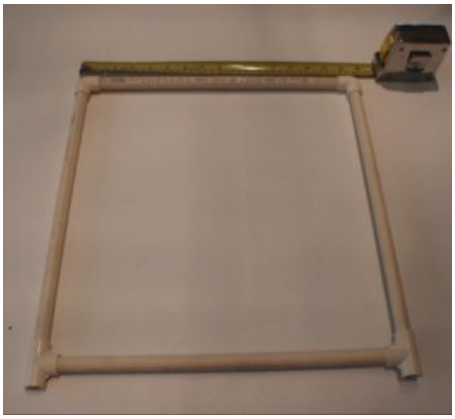
**A step by step method to make Bylot Arthropod Traps (i.e., modified Malaise trap)**
*by Jean-François Lamarre and Elise Bolduc, based on previous experience with modified Malaise traps.*

Material needed per trap (40 cm X 40 cm)

- 2 90° elbow – for ½ inch CPVC tubing
- 2 T elbow – for ½ inch CPVC tubing
- 4 36 cm long ½' inch CPVC tubing
- 2 10 inch nails (to anchor down trap)
- 1 Rubbermaid drawer organizer (15' X 3' X 2'---L3-2917-RO-WHT)
  http://www.rubbermaid.com/Category/Pages/ProductDetail.aspx?Prod_ID=RP091340
  http://www.containerstore.com/shop/office/deskAccessories/drawerOrganizers?productId=10000149
- 1 Screen 45 cm X 92 cm(out of a roll of 92 cm wide)
- 2 rebar (1 ft ½ inch diameter)
- Rope (approx. 2mm wide, 5 m per trap)
- 4 small Zip ties (to anchor the screen to rebar)
- 1 PVC pipe glue (for securing the elbows)
- 1 Large sewing needle
- 1 spool of heavy gauge thread for sewing screen

**FRAME AND SCREEN**

1. Cut the tubing in length of 36 cm.



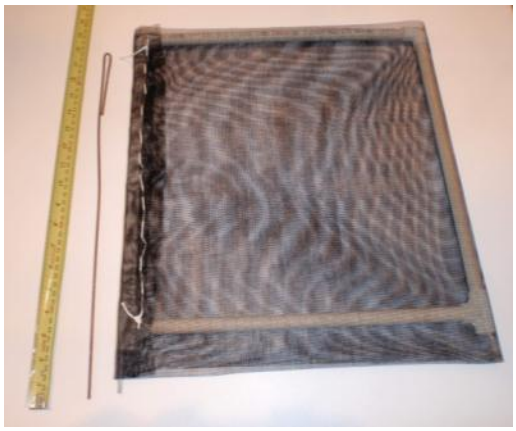
2. Assemble with 90° and T elbow. Make sure elbows are straight. Use PVC glue if necessary (I didn't use any: connections were tight enough)

3. Cut a slice of 45 cm X approximately 92 cm (there is 92 cm rolls sold) of normal window screen (mesh size approx. 2 mm).

4. Use approx. 80 cm of small rope to sew both screen edges together to wrap the tubing frame with a big needle.

5. Secure each end with a few knots. Note that the screen is meant to extend below the bottom so that the inverts are guided into the solution in the pitfall container.

6. Attach 2 zipties on each side of the frame for later use anchoring the screen to the rebar. The rebar should be pounded in the ground just outside the pitfall. Picture shows wire but rebar is preferred.

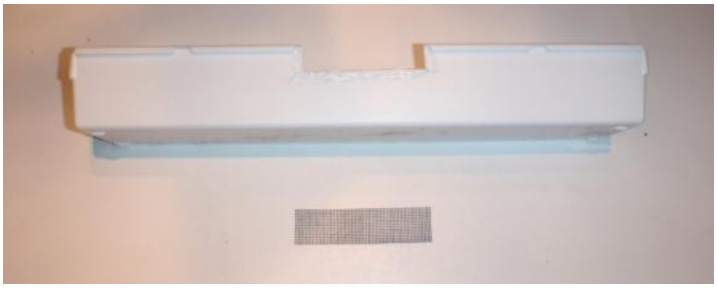


7. Tie a knot on the top tubing (3 turns around the tube and 2 flat square knots).

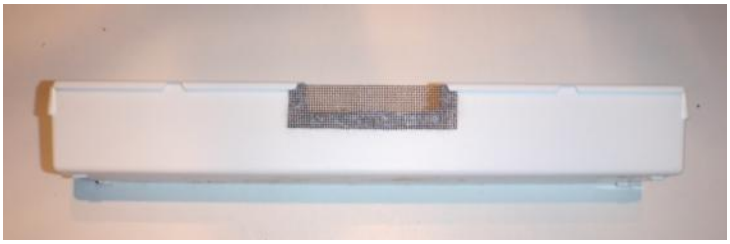
8. Tie both end of this rope together. This loop will be used to keep the frame vertical by providing easy location to secure ropes for anchoring trap.

**PITFALL and overflow**

9. Cut a hole into the side of the pitfall, approx 80 mm X 17 mm. Use a hacksaw to make vertical cuts and a knife to score the horizontal cut (then bend over and break off)



10. Cut a piece of screen approx. 10 cm X 2.5 cm, apply glue (Loctite 454 instant adhesive) on the outside of the container surrounding the edge of the hole and install the screen. Use plastic from the hole to push the screen in place and add glue to secure it. A catalyst may be used to make the glue set faster. The trap is now ready to be assembled outside in your favorite field site.



**INSTALLING THE TRAP**

11. Determine the direction of the dominant winds and configure placement of trap and screen to be **perpendicular** to the dominant wind.
12. Dig just the size of your pitfall container and avoid disturbing the surrounding area.
13. Install the pitfall container in the hole and replace soil so the pitfall is even with the ground.
14. While frame is in the ground pound one piece of rebar just outside each end of the pitfall container on the short ends of the rectangular pitfall container.
15. Install the frame. Use zip ties to secure frame to rebar, small holes in the netting for the zip ties, will be okay. Keep zip ties loose enough so frame can slide off rebar.
16. Use nails (1 or 2 on each side) and rope to keep the frame straight with the loop made on the top tubing of the frame. You may use simple knots to allow an easy removing of the frame to make it fast and easy to remove the container from the ground and drain its content.

