March 28, 2025

RE: Agreement No. **22-CS-11041300-032**

Agreement Title: **Native Plant Restoration**

Performance Period: **01/01/2024 - 12/31/2024**

Annual Performance Report

During this performance period, the following activities were completed:

Timeline	Activities
Winter 2024	• Compiled and distributed 2023 annual report. • Conducted outreach activities with potential partner organizations from the federal, state, non-profit, and education sectors. • Attended the Idaho Noxious Weeds Conference and the National Native Seed Conference. • Allocated new BIL funding to FS agreement. • Secured a larger workshop/storage space and began the process of improving the facilities to meet the program's needs. • Designed and constructed new seed cleaning equipment. • Researched and began implementing new technologies for seedball application. • Began developing seed zones and selected sites for future seedball dispersion. • Developed seedball monitoring protocol.
Spring 2024	• Conducted 2023 seedball monitoring. • Assessed equipment inventory and purchased new materials for seed collection, seedballs, and seed cleaning. • Initiated the hiring process for seasonal interns and technicians. • Developed and dispersed the Native Plants Volunteer Mailing List.

	• Attended trainings via the CPC's Rare Plant Academy. • Processed new FS agreement modifications to expand the program's funding and scope of work. • Uploaded seed cleaning and collection methods to RNGR database. • Worked with USDA Seed specialist to select seed cleaning devices and target species for restoration efforts. • Designed new ArcGIS Field Maps data collection layers for phenology monitoring, seed collection, voucher collection, and seedball dispersals. • Finalized seedball sites and target species for Fall/Winter 2024. • Began phenology monitoring and population mapping.
Summer 2024	• Obtained defensive driving, ATV & UTVS handling, CPR/First Aid, Weed ID, and Chainsaw training. • Applied for and received the 2024 NEEF Pollinator Grant. • Prepared and implemented field safety plan. • Trained and onboarded two SVS interns and three SVS technicians. • Continued phenology monitoring and population mapping. • Conducted seed collection efforts and hosted two volunteer seedball events with 11 volunteers. • Wrote new BIL funding proposal to support work with the Shoshone/Bannock Tribe on the restoration of culturally significant plant species and co-management efforts. Attended field visits with tribal members.
Fall 2024	• Continued seed collection and population monitoring. • Began processing seed including cleaning, weighing, counting, measuring and testing collections. • Designed 12 seed mixes made up of available species from collections. • Produced thousands of seedballs for each mix. • Hosted two seedball volunteer events. • Dispersed seedballs on the landscape. • Purchased equipment for seed storage.

Winter 2024	• Compiled field data from the 2024 field season. Uploaded relevant data to the appropriate databases. • Workshop renovated and equipment installed. • Initiated seed moisture testing for storage.

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Program Goals

Our program's primary goal is to reintroduce native species to some of the most degraded landscapes within the greater Salmon area. Increasing species biodiversity and reconstituting healthy plant communities is known to increase ecosystem resiliency to wildfires and biological invasions. Additionally, well balanced plant communities are more capable of supporting pollinator species throughout the season, providing more heterogeneous habitat for wildlife, and reducing erosion and soil damage.

With the introduction of funding from the 2024 NEEF Pollinator Grant supported by Toyota, we worked on picking target locations for restoration that would allow us to focus our efforts and make a more noticeable dent in the damage observed on the landscape. We chose the hillsides of the Salmon River Corridor from North Fork to Panther Creek as our primary restoration effort. This area provides a unique restoration challenge as most of the hillsides are both extremely steep and rocky; and the area has a long history of high intensity wildfire and over grazing by sheep, rendering the landscape extremely forb depauperate with a nearly depleted seedbank. Our goal here is to connect pollinator habitat from the Salmon River to the ridges beyond by staging key planting areas in advantageous locations essentially leading pollinators to new, previously unreachable habitat.

Additionally, in an effort to combine this program's work with SVS's Milkweed Monitoring Program, we undertook an initiative to spread Western Monarch Butterfly habitat through the Salmon River Corridor. Observations by our Milkweed Monitoring Program suggest that Monarch use of milkweed plants in the Salmon area has diminished greatly over time, and we seek to increase their usable habitat by planting milkweed in key locations to connect the existing, though limited, milkweed populations within the corridor.

2023 Seedball Monitoring

March 2024 was abnormally warm in the Salmon Valley, and most of our snow cover melted by the end of the month. Around the same time, we began revisiting our seedball treatment sites from 2023 to see what, if any, was hopefully growing. Because of our transects, seedballs from the year before were easy to locate and we were quickly excited by the results. Approximately 80% of the locatable seedballs along our transect near Panther Creek exhibited some amount of germination, many with 10-20 seedlings each.

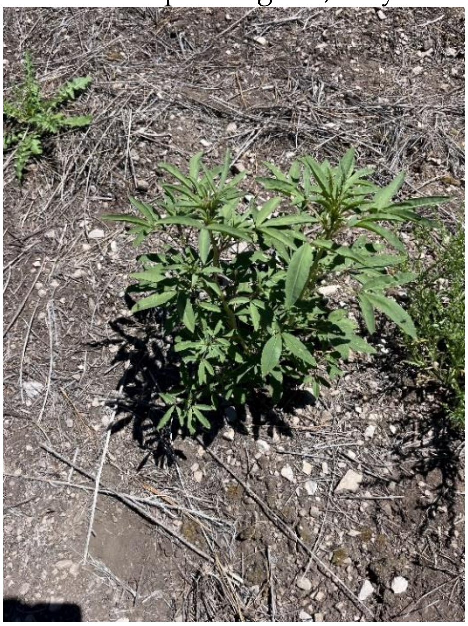


Unfortunately, a prolonged period of minimal precipitation and regular frosts during April and May resulted in significant seedling mortality. Upon revisiting these sites in early June, we observed far fewer surviving seedlings, many of which were already senescing due to drought.