## *Appendix M. Lemming live trapping*

(modified from trapping methods proposed by Kaithryn Ott for Barrow)

Use live trapping on trapping grids to obtain density estimates of each rodent species present. Information on sex, age and breeding condition can be collected if desired. Live trapping is considered the most accurate approach for obtaining a true estimate of the number of lemmings that occur in an area, and will be used at a limited number of sites (most likely Nome, Barrow, East Bay, and Bylot Island) to correct our less precise indices.

***Consistency in approach between camps***

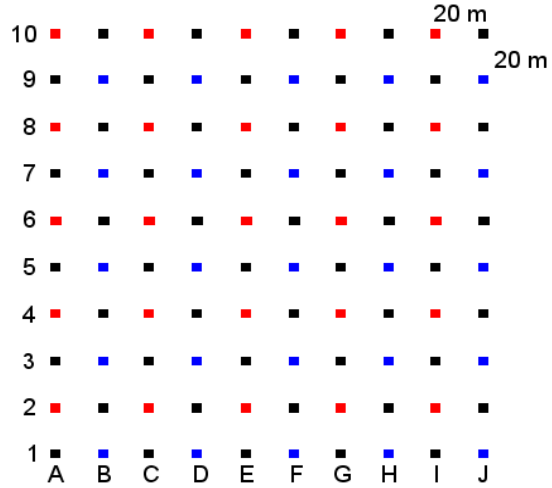
There are a number of constraints that will make following the protocol difficult for all camps. Among these constraints are trap type. Longworth traps are recommended due to the fact that they have a box on the end that can be filled with food and bedding to minimize lemming mortality due to exposure. We recognize, however, that these traps are expensive and that several camps may need to use the cheaper and potentially more accessible Sherman traps. Another constraint might be the topography of the study site. Although we recommend that square trapping grid below, traps set along a transect might be preferable in areas with lots of wetlands. Also, some researchers may prefer using a "chevron" trapping grid (L-shaped). Despite these differences, our hope is that each camp can generate a density estimate that is both reliable and comparable. At present I do not know if there are any differences in how effective Sherman and Longworth traps are in capturing animals but given we are not in a position to change this variable, we will need to live with this.

***Time period***

Live trapping should be done twice every year, once soon after spring melt, and once before the snow sticks in late summer. This is the best way to infer stage in a cycle, because populations can change dramatically during the summer, and mid-summer trapping alone does not give a picture of population trend. Given the time constraints of most ASDN camps, we anticipate only a single round of live-trapping to be conducted (shortly after snow melt) but encourage more if possible.

***Trapping grid setup***

Use a 100 m long measuring tape and a compass to measure out a 10x10 trapping grid with 20 meter spacing (3.24 ha), or a grid of larger square shape, such as a 16x 16 grid with 20 m spacing (9 ha). This can be done with 2 people but 3 people are most efficient. Mark each grid point with a wooden stake (wooden surveyor's lathe, 60 cm lengths) labeled with the row and column. Place a Longworth (recommended) or Sherman trap at a suitable site within 5 m of every second grid station for a total of 50 or more depending on the size of the grid (or every grid station if the population density is high and the large majority of traps are occupied each check). Stagger the trap placement so they are evenly spaced over the area (black squares represent traps in the schematic below). It is useful to color code the rows to help with orientation while in the middle of the grid. One way to do this is to temporarily place red and blue surveyor flags next to each station. These can be picked up at the end of trapping session (or laid flat on the ground) to minimize wind damage to the flags.



***Setting traps***

Oats – Place a small handful of whole oats in the box of the Longworth traps or at the rear portion of the Sherman traps (careful not to get under treadle).

Bedding - Line the box of the Longworth traps or the back of the Sherman traps (again careful not to get under treadle) with upholstery cotton or wool fleece for bedding. Use just enough cotton or fleece to create a warm nest. If too much cotton is stuffed in the box, the lemmings may view it as a blockage as opposed to nesting material.

Apple – Place a chunk of apple in the box in front of the bedding. We get 32 squares from a large apple. Cut in squares rather than slices to minimize surface area and desiccation.

***Trap assembly***

Longworth traps: Unlock the locking mechanism of the Longworth tunnel and check that the door falls properly when the treadle is depressed. Adjust the mechanism if necessary and when all is in good working order, set the trap and connect the tunnel with the box. Make sure that the apple is not too close to the treadle.

Sherman traps (3”x3.5”x9” folding aluminum): unfold flattened trap so it becomes rectangle in shape. Adjust treadle holder so the door will snap shut easily. When setting traps in field, be sure food and cotton are not too close to entrance so as to prohibit door from closing.

***Trap placement***

Place traps as close to the stake marking the location as possible (maximum 5 meters radius from stake). Search the area carefully for fresh sign such as an active burrow or runway. If you find a good runway, place the trap right on the runway. If there is a burrow, place the trap in front of the entrance but do not block the entrance. If there is no sign, place the trap under cover if possible. Make sure the trap is flat and stable and make sure that the door hasn’t fallen shut or the apple rolled under the treadle while placing the trap. Place a board or cover over the live trap to shelter it from too much sun or rain.

***Checking traps***

A trapping session should last 2 days (48 hours) and all traps should be checked every 4-6 hours. ( If you do not want to check traps at night (we prefer not to), it is acceptable to set the traps early in the morning, check every 4-6 hours and then lock the traps open at the last evening trap check. Traps will have to be re-opened the next morning.) You should have at least 12 trap checks in a 2 day trapping period. Do not trap more than 2 days in a row or “trap-happy” individuals will be caught too often and may die in the trap. At the last trap check the doors should be locked open and the traps should be left in 2 pieces so that there is no chance of an animal being caught by mistake.

Place a few cm of oats in the bottom of your trapping bucket and walk past every trap. If you find a trap with the door closed, gently open the trap in the bucket and tip the animal out onto the oats. The oats provide a soft landing surface for them. At this point you can check species and get your tagging equipment prepared. Scoop the animal up with a gloved hand (see footnote 1 below). Use the other un-gloved hand to get a good grasp of the tail. Be careful not to squeeze the animal and keep the eyes covered to minimize squirming. Check carefully for an eartag and record the number if it’s already tagged. If a new tag is required and the species is *Dicrostonyx* you will need 2 people to tag properly. The person handling the lemming can use both hands to hold it so that its right ear is exposed but eyes covered. The person tagging can use the blunt tweezers to gently extend the ear flap with one hand and tag with the other hand. Make sure the tag point has pierced through the ear flap and tag hole and folded over properly (check each tag’s alignment before placing in the pliers). Be careful to not catch too much skin in the tag (increases chance of infection) but place the tag far enough in so that it doesn’t

easily rip out. The ears of brown lemmings and voles are big enough that they can be successfully tagged without the use of tweezers. Record the tag number.

Check the sex, reproductive condition and weight of the animal. Record the data and release the animal. Reset the trap with fresh bait and dry bedding.

If the animal caught already has a tag and was caught earlier in the same trapping period, you do not need to check the sex and weigh the animal. Simply record the tag number, location, and which check number it is then release the animal.

***Equipment needed***

- 100m measuring tape and compass for setting up trapping grids
- 20 litre bucket with minimum 40 cm high sides for holding animals
- Traps and trap boards
- Whole oats
- Apple (preferably Granny Smith)
- Upholstery cotton or wool fleece for bedding
- Ear tags & tagging pliers (or injectable transponder tags and readers if you have lots of money)
- Blunt tweezers for holding tiny *Dicrostonyx* ears when tagging
- 100 g Pesola scales for weighing adults; 30 g scale for juveniles; 300 g scale for some pregnant females.
- Light drawstring bag for holding the animal while weighing
- Trapping gloves
- Bandaids

***Data analysis***

Data should be entered into a database and checked immediately for errors. Density estimates can be computed using Program Capture or Program MARK, or by the Petersen or Schnabel estimators.

Footnote 1: another method for handling is to place a clear plastic bag around the trap. Hold the bag shut and work through the bag to break the trap open and release the animal into the bag. Scruff the animal through the bag so that you can open the bag and remove the trap without letting the animal escape.

## *Appendix N: Methyl Mercury Sampling and Shipping Protocol 2013*

**Note: Methods for blood collection differ depending on your method of storage, freezer or cryoshipper. Please follow the appropriate directions in the section on blood collection.**

**Sample Size (refer to 2013 sample collection protocol that was sent to the PI for your field site):**

- Sample species with at least **10** individuals available to sample at your site (blood and feather samples)
- Sample **30** individuals from each species if available (blood and feather samples)
- Sample up to **50** individuals from each species if possible and time and resources allow (sample blood only after 30 individuals have been sampled for feathers)

**Mercury Feather Samples**

Feather Sample Collection: Pull the entire flight feather or primary covert from each wing, collecting a total of two feathers from each bird (feathers should be pulled in a symmetrical fashion; see Figure 4, and refer to Appendix A for details on which feathers to collect). Place ONE feather in the brown manila envelope provided.

Fill out the (larger) label provided and affix it to the envelope. Be sure to circle "**Mercury**" on the label. This feather should be sent to BRI (see below). The second feather collected should be placed in a second brown manila envelope provided with "ASDN archive" circled on the label. This feather should be sent to USFWS Migratory Bird Management Office.

**Special considerations for Dunlin in flight feather molt:**
For Dunlin only: Collect 5 black breast feathers; place all 5 breast feathers in an envelope (flight feathers not needed for Dunlin) and label as above. Be sure to circle "**Mercury**" on the label. These feathers should be sent to BRI (see above under blood sample shipping).

A summary of what feathers to collect for methyl mercury from each species is listed below.

| Species | Feathers collected from both wings (see Figure 4) |
|---|---|
| DUNL (in primary molt) | Innermost new and old primary (see details above), black breast feathers |
| DUNL (not in primary molt) | 1st primary, black breast feathers |
| All other captured species | 10th secondary |

**Shipping Samples:**
Send samples directly to: Jennifer Goyette
Biodiversity Research Institute
652 Main Street
Gorham, ME 04038 USA

Please ship all remaining sampling supplies back to BRI with the collected samples and email an inventory of samples with associated data collected in the field.

Contact: Jennifer Goyette and Kevin Regan (BRI's Mercury Lab Manager) by e-mail: jennifer.goyette@briloon.org; kevin.regan@briloon.org or phone: 207-839-7600 ext.107 (Kevin; mercury lab). When you ship samples let them know when to expect the shipment to arrive. Avoid having samples arrive at BRI's Mercury Lab over the weekend (i.e. ship samples no later than Tuesday). Blood samples do not need to remain frozen during shipping, but do need to remain cool; they should be packed securely in packing material (provided in supply shipment) to prevent breakage and shipped in a hard-sided cooler filled with frozen ice packs. Samples should be shipped 2-day if possible. Prior to shipping, place all feather samples for mercury in a plastic bag for protection. If necessary, feather samples can be shipped separately from the blood samples in an envelope and do not require 2-day shipping. If you are unable to ship via FedEx or if shipping blood samples from your site will require longer than 4 days, please contact: Jennifer Goyette ( phone: 414-526-0808).

Shipping samples via FedEx: Please mark "Bill Recipient" and include BRI's FedEx account number: 225789-827 (to be used only for shipping samples to BRI). Under "Reference Number" write "MARI IN13-007".

## *Appendix O: Pond hydrology*

This supplement describes methodology that was used for collecting data to complement the aquatic invertebrate emergence phenology studies. **This project is not active in 2013**, but the protocol is included here for reference for those sites that participated.

Please don't hesitate to contact Daniel Rinella (University of Alaska Anchorage; rinella@uaa.alaska.edu; 907-748-2154) if you have any questions regarding these activities.

| | Activity | Frequency |
|---|---|---|
| A. | Temperature monitoring | Install at beginning of season and remove at end of season |
| B. | Water level monitoring | Each time invertebrate sampling is conducted |
| C. | Habitat measurements | After ice is melted in the pond (once per year) |

**A. Temperature monitoring**

- Equipment list provided by **ASDN**: 5 Hobo temperature loggers, plastic cable ties
- Equipment list provided by **camp**: 5 rebar (4- ft) stakes, hammer for driving stakes, chest waders for deploying loggers.

Deployment: For each pond, record temperature logger number and other requested information on the *Pond water level and temperature monitoring datasheet*. Note that each pond will have its own datasheet.

Deploy one temperature logger in each of the 5 ponds at the beginning of the field season. Temperature loggers should be deployed in a location with relatively deep water so they do not go dry when water levels fall. Attach the temperature logger directly to the rebar stake using two cable ties, making sure the sensor end (i.e., the end with the hole) is pointing down. Sink the stake into the pond bottom so that the temperature logger is just above the pond bottom. Check periodically to ensure that temperature logger remains near the pond bottom; adjust the depth if necessary. If water levels fall enough that any temperature logger is in danger of going dry, move it to a deeper location. Be sure to record what # logger went in what pond.