Senescing seedlings are visible in the above photos. While revisiting the site in mid July, we observed no surviving seedlings and we believe conditions were simply too inhospitable this year for seedlings to effectively establish.

Other locations displayed different results. At our transect seedballs near Kriley Gulch and Buster Bulch, the harshest sites we applied treatments to in 2023, we observed no germination at all. However, Carmen Creek, we observed stellar success. During visits in early June, we discovered dozens of well developed *Cleomella serrulata* and *Grindelia squarrosa* seedlings. On harsher areas of the planting site, only *Grindelia squarrosa* was observed to have germinated.



Juvenile *Cleomella serrulata* plant.

Juvenile *Grindelia squarrosa* plants.

Revisiting the site later in July, we observed high seedling survival and robust plants. *Cleomella serrulata* is an annual and is expected to produce flowers on a yearly basis. *Grindelia* squarrosa is a short-lived perennial and does not typically produce flowers in its first year.

Flowering *Cleomella serrulata* plant.

Robust, adult *Grindelia squarrosa* plant.

Adaptive Management

From monitoring our 2023 treatments, it quickly became clear how difficult getting seeds to survive in the unforgiving conditions of the Salmon Challis National Forest was going to be. To negate the high levels of observed seedling mortality, we have to alter our strategy. We landed on a few takeaways and lessons from 2023:

- Less seed per seedball. Too much germination in one place may be leading to too much competition between seedlings.
- Sites with shallow or limited soil development will be our hardest challenges.
- We need to incorporate more technologies in our seedballs.
- Selecting our restoration sites ahead of time will allow us to avoid wasting time collecting seed from species that are not appropriate.
- We need to put far more seedballs out on the landscape. Increasing both our overall acreage and the density of plantings. The more seedballs we put out the more seed has the chance to germinate, grow, and establish.

Population Mapping and Phenology Monitoring

The ever-expanding field of seed science has revealed that locally collected native seed is of vital importance to revegetation efforts and overall seedling survival, especially in areas that display harsh or unique conditions. Salmon is undoubtedly both harshly arid and ecologically unique; at both high elevation and high latitude, we experience extremely hot and extremely cold temperatures yet surprisingly little rainfall with an average precipitation of 9-11 inches a year. With its steep topography, rocky soils, and orographic geography, these factors all coalesce into a difficult place for plants to grow. As such locally adapted genetics are of the utmost importance.

Phenology monitoring requires repeated site visits in order to determine the ideal timing for seed collection. Plant phenology is dependent on precipitation, temperature, elevation, aspect, soil, and seasonality making it difficult to accurately predict and can vary significantly from year to year. Often, this means that you may be able to collect seed from a species at Site B long after that species has entirely senesced at Site A, if the individual site conditions are different enough. This work requires a large amount of exploration and mapping, then if the site is deemed a priority, repeated visits to time collection efforts.

We learned a lot about what plant species would be appropriate for our efforts from last year's work, and our list of target species for collection was greatly amended. Our focus is still on rangeland and high desert plant species with an emphasis on early seral species and nitrogen fixers. However, species were removed from the list due to a variety of factors including poor suitability to restoration sites, low seed viability, and low germination rates. The goal is to create well rounded, balanced plant communities and this requires the use of forbs, grasses, and shrubs, including both annual and perennial species. Other factors such as fire adaptations, seasonality, and ground cover impacts were also considered. Our target list for the 2024 field season is shown below as well as the species we removed from 2023's list.

2024 Species Target List

Scientific Name	Common Name	Scientific Name	Common Name
Achillea millefolium	Western Yarrow	*Fritillaria pudica*	Yellow Bells
Achnatherum hymenoides ★	Indian Ricegrass	~~*Geum Triflorum*~~	~~Prairie Ground Smoke~~
Agoseris glauca	Pale Agoseris	*Grindelia squarrosa* ★	Curly Cup Gumweed
Allium sp.	Wild Onion	*Gutierrezia sarothrae*	Broomrape Snakeweed
~~*Antennaria sp.*~~	~~Pussy Toes~~	*Helianthus annuus* ★	Annual Sunflower
~~*Arnica cordifolia*~~	~~Heart Leaf Arnica~~	*Heterotheca villosa*	Hairy Goldenaster
~~*Arnica sororia*~~	~~Twin Arnica~~	*Ionactis alpinus*	Lava Aster
Artemisia arbuscula	Low Sagebrush	~~*Ipomopsis aggregata*~~	~~Scarlet Gilia~~
Artemisia dracunculus ★	Tarragon	*Lewisia rediviva* ★	Bitterroot
Artemisia tridentata	Big Sagebrush	*Lithospermum ruderale*	Western Stoneseed
Artemisia tripartita ★	Three Tip Sagebrush	*Lupinus caudatus*	Tailcup Lupine
Asclepias speciosa ★	Showy Milkweed	*Lupinus polyphyllus*	Bigleaf Lupine
Astragalus atropubescens ★	Kelsey's Milkvetch	*Machaeranthera canescens* ★	Hoary Tansyaster