Retrieval: pull the rebar stake from the pond, cut the cable ties holding the temperature logger, and return all the temperature loggers to USFWS Alaska Regional office in Anchorage.

**B. Water level monitoring**

- Equipment list provided by **ASDN**: short piece of wire, nylon string, line level, meter stick or tape measure,
- Equipment list provided by **camp**: 5 rebar (4-ft) stakes, hammer for driving stakes, pliers for twisting wire

1. Prior to the first water level measurement, drive the rebar securely into the pond shoreline. It is preferable to place it along the shoreline where there is a sharp edge to limit the wading distance required as lake levels drop over the summer. Once the rebar is in place, make a permanent mark on the rebar near the ground by wrapping a piece of wire around the stake and twisting it in place with pliers. The wire will serve as an elevation benchmark for the duration of the study, so leave the rebar in place. Record GPS coordinates of the benchmark on the *Pond water level and temperature monitoring datasheet*.
2. The vertical distance between the benchmark (i.e., the wire) and the pond surface will be measured each time aquatic invertebrate sampling is conducted. To do this, tie a string to the rebar at the benchmark (i.e., the wire), unspool the string until it extends over the water surface, and hang the line level on the string. Raise or lower the pond end of the string until the bubble is centered within the line level. You may need to clear some vegetation to have a clear path for the string.
3. Once the line is level, measure the vertical distance between the line and the water surface (Figure 19). Record this distance (along with the date) and any comments on the *Pond water level and temperature datasheet.* These repeated measurements will allow us to track changes in water level throughout the sampling season.

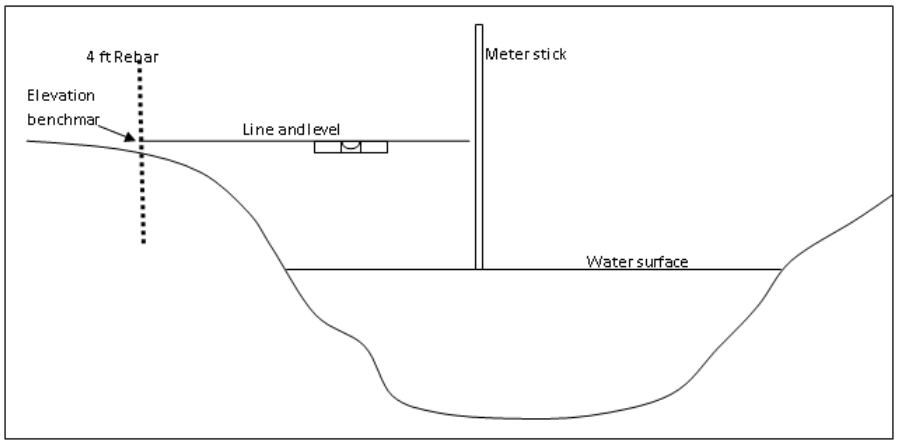



Figure 19. Schematic showing method for measuring vertical distance between elevation benchmark and pond water level.

**C. Habitat measurements**

- Equipment provided by **camp**: meter stick (or longer graduated stick) for measuring depth, camera, GPS, chest waders for person conducting measurements
- Procedure for each pond: (a) record a GPS track of the shoreline perimeter to calculate pond's surface area, (b) record depth measurements and substrate type at a systematic array of 15 points distributed across the pond, (c) take a series (6 – 8) of photos.
    1. Walk the shoreline with a handheld GPS and have it calculate the pond's surface area. Record the surface area measurement on the *Pond habitat datasheet*. The procedure for measuring areas varies by GPS make and model, so check your owner's manual. If you have a Garmin etrex GPS, go to the main menu, select *Tracks*, press the menu button again, select *Area Calculation*, select *Start*, walk the pond's perimeter, then select *Stop*. The etrex will then calculate the area.
    2. For the depth and substrate measurements, visually locate three equally spaced transects that will run the length of the lake and take measurements at five points roughly equally spaced along each transect. Thus, measurements will be taken at 15 points in a grid pattern equally distributed across the lake (Figure 20). Record the approximate distance between each of the three transects and the approximate distance between each of the 5 sampling points on each transect on the datasheet. At each of the points, lower a meter stick (or make a longer graduated stick if necessary) to the bottom and record the depth and type of bottom substrate on the *Pond habitat datasheet*. The various substrate categories are given on the datasheet; be sure to record the two-letter code. If pond is too deep or soft to safely wade, then note on the datasheet and skip the depth and substrate measurements. Also, record the predominant color of the bottom substrate on the datasheet.
    3. Take 6 – 8 photos of the pond from various angles and distances. Include a few photos that show the crew taking habitat measurements, measuring the pond water level, and collecting invertebrate samples. Include any other interesting photos from the pond. Store all the photos from a given pond in a folder bearing the camp code and pond number.

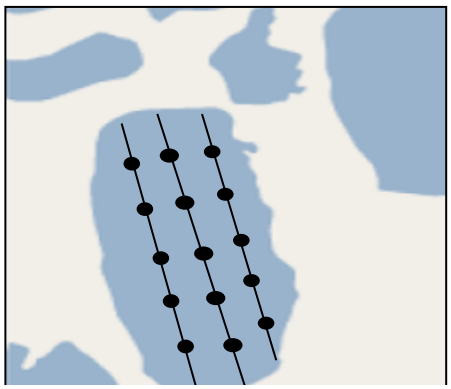


Figure 20. Schematic showing layout of 5 sampling points on each of three equally spaced transects

**Pond notes.**

Please record notes regarding each pond on the *Pond notes datasheet*. Include comments on water level fluctuations, periods of heavy aquatic insect emergence, bird activity in and around the pond, or anything else of interest. Please include dates of observations and feel free to attach additional sheets as necessary.

**Storage of pond temperature loggers**

Store temperature loggers in a quart size plastic bag with the camp site code labeled on the outside. Include a hard copy of the raw data (3 different forms, water level, habitat and pond notes).

## *Appendix P: Retrieval and return of DUNL geolocators*

*Rick Lanctot, Shorebird Coordinator, USFWS, Alaska*
*Stephen Yezerinac, Biology Department, Mount Allison University, Canada*

As you know, in 2011 and 2012 we recovered many geolocators deployed on Dunlin in 2010. 2013 represents another opportunity to get loggers from birds that were missed previously. Although the logger batteries are now dead, the data should still be recoverable and valuable for analyzing individual movements and population segregation. Last year's data indicate that populations of *pacifica* are partially segregated in winter. It's difficult to discern such a pattern for *arcticola,* though also perhaps less likely given the geography of these populations. So strengthening the analysis with more loggers would be valuable. Thus, we encourage you to catch geolocator birds as soon as the nest is discovered.

*1. Remove the entire band with the attached geolocator*. We suggest using cuticle scissors or some other fine-tipped, sharp scissors to snip the rings around the bird's leg. Of course, it's critical to snip the rings without injuring the leg or damaging the geolocator.

*2. Label the geolocator flag with the bird's band number*. Use the same fine-tipped, permanent marker used for the blood vials and write the band number directly on the flap of the geolocator band. **It is critical that the geolocator be identified with the band number of the bird that carried it**; the geolocator will no longer have a readable unique ID and unless we can link the instrument to last year's records via the Band Number the data will be useless.

*3. Place each geolocator in its own coin envelope or plastic baggy*. It's possible that some geolocators may be loosely held to the flag, so it is critical that each geolocator be kept separate in its own coin envelope or plastic baggy.

*4. Label the envelope or baggy with the following:*

Species _____
Band number _____
Date _____
24 hr time of retrieval _____
General Location (e.g. Barrow) _____
GPS location (in NAD83) _____ (in correct time zone)

*5. Mail the loggers back for data download ASAP once the field season ends.*

Before mailing:

- ✓ Please check that you have filled out the data labels completely. The Band Number, Date, time and location are critical information for interpreting the data from the geolocator.
- ✓ Pack the geolocators so they are not loose and so that they have some cushioning to prevent damage in transit.
- ✓ Please email <Richard_Lanctot@fws.gov> or <syezerinac@mta.ca> to let us know to expect the shipment.

- If mailing from USA field site, send to

  Rick Lanctot
  Alaska Shorebird Coordinator
  U.S. Fish and Wildlife Service
  Migratory Bird Management Division
  1011 East Tudor Road, MS 201
  Anchorage, Alaska 99503
  Phone: 907-786-3609

- If mailing from Canadian field site, send to

  Stephen Yezerinac
  Biology Department
  Mount Allison University
  63B York St.
  Sackville, NB, E4L 1G7
  Canada
  Phone: 506-364-2260

If you need financial assistance shipping the geolocator, contact Richard_Lanctot@fws.gov for a FedEx account number to use. Thanks so much for your assistance!

## *Appendix Q: Do migratory shorebirds disperse Moss (Bryophyta) diaspores?*

Request for Feather Samples and Project Description

a) Background: Many bryophytes (mosses, liverworts, and hornworts) display broad infraspecific geographic disjunctions (Schofield 1992). Such disjunctions may be explained by vicariance paired with evolutionary stasis of fragmented populations, or recent to ongoing long distance dispersal (LDD). Molecular studies have overwhelmingly pointed toward LDD as leading to broad infraspecific disjunctions in bryophytes

(Heinrichs 2009; Shaw 2001), including the most extreme disjunction, the bipolar pattern (Piñeiro et al. 2012; Kreier et al. 2010). Preliminary data from the doctoral thesis work of L. R. Lewis suggests that LDD is the process resulting global disjunctions in a dispersal limited entomochorous dung moss (Tetraplodon mnioides), perhaps resulting in at least three southern hemisphere colonization events. The process of LDD requires a vector, such as wind or migratory birds (human dispersal should also be considered, but is beyond the scope of this proposal). A lack of wind connectivity between extreme northern and southern high latitude regions suggests that dispersal by birds may be more likely. Migratory shorebirds have been proposed as potential vectors for bipolar dispersal, most recently by Popp et al. (2011) for the vascular plant Crowberry (Empetrum), but absence of field collected or experimental data allows for only speculation.

b) Justification & relevancy of the project: We are requesting feather samples from migratory shorebirds, in order to screen the feathers for diaspores, culture the diaspores, and use molecular markers to identify the bryophyte species being carried by the sampled birds. This project is being proposed as a side project to the ASDN, as migratory shorebirds encountered by field camps could be sampled (see sampling protocol below) to provide feathers for screening.

c) Objective: Provide evidence for dispersal of bryophyte diaspores by migratory shorebirds.

d) Methods:

a. Requested Feather Sampling Protocol: This side project will not affect the core objectives of the ASDN.

Location: Along migratory paths of the target bird species. Collections made from birds about to migrate south or birds recently arrived from southern range would be especially desirable.

Feather sampling: Please pluck 3-5 feathers from the base of the breast bone and put them in a single envelop.

Feather Storage: Dry, at room temperature in paper bag or envelope. Envelopes will be prepared and provided for feather collection.

Collection Information: Bird species, locality, date, collector, any relevant molt information.

b. Feather screening, diaspore collection, culture, and ID: The Side Project Investigator, L. R. Lewis, will direct this portion of the methods.

Location: The labs of Dr. B. Goffinet and Dr. L. Lewis at University of Connecticut, Storrs.

Primary Investigator: L. R. Lewis

Procedure Note: The culture protocols, referred to below, have been optimized in the Goffinet Lab. Diaspore detection is currently being optimized on feather samples from the UCONN salvage collection.