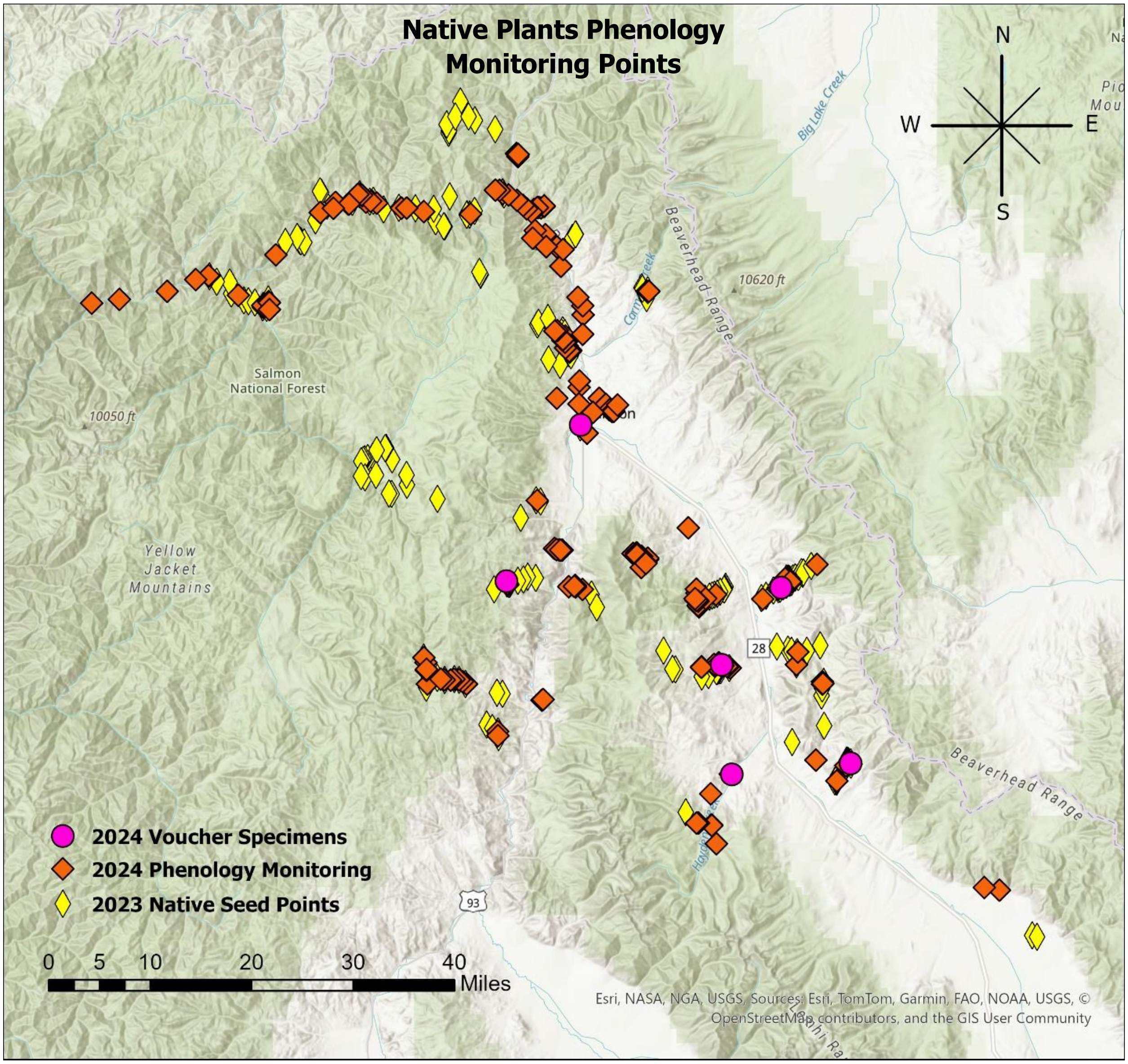
Scientific Name	Common Name	Scientific Name	Common Name
Astragalus beckwithii	Beckwith's Milkvetch	*Mentzelia dispersa*	Bushy Blazing Star
Astragalus purshii ★	Wooly Milkvetch	*Mentzelia laevicaulis* ★	Smooth Stem Blazing Star
Astragalus scaphoides	Bitterroot Milkvetch	*Oenothera villosa*	Hairy Evening Primrose
Balsamorhiza sagittata ★	Arrowleaf Balsamroot	*Oreocarya glomerata*	Northern Cyptantha
Calochortus eurycarpus	Mariposa Lily	*Oxytropis lagopus*	Haresfoot Locoweed
~~*Calochortus nitidus*~~	~~Broadfruit Mariposa Lily~~	*Panicum capillare*	Witch Grass
Camassia quamash ★	Camas	*Penstemon aridus*	Stiffleaf Penstemon
Chaenactis douglasii ★	Douglas' Dusty Maiden	*Penstemon deustus* ★	Hotrock Penstemon
Chrysothamnus viscidiflorus	Yellow Rabbitbrush	*Penstemon eriantherus*	Fuzzy Tongue Penstemon
~~*Cirsium cymosum*~~	~~Peregrine Thistle~~	~~*Penstemon lemhiensis*~~	~~Lemhi Penstemon~~
Cleomella serrulata	Rocky Mountain Bee Plant	*Phacelia hastata* ★	Silverleaf Phacelia
~~*Collinsia parviflora*~~	~~Maiden Blue-Eyed Mary~~	*Phacelia heterophylla* ★	Varileaf Phacelia
~~*Collomia linearis*~~	~~Pink Tiny Trumpets~~	*Phacelia linearis* ★	Linear Leaf Phacelia
~~*Crepis acuminata*~~	~~Tapertip Hawksbeard~~	~~*Phlox longifolia*~~	~~Longleaf Phlox~~
Elymus elymoides ★	Bottlebrush Squirreltail	*Plantago patagonica* ★	Woolly Plantain
Eremogone kingii	King's Sandwort	*Polanisia dodecandra*	Red Clammyweeed
Ericameria nauseosa ★	Rubber Rabbitbrush	*Pseudoroegneria spicata*	Bluebunch Wheatgrass
Erigeron compositus	Dwarf Leaf Fleabane	~~*Senecio integerrimus*~~	~~Western Groundsel~~
Erigeron linearis	Straight Leaf Fleabane	*Sporobolus cryptandrus* ★	Sand Dropseed
Erigeron pumilis ★	Shaggy Fleabane	*Stenotus acaulis* ★	Mock Goldenweed
Eriogonum microtheca	Slender Wild Buckwheat	~~*Tetradymia canescens*~~	~~Spineless Horsebrush~~
~~*Eriogonum ovalifolium*~~	~~Cushion Buckwheat~~	*Thelypodium laciniatum*	Lanceleaf Thelypody
Eriogonum strictum ★	Blue Mountain Buckwheat	*Townsendia parryi*	Parry's Ground Daisy
~~*Eriogonum umbellatum*~~	~~Sulfur Buckwheat~~	*Verbena bracteata*	Big Bract Verbena
Frasera albicaulis	White Stemmed Frasera	*Vulpia octoflora*	Six Weeks Fescue