1. Feathers will be visually screened for diaspores with dissecting and light microscopes in a ventilated sterile fume hood to prevent contamination.
2. Feathers will be votexed and centrifuged in distilled water to remove particulate matter over two replicates. The resultant debris removed from the feathers will placed on sterile growth media.
3. Cultures will be maintained in a sterilized growth chamber, under controlled conditions. Conditions will simulate a cool temperate environment.
4. Cultures will be monitored weekly for germinating diaspores.
5. Upon germination, a modified single spore isolate protocol will be followed to create single strain cultures.
6. When adequate biomass has developed, a small voucher will be dried and saved, and a portion of the material will be sequenced for the rps4 (chloroplast locus) and its2 (nuclear ribosomal locus) and blasted against the NCBI Genbank database for identification. Additional material will be kept in culture for the potential of providing morphological characters for identification.

e) Requested Samples:

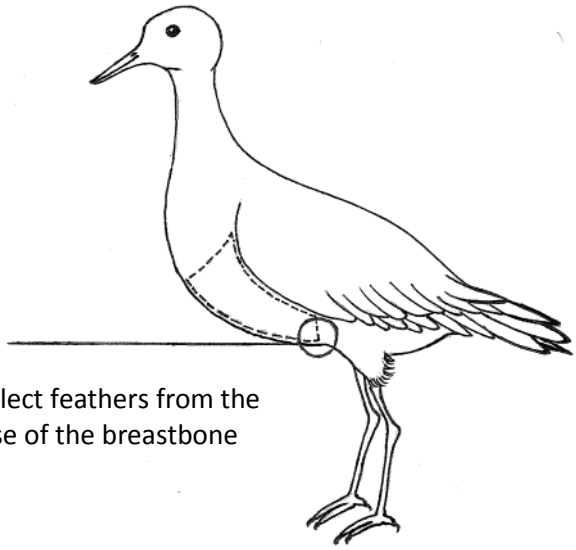
**A sample of 3-5 feathers from base of breastbone.**

Collection Note: If the sampled bird has begun prebasic molt then select older-generation feathers. If newly grown feathers are the only option, please make note.

Quantity: The success of this study depends on having a large sampling of feathers. As many samples as can be reasonably collected would be appreciated. If collaboration is agreed upon, sampling kits will be provided to match the number of samples field researchers are willing and able to collect.

Target bird species

- Surfbird Aphriza virgate
- Ruddy turnstone Arenaria interpres
- Baird's sandpiper Calidris bairdii
- White-rumped sandpiper Calidris fuscicollis
- Sanderling Calidris alba
- Red knot Calidris canutus
- Semipalmated Sandpiper Calidris pusilla
- Pectoral Sandpiper Calidris melanotos
- Least Sandpiper Calidris minutilla
- Western Sandpiper Calidris mauri
- Stilt Sandpiper Calidris himantopus
- Dunlin Calidris alpine
- Semipalmated Plover Charadrius semipalmatus
- Long-billed Dowitcher Limnodromus scolopaceus
- Hudsonian godwit Limosa haemastica
- Bar-tailed Godwit Limosa lapponica
- Whimbrel Numenius phaeopus
- Red-necked Phalarope Phalaropus lobatus
- Red Phalarope Phalaropus fulicarius
- American golden plover Pluvialis dominica
- Black-bellied Plover Pluvialis squatarola
- Lesser yellowlegs Tringa flavipes
- Buff- breasted Sandpiper Tryngites subruficollis



Collect feathers from the base of the breastbone

f) Expected results & benefits: This study has the potential to offer the first evidence for LDD by bird, contribute to the PhD thesis of L. R. Lewis, investigating the phylogeography of the bipolar moss species Tetraplodon mnioides, and provide undergraduate research opportunities.

g) Timeline for completion of the project:

- Fall 2012: Feather screening protocol optimization
- Spring 2013: Finalize screening protocol and begin Screening Feather Samples
- Fall 2013: Feather Screening
- Spring 2014: DNA analysis of diaspore cultures
- Summer & Fall 2014: Manuscript preparation and publication

h) Anticipated products: The results of this study will be published in order to contribute to the ongoing discussion regarding the potential of migratory shorebirds as dispersal vectors for bryophyte species. While the focus here is on bryophytes, this study will have relevance for all plant biogeographers.

i) Funding: This side project is being funded by the National Science Foundation Graduate Research Fellowship, Awarded to L. R. Lewis. And Federal Work Study awarded to undergraduate researchers.

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Statement of Authorship: Authorship will be determined as specified in the STATEMENT OF AGREEMENT FOR ARCTIC SHOREBIRD DEMOGRAPHICS NETWORK SIDE PROJECTS (26 October, 2011)

j) Literature cited in this Appendix:

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## *Appendix R: Nest Fate and Egg Fate Scenarios*

The following represents various scenarios to help ensure consistency in how nest fates and the cause of nest failure are classified, and to help ascertain fate of individual eggs.

**Abandonment Scenarios**

Scenario 1

Nest was thought to be abandoned prior to estimated hatch date so researcher.turned eggs out and next visit found eggs still turned out. (this approach will not work on 3 egg clutches since it is common for one egg to sit with the big end inward). In all cases, information on the warmth of the eggs and the presence of adults should also be used to ascertain that a nest has been indeed abandoned.

Day last active=day visited nest prior to turning eggs out
Fate date= day turned eggs out initially

Nest fate=failed
Fail cause=abandonment

#of eggs abandoned=4
# of eggs failed to hatch=0
# of eggs predated=0
# eggs hatched=0
# eggs observer=0
# eggs unknown=0

Scenario 2

Same as scenario 1, but eggs were monitored past estimated hatch date and there was no evidence of nest being active (no data on whether adult was seen; may have written turned eggs out, but following visit didn't write if eggs were still turned out; no data on egg warmth, etc.).

Day last active=day prior to estimated hatch date
Fate date=day after estimated hatch date
(rationale: so program Mark records nest alive until expected hatch date)

Nest fate=failed
Fail cause=abandonment

# eggs abandoned=4
# eggs failed to hatch=0
# eggs predated=0
# eggs hatched=0
# eggs observer=0
# eggs unknown=0

Fail to Hatch Scenarios

Scenario 1

Nest was monitored up to or past expected hatch date. Adult observations suggest parents were present up to expected hatch date. No eggs ever hatched. Either collected or assessed for fertility in field AFTER expected hatch date.

Day last active=day prior to estimated hatch date

Fate date=day after estimated hatch date
(rationale: so program Mark records nest alive until expected hatch date)

Nest fate=failed
Fail cause=failed to hatch

# eggs abandoned=0
# eggs failed to hatch=4
# eggs predated=0
# eggs hatched=0
# eggs observer=0
# eggs unknown=0

**Hatching Scenarios**

Scenario 1
Nest was monitored and found with 3 chicks in nest cup. 4th egg never hatched.

Day last active=day found with chicks
Fate date=day found with chicks

Nest fate=hatched
Fail cause=n/a

# eggs abandoned=0
# eggs failed to hatch=1
# eggs predated=0
# eggs hatched=3
# eggs observer=0
# eggs unknown=0

Scenario 2
Nest was monitored for 2 consecutive days. On last visit eggs were present, but the next day nest was empty but evidence indicated hatching (e.g., brood found nearby, egg caps, egg bits, nest flattened, etc.)