☐ – Forb Species ☐ – Grass Species ☐ – Shrub Species

~~Species~~ – removed from list ★ – Priority

The species we decided not to target were removed from the list due to poor compatibility with the restoration site's specific parameters or for other reasons. In some cases, the soil types, elevation, or the aspect of the Salmon River Corridor's hillsides were not suitable for the selected species. In other cases, we learned that the average % of pure live seed from collections was extremely low. Lemhi penstemon was deemed inappropriate for collections because planting it in areas where one of its two parent species are present (*Penstemon cyaneus* and *Penstemon payettensis*) can lead to detrimental effects on overall population genetics. Species were designated as a priority due to ease of collection, % PLS output, and suitability for restoration sites.

Native Plants Monitoring Points



As of 12/31/2024, 194 locations were mapped and revisited at regular intervals to track the phenology of the target species located there. Sites can be found on the above map and are located on both BLM, FS, and IDFG lands.

2023's phenology tracking points are shown on the map to indicate locations that were deemed unnecessary and not revisited in 2024 or new areas we chose to explore that were not visited in 2023. Sites assessed for suitability and were deemed unnecessary include Deep Creek, Napias Creek, William's Creek, Hawley Creek, Ramsey Mountain, and Granite Mountain. Newly visited locations include Wagonhammer Creek, Henry Creek, the Middle Fork, Black Rock, Trail Gulch, Discovery Hill, Baby Joe Gulch, and Hayden Creek. Voucher collections are also shown on the above map though they have yet to be processed and identified.

Seed Collection

Protocol

As mentioned in previous reports, the Forest Service does not have an established protocol for seed collection and as such, this program had limited guidelines for how to operate during this phase. Instead, we were instructed to adapt the BLM's Seeds of Success (SOS) protocol to suit the program's needs. The SOS's protocols are well established and operates under the following principles:

- Seeds may only be collected from wild populations.
- Each collection must sample at least 50 individual plants.
- No more than 20% of ripe seed should be collected from a population.
- At least three populations must be sampled per species.
- Populations should be at least one kilometer apart.
- Ideal collections consist of at least 10,000 seeds.

These guidelines prevent over sampling plant populations and causing detrimental effects to population dynamics while ensuring that adequate genetic diversity is sampled during collections. All volunteers and technicians working with the program are trained to follow these rules and the only times the program breaks them is when we make opportunistic collections resulting in less than three populations sampled for a particular species by the end of the season.

Seed Collection Strategies

Plant species have evolved to produce seed in a remarkably diverse number of ways which translates into a diverse range of seed collection strategies. Even closely related species may require very different collection tactics. Some of our strategies may be found in the following list and more specific information can be made available upon request. As a general rule of thumb, seed that does not readily fall from its container is likely not ripe and should not be collected.

- Aster species like *Erigeron pumilus*, *Ionactis alpina*, and *Machaeranthera canescens* produce seed with fluffy pappus, creating a dandelion-like head from which ripe seeds are easily stripped off by hand. This is the case for most of the aster species we target with the exception of *Chaenactis douglasii* whose pappus is not made of capillary (hair-like) bristles but of papery chaff. Though the seed is collected in the same manner, it is much more difficult to clean. In future years we would like to try collecting these species with a backpack vacuum though we have yet to attempt it.
- Aster species in the sunflower tribe like *Balsamorhiza sagittata* and *Helianthus annuus* produce seeds without any pappus. We've found that clipping the dehiscent heads into a bucket or bag to be the most efficient collection method.
- Pea family species like *Astragalus purshii* or *Lupinus caudatus* produce seeds in pods that can either be picked up off the ground, pulled from the plant, or clipped off with shears. These seeds are not typically ripe until their pods begin to dehisce and open though, if

the pod is closed but you can hear the seeds rattling around inside, they are likely still ripe.

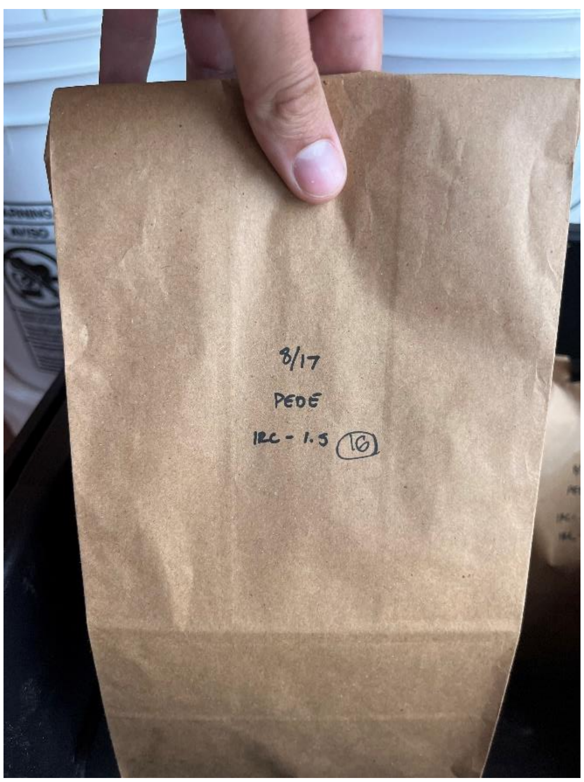
- *Penstemon* seeds grow in small pods that can be easily shaken out into a bag. However, the seeds are so small and numerous that we found it easier to clip entire stems to avoid losing wayward seeds.
- Plants like *Collomia linearis* or *Plantago patagonica* are either so sticky or hold on to their seeds so tightly, that the entire plant must collected to allow time to dry before seeds may be removed from their receptacles.
- Borages like *Oreocarya glomerata* or waterleaf species like *Phacelia heterophylla* produce fresh flowers along the end of the stem while older ones are actively fruiting further down making it difficult to time collections. Unripe seeds will not ripen if the stem is clipped so we found that lightly hand stripping the stems allowed ripe capsules to dislodge while leaving the unripe ones remaining. Wear gloves as their stiff bristles can irritate the skin.
- *Ericameria nauseosa* produces so much seed that placing a large bucket downwind and whacking the plant with a racket works excellently.
- Milkweed species cannot be collected until the pod has begun to open on their own, but the seed is much harder to collect and process once the seed has begun to freely fall from the pods making timing the collections critical. Rubber bands may be placed around unripe pods to prevent them from opening up until you return at a later date.
- Hand stripping was the best method we found for all grass species sampled.