Day last active=day found nest empty
Fate date=day found nest empty

Nest fate=hatched
Fail cause=n/a

# eggs abandoned=0
# eggs failed to hatch=0
# eggs predated=0
# eggs hatched=4 (unless the fate of some eggs determined for some other reason prior to this event or eggs left in nest)
# eggs observer=0
# eggs unknown=0

Scenario 3

Nest was monitored with ≥3 days between nest checks and found to be empty. Upon looking, a brood was found near the nest with an adult marked that corresponded with nest. If brood was found far (>100m) from the nest or if brood was seen with adult ≥1 day after nest found empty use scenarios 4-6 below.

Day last active=day prior to finding brood
Fate date=day prior to finding brood

Nest fate=hatched
Fail cause=n/a

# eggs abandoned=0
# eggs failed to hatch=0
# eggs predated=0
# eggs hatched=4 (unless the fate of some eggs determined for some other reason prior to this event or eggs left in nest)
# eggs observer=0
# eggs unknown=0

Scenario 4
Nest was monitored with ≥3 days between nest checks and found to be empty. However, during the last visit eggs were found to be pipped or heavily starred, and at least some of the other available information suggests it hatched (egg caps, egg bits, nest flattened).

Day last active=one day after visit when eggs found pipped or heavily starred
Fate date=one day after visit when eggs found pipped or heavily starred

Nest fate=hatched
Fail cause=n/a

# eggs abandoned=0
# eggs failed to hatch=0
# eggs predated=0
# eggs hatched=4 (unless the fate of some eggs determined for some other reason prior to this event or eggs left in nest)
# eggs observer=0
# eggs unknown=0

Scenario 5
Nest was monitored with ≥3 days between nest checks and found to be empty. However, during the last visit eggs were found to be lightly starred, and at least some of the other available information suggests it hatched (egg caps, egg bits, nest flattened).

Day last active= two days after visit when eggs found lightly starred
Fate date=two days after visit when eggs found lightly starred

Nest fate=hatched
Fail cause=n/a

# eggs abandoned=0
# eggs failed to hatch=0

# eggs predated=0
# eggs hatched=4 (unless the fate of some eggs determined for some other reason prior to this event or eggs left in nest)
# eggs observer=0
# eggs unknown=0

Scenario 6
Nest was monitored with ≥3 days between nest checks and found to be empty. During the last visit where the nest had eggs, no eggs were found starred or pipped. However, the available information during the very last visit suggests the nest hatched (egg caps, egg bits, nest flattened, unmarked brood nearby).

Day last active= day prior to finding the nest empty or the median date between visits depending on which is closest to the expected hatch date.
Fate date= day prior to finding the nest empty or the median date between visits depending on which is closest to the expected hatch date.

Nest fate=hatched
Fail cause=n/a

# eggs abandoned=0
# eggs failed to hatch=0
# eggs predated=0
# eggs hatched=4 (unless the fate of some eggs determined for some other reason prior to this event or eggs left in nest)
# eggs observer=0
# eggs unknown=0

**Partial Predation Scenarios**
Scenario 1
Nest loses 1 egg on a visit, and may or may not lose another egg the next visit (apparent partial predation). There are no reasons listed in notes that suggest eggs damaged by observers. Then, adult sits on nest until well past expected hatch date, then finally abandons nest on its own.

Day last active= day prior to estimated hatch date
Fate date= day after estimated hatch date
(rationale: so program Mark records nest alive until expected hatch date)

Nest fate=failed
Fail cause=predation/failed to hatch

# eggs abandoned=0
# eggs failed to hatch=# eggs left in nest when adult abandoned
# eggs predated=# eggs that disappeared from nest
# eggs hatched=0
# eggs observer=0
# eggs unknown=0

Scenario 2

Nest loses 1 egg on a visit, and may or may not lose another egg the next visit (apparent partial predation as in scenario 1). However, this time the adult continues to incubate the nest and it hatches (despite predation we still call this hatching as long as one chick hatches in nest).

Day last active = hatch date (see above hatching scenarios)
Fate day = hatch date (see above hatching scenarios)

Nest fate = hatched
Fail cause = n/a

# eggs abandoned =0
# eggs failed to hatch=0
# eggs predated = # eggs that disappear from nest = x
# eggs hatch=# chicks found in nest or 4-x
# eggs observer=0
# eggs unknown=0

Scenario 3
Nest loses 1 egg on a visit, and may or may not lose another egg the next visit (apparent partial predation as in scenario 1). However this time the adult abandons the nest soon after predation event.

Day last active = date prior to partial predation event
Fate day = date seen partially predated

Nest fate = failed
Fail cause = predation/abandonment

# eggs abandoned=# eggs left in nest after partial predation
# eggs failed to hatch=0
# eggs predated=# eggs removed by predator
# eggs hatch=0
# eggs observer=0
# eggs unknown=0

There are potential more scenarios that involved partial loss of nest to multiple factors – see fail cause list below.

**Unknown Scenarios**

Scenario 1
Nest visited 4 days prior to hatch and no eggs are starred. During next visit (typically 2 days later), no eggs are in nest and there is not enough evidence to classify nest fate (see ASDN protocols in Appendix F) or there are mixed signals for whether nest hatched or not.

Day last active= day when nest was last still active
Fate date= day found nest failed

Nest fate=Unknown
Fail cause=n/a

# eggs abandoned=0

# eggs failed to hatch=0
# eggs predated=0
# eggs hatched=0
# eggs observer=0
# eggs unknown=4

There are more scenarios where eggs went missing prior to the unknown scenario above – treat like partial predation scenarios above.

**Egg Fates**

#Hatched = # of eggs that resulted in chicks based on observations of chicks in nest or assumptions based on hatching that relies on evidence indicating hatching (e.g., starred or pipped eggs, egg caps, egg bits, flattened nest)(see above). For example, we may assume 4 eggs hatched if 4 eggs were present in the nest on a prior visit and last visit indicates hatching as egg bits were found in nest.

#Abandoned = # of eggs in nest when adult abandoned (as long as abandonment was determined to have occurred prior to expected hatch date).

#Fail to hatch = # of eggs left in nest after chicks depart a "hatched" nest, OR # of eggs in nest when nest was determined to have failed to hatch (do not include eggs that failed to hatch due to observer-related causes).