***Astragalus aquilonius* plant with unripe seed pods. Seed is ripe when pods are dry and opening.**

Volunteer, Kenia, collecting stems from *Plantago patagonica* near Diamond Creek.

Technicians, Maggie & Max, collecting seed from *Ionactis alpina* near Reese Creek.

Ericameria nauseosa seed collected in large quantities by whacking shrubs with a racket.



Collection bags record the date, species collected, site name, and the # of individuals sampled.

Milkweed pod rubber-banded closed to prevent seed from escaping between site visits.

Seed Collection Results

Between June and November of 2024, the program collected seed from 37 different plant species sampling a total of 61,299 individual plants. 22 of the species collected this year were not sampled in 2023, demonstrating the level to which the program pivoted following our pilot year. With the additional help of technicians, interns, and volunteers, this season we were able to sample seed from over twice as many individual plants. Collections occurred between 6/12/2024 and 11/7/2024 at 68 unique locations. The following table provides data on seed collections from 2024; species are listed in the order they were collected. More information available upon request.

2024 Seed Collection Results

Scientific Name	Common Name	Type	Annual or Perennial	First Collection	Last Collection	# Plants Sampled
Erigeron compositus	Dwarf Leaf Fleabane	Forb	P	6/12/24	6/13/24	841
Achnatherum hymenoides	Indian Ricegrass	Grass	P	6/17/24	7/15/24	1842
Stenotus acaulis	Mock Goldenweed	Forb	P	6/18/24	10/24/23	1400
Elymus elymoides	Bottlebrush Squirreltail	Grass	P	6/25/24	7/16/24	1274
Vulpia octoflora	Six Weeks Fescue	Grass	A	6/25/24	6/26/24	6980
Townsendia parryi	Parry's Ground Daisy	Forb	P	6/25/24	--	10
Ionactis alpina	Lava Aster	Forb	P	7/1/24	7/11/24	271
Oxytropis lagopus	Haresfoot Locoweed	Forb	P	7/1/24	--	88
Astragalus aquilonius	Lemhi Milkvetch	Forb	P	7/2/24	--	1075
Erigeron pumilus	Shaggy Fleabane	Forb	P	7/3/24	7/9/24	3118
Chaenactis douglasii	Douglas' Dusty Maiden	Forb	P	7/3/24	7/11/24	1792
Phacelia heterophylla	Varileaf Phacelia	Forb	P	7/8/24	7/22/24	2351
Pseudoroegneria spicata	Bluebunch Wheatgrass	Grass	P	7/8/24	7/15/24	2918
Balsamorhiza sagittata	Arrowleaf Balsamroot	Forb	P	7/9/24	7/12/24	2174
Astragalus beckwithii var. sulcatus	Grooved Milkvetch	Forb	P	7/10/24	--	1
Astragalus atropubescens	Kelsey's Milkvetch	Forb	P	7/10/24	7/25/24	2679
Erigeron linearis	Desert Yellow Fleabane	Forb	P	7/11/24	--	14
Penstemon aridus	Stiffleaf Penstemon	Forb	P	7/15/24	8/20/24	2190
Astragalus purshii	Woolly Milkvetch	Forb	P	7/15/24	7/17/24	1013
Plantago patagonica	Woolly Plantain	Forb	A	7/17/24	8/8/24	20743
Eremogone kingii	King's Sandwort	Forb	P	7/17/24	--	1002
Achillea millefolium	Western Yarrow	Forb	P	7/18/24	--	527
Penstemon eriantherus	Fuzzy Tongue Penstemon	Forb	P	7/18/24	7/22/24	447
Oreocarya glomerata	Northern Cryptantha	Forb	P	7/22/24	--	378
Camassia quamash	Camas	Forb	P	7/22/24	7/23/24	762
Penstemon deustus	Hotrock Penstemon	Forb	P	7/24/24	8/1/24	663
Thelypodium Lanciniatum	Lanceleaf Thelypody	Forb	P	7/31/24	--	409

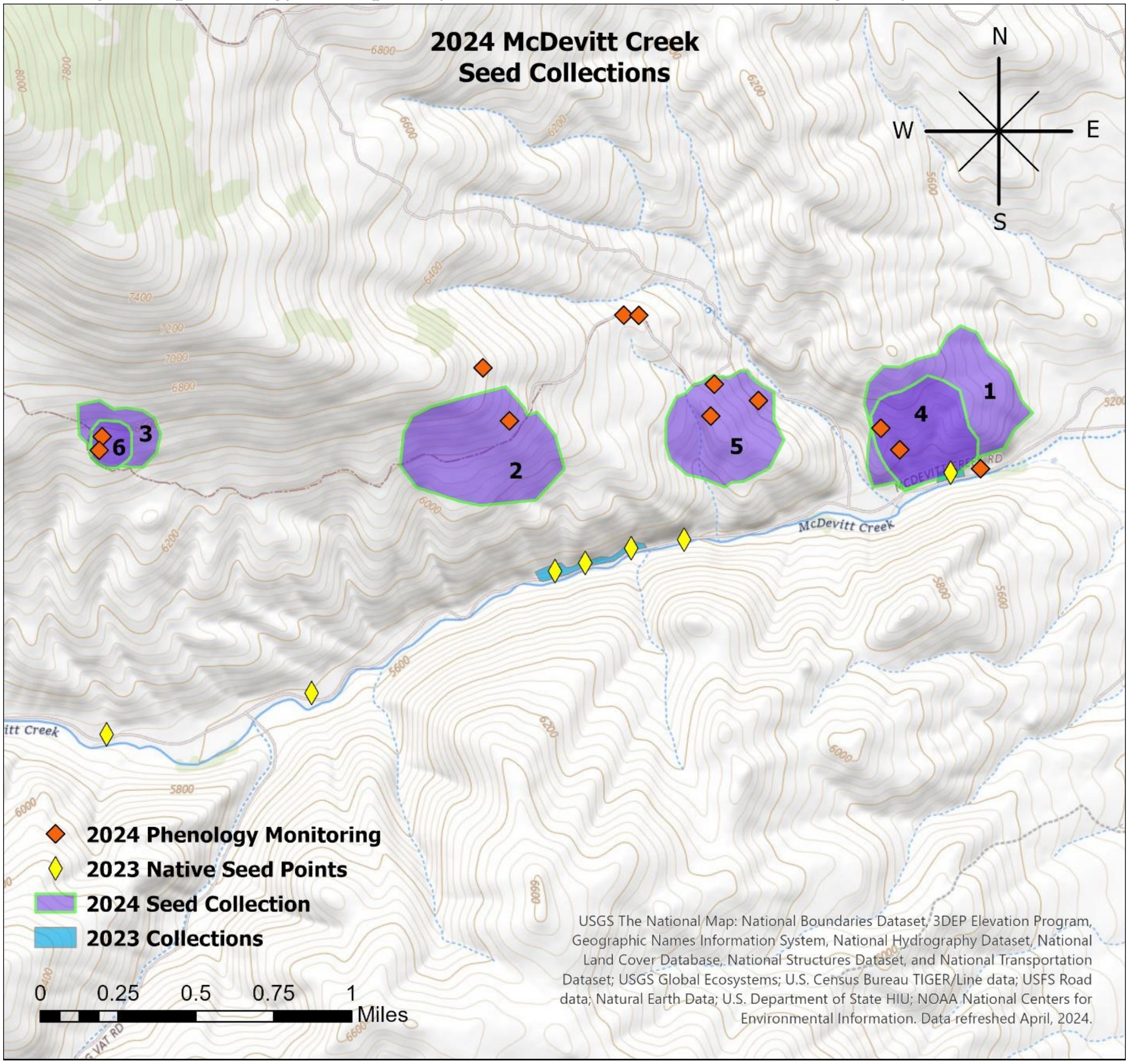
Scientific Name	Common Name	Type	Annual or Perennial	First Collection	Last Collection	# Plants Sampled
Helianthus annuus	Annual Sunflower	Forb	A	8/27/24	9/27/24	476
Verbena bracteata	Big Bract Verbena	Forb	P	8/27/24	9/23/24	67
Asclepias speciosa	Showy Milkweed	Forb	P	9/5/24	9/25/24	1265
Oenothera villosa	Hairy Evening Primrose	Forb	B	9/5/24	--	9
Grindelia squarrosa	Curly Cup Gumweed	Forb	B	9/23/24	--	449
Panicum capillare	Witch Grass	Grass	A	9/10/24	--	305
Ericameria nauseosa	Rubber Rabbitbrush	Shrub	P	10/2/24	10/16/24	156
Machaeranthera canescens	Hoary Tansyaster	Forb	P	10/15/24	10/20/24	381
Eriogonum strictum var. proliferum	Blue Mountain Buckwheat	Forb	P	10/16/24	10/17/24	539
Sporobolus cryptandrus	Sand Dropseed	Grass	P	11/7/24	--	690

Collection Notes

- Many collections were opportunistic in nature meaning we had not intended to come across that species that day but found it ready and in population sizes appropriate for collection. This is primarily shown in the table above by records lacking a "Last Collection" entry, though in some cases we simply got what we needed in one day and declined to collect more.
- Many species exhibited remarkably different population sizes from 2023 to 2024. *Phacelia linearis* was practically nonexistent on the landscape compared to what we observed in 2023. *Helianthus annuus*, *Ionactis alpina*, *Mentzelia laevicaulis*, *Mentzelia dispersa*, and *Astragalus scaphoides*, all species we found in high numbers in 2023 were extremely difficult to find upon revisits to those locations this year. Other species like *Machaeranthera canescens*, *Eriogonum strictum*, *Plantago patagonica*, *Vulpia octoflora*, and *Chaenactis douglasii* were found in extremely high numbers compared to the year prior.
    - This is important to note because it highlights how difficult it is to predict what will be on the landscape from year to year and how dependent each of our target species is on specific conditions that we don't fully understand.
- We used SVS's mailing list to put out a call for volunteers to collect seed from *Asclepias speciosa* as many people have it growing in their garden. Data from those collections are not listed in the table above as we lack the necessary information.
- Collections shown above in which under 20 species were sampled were not intended for use in 2024 seed mixes. They were mostly educational/opportunistic in nature.
- *Phacelia hastata* and *Phacelia heterophylla* can technically only be distinguished by their taproot, effectively killing the plant in order to determine its proper designation. Though the two species may have significant crossover in leaf shape, growth form, flower color, and habitat, we decided to ID based on which side of the scale the morphological features leaned. As such, all collections were IDed to *Phacelia heterophylla*.