#Predated = # of eggs that disappear from a nest prior to expected hatch date (this could be a few eggs or all the eggs if an entire clutch disappears), unless there are observations of an egg being infertile or destroyed in a previous visit – then assume adult carried egg away and it failed to hatch.

#Observer=# eggs destroyed by researchers (e.g., stepping on eggs, cracking eggs during measurement, etc.).

#Unknown=fate of egg could not be determined. Could have hatched or been depredated. Use when fate is undetermined.

#Lost = # eggs whose fate were definitely not hatched but could not be ascribed to being abandoned, fail to hatch, predated, observer, or gone for some other reason.

**Egg Rules:**

1) All egg fate categories should add to maximum clutch size for that nest. (i.e., one egg has a single egg fate)
2) If it is uncertain whether an egg disappeared due to being infertile, being damaged by observers, or some other reason, then use the lost category. This may be popular in early years of Barrow or ASDN.

**Nest Fate categories**

Failed=if no chicks hatch
Hatched=if at least one egg hatches
Unknown = could not determine if nest hatched or was depredated based on available information
Undetermined = field researchers left study site prior to the end of nest monitoring, nest was off plot/study area, or nest used for some other purpose (e.g., practice banding, etc.) and then not monitored.

**Fail cause categories** (assign when nest fate = failed)

abandonment
failed to hatch

predation
observer
weather
collect
trampling
n/a = not applicable since the nest did not fail (goes with nest fate = hatched, unknown, and undetermined)

Use these combo cases or others when eggs had multiple fates and cause of nest failure may not be clear. List the fail causes in the order that they occur (e.g., nest was partially predated then failed to hatch =predation/failed to hatch)

predation/failed to hatch
predation/abandon
observer/predation

**Special Note for Phalaropes:** Willow English reported important results from temperature monitors in RNPH nests at Nome, which make it clear when a nest is abandoned or not. She had several nests that would have been deemed abandoned under the protocol for being cold with parent absent for several visits in a row, however based on the temperature logger they were not abandoned and later hatched. She also had a few nests where she turned one or more eggs pointy end out, and found them unchanged yet the nest was not abandoned. In general she has found RNPH to be much less particular about the organization of their eggs compared to WESA and SESA. This could potentially cause nests to be mistakenly called abandoned so she thought it was important to note this for the group.

## *Appendix S: Retrieval and return of geolocators for the AMGP project*

As you know, this season we need to recapture American Golden-Plovers and retrieve the geolocators that were deployed in previous years. **The success of these studies will be proportional to the number of geolocators retrieved**. We need to get data from many birds in each population to accurately assess migration routes, stopover sites, and winter segregation of populations. So we'd like to encourage everyone to make extra effort to get the geolocator birds this season.

*1. Remove the entire band with the attached geolocator*. We suggest buying cuticle scissors from your local drugstrore, or some other fine-tipped, sharp scissors to use to snip the rings around the bird's leg. Of course, it's critical to snip the rings without injuring the leg or damaging the geolocator.

*2. Label the geolocator flag with the bird's Band Number*. Use the same fine-tipped, permanent marker used for the blood vials and write the Band Number directly on the flap of the geolocator band. **It is critical that the geolocator be identified with the Band Number of the bird that carried it**; the geolocator will no longer have a readable unique ID and unless we can link the instrument to last year's records via the Band Number the data will be useless.

*3. Place each geolocator in its own coin envelope or plastic baggy*. It's possible that some geolocators may be loosely held to the flag, so it is critical that each geolocator be kept separate in its own coin envelope or plastic baggy.

*4. Label the envelope or baggy with the following:*

Species _____
Band Number _____
Date _____
24 hr Time of Retrieval _____
General Location (e.g. Barrow) _____

GPS location (in NAD83) _____ (be sure you are in correct time zone)

*5. Store the loggers in the refrigerator (NOT freezer) to preserve battery life.* The logger batteries will die sometime this Spring or Summer. While the data *may* be downloadable after the battery expires, this is a much more complicated process and has an accompanying loss of accuracy.

*6. Mail the loggers back for data download ASAP*. If you have field crew leaving before the end of the season, send out the first batch of loggers with them. Before mailing please check the following:

- Please check that you have filled out the data labels completely. The Band Number, Date, time, and location are critical information for interpreting the data from the geolocator.
- Pack the geolocators so they are not loose and so that they have some cushioning to prevent damage in transit.
- Please email Jean-François Lamarre (jflamarre@gmail.com) to let me know to expect the shipment and if you need financial assistance to ship the geolocators.
- If mailing internationally, be sure to attach the letter (next page) to the Custom's officer to the outside of the package. Send to:

Élise Bolduc / Jean-François Lamarre
University of Québec in Rimouski (UQAR)
300, allée des Ursulines, Rimouski (Québec) Canada G5L 3A1
Téléphone : 418 723-1986 ext 1911or 1909

Thanks so much for your assistance!

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25 March 2013

To: Customs Officials in the United States of America and Canada

Subject: Import and export of Light Level Recorders

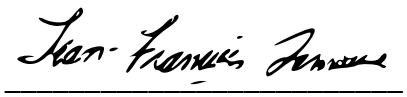
Dr. Joël Bêty, professor at the University of Québec at Rimouski and myself ( Ph. D. Student Jean-François Lamarre) have recently initiated a study to determine in precise detail the migration pathway of American Golden-Plovers (*Pluvialis dominica*). To do so, biologists have equipped plovers with a single Light Level Recorder. Birds equipped with these devices in Alaska will hopefully successfully complete migration and then return to their country of origin to be captured again, at which time data will be downloaded. The levels of light recorded by these devices will allow biologists to estimate the latitude and longitude of each bird on a daily basis, providing valuable information on how and where a bird migrates.

In order to have the best understanding of the species' movements, we teamed up with several collaborators from the USA and Canada. Plovers are nesting in the Arctic from Alaska to Nunavut. We sent devices to these collaborators that installed the devices on the birds. Once the birds come back from their migration, biologists will retrieve the devices and send them back to us for analysis.

Thus this letter is to inform customs officials that these devices are thus being sent to the host country temporarily (for a year or two) and they will be returned to Canada. The devices do not transmit any frequencies but only record light intensity. Accordingly, we hope that you will not impose any import duty on these items nor hold the item for any further clearance. If you have any questions, please do not hesitate to call or write an e-mail to us.

Thank you for your help with this study.

Sincerely,



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