Seed Collection Maps

Collection polygons were generated in the field using ArcGIS Field Maps. They are intended to be used for tracking various data points related to our field work including cost per acre. 115 unique collections were made spanning 1,539 acres during the 2024 field season. These maps illustrate our year-to-year shifts and the diversity of collections that can be made within the same region if phenology is frequently monitored and sites revisited regularly.



1 – 7/2/24 – Collected *Astragalus aquilonius* from 1075 plants at MCC-2 over 58.1 acres.

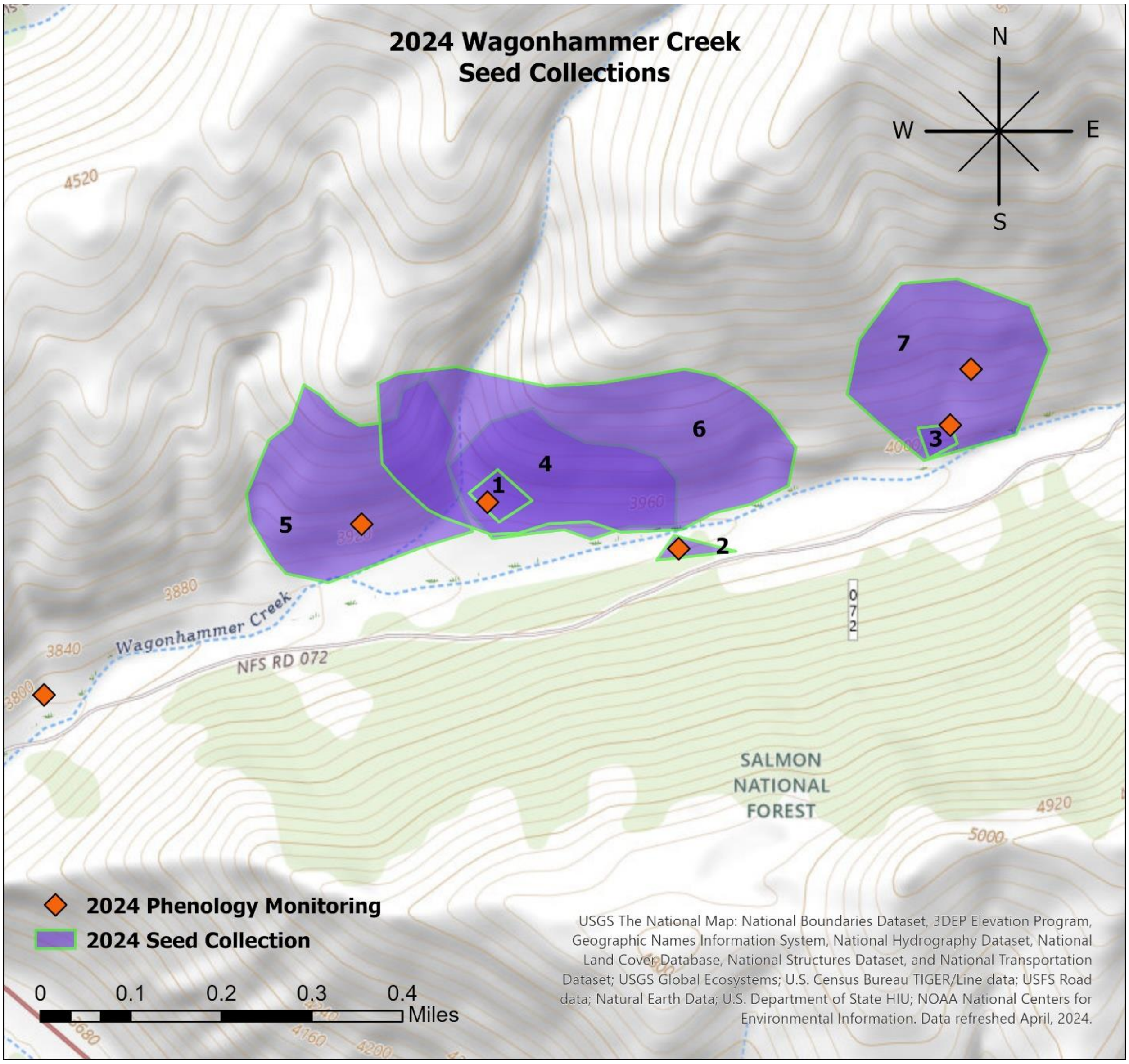
2 – 7/2/24 – Collected *Stenotus acaulis* from 63 plants over 47.21 acres.

3 – 7/2/24 – Collected *Stenotus acaulis* and *Ionactis alpina* from 482 plants over 13.86 acres.

4 – 7/22/24 – Collected *Phacelia heterophylla* from 812 plants at MCC-2 over 28.22 acres.

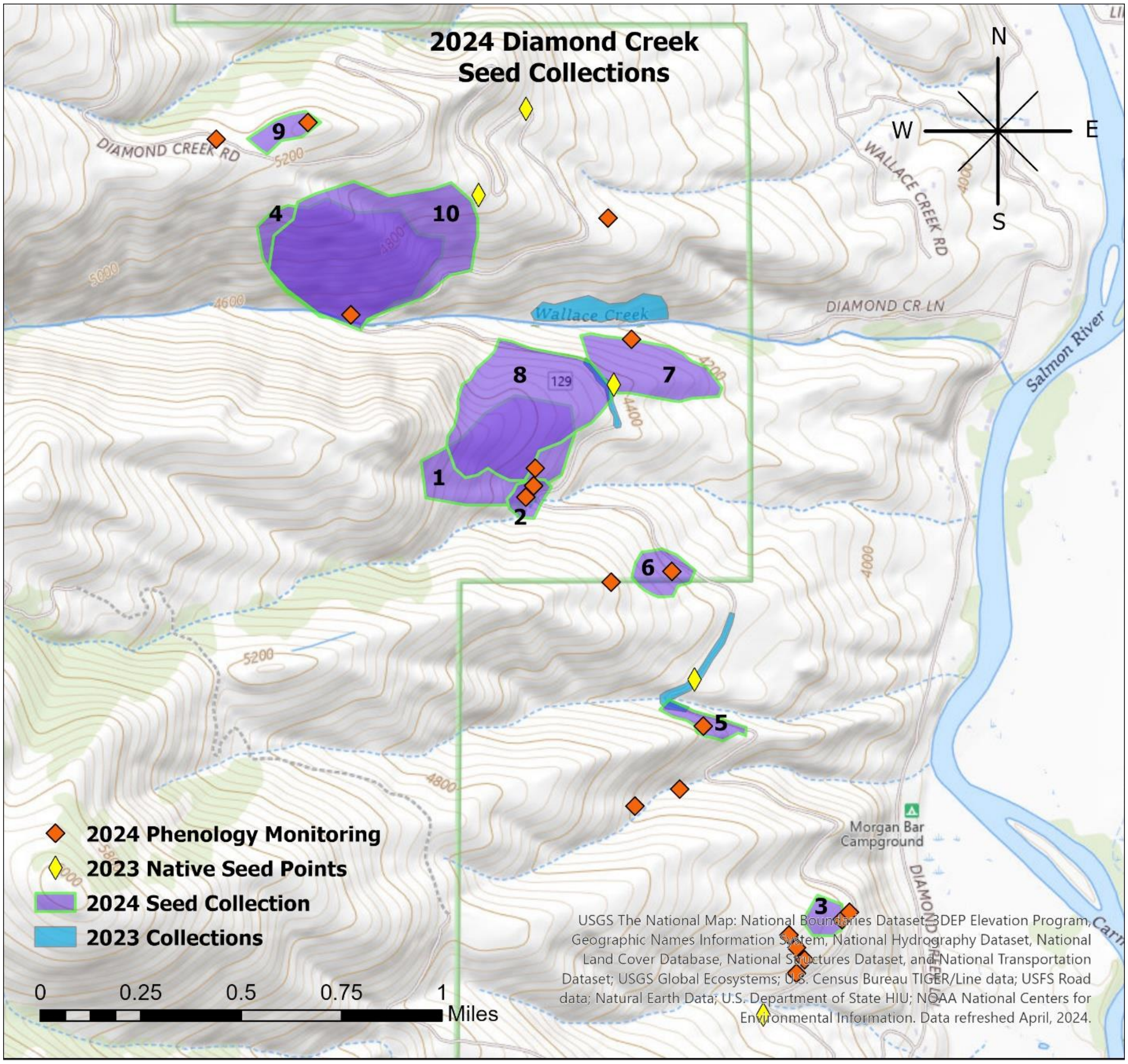
5 – 7/22/24 – Collected *Astragalus atropubescens* from 505 plants at MCC-3 over 33.78 acres.

6 – 8/20/24 – Collected *Penstemon aridus* from 612 plants at MCC-6 over 5.56 acres.



1 – 7/3/24 – Collected *Chaenactis douglasii* from 48 plants at WAH-2 over 0.64 acres.
2 – 7/3/24 – Collected *Erigeron pumilus* from 55 plants at WAH-4 over 0.31 acres.
3 – 7/3/24 – Collected *Chaenactis douglasii* from 16 plants at WAH-3 over 0.35 acres.
4 – 7/10/24 – Collected *Chaenactis douglasii* from 601 plants at WAH-2 over 7.86 acres.
5 – 7/11/24 – Collected *Chaenactis douglasii* from 628 plants at WAH-2 over 11.74 acres.
6 – 8/1/24 – Collected *Penstemon deustus* from 187 plants at WAH-2 over 21.6 acres.
7 – 8/1/24 – Collected *Penstemon deustus* from 148 plants at WAH-3 over 10.15 acres.

Wagonhammer creek was not visited for monitoring or collections in 2023, but we found it to be an extremely biodiverse site for many of our target species in 2024.



1 – 6/17/24 – Collected *Achnatherum hymenoides* from 12 plants at DIC-20 over 23.07 acres

2 – 7/10/24 – Collected *Chaenactis douglasii* from 13 plants at DIC-20 over 2.25 acres.

3 – 7/10/24 – Collected *Astragalus beckwithii* var. sulcatus from 1 plant at DIC-0 over 2.43 acres.

4 – 7/10/24 – Collected *Chaenactis douglasii* from 496 plants at DIC-35 over 31.18 acres.

5 – 7/10/24 – Collected *Astragalus atropubescens* from 223 plants at DIC-65 over 2.61 acres.

6 – 7/17/24 – Collected *Plantago patagonica* from 6,562 plants at DIC-5 over 4.13 acres.

7 – 7/22/24 – Collected *Oreocarya glomerata* and *Astragalus atropubescens* from 216 plants at DIC-21 over 11.73 acres.

8 – 7/25/24 – Collected *Astragalus atropubescens* from 942 plants at DIC-21 over 29.5 acres.

9 – 8/8/2024 – Collected *Plantago patagonica* from 10,960 plants at DIC-72 over 2.84 acres.

10 – 10/15/24 – Collected *Machaeranthera canescens* and *Ericameria nauseosa* from 353 plants at DIC-35 over 42.63 acres.

Seed Cleaning

Before collected seed can be put towards any specific purpose, it must be processed, dried, and cleaned of debris, chaff, pappus, and other detritus that is collected along with the seed during field work. Because species produce fruit in such a wide variety of ways, we employ a plethora of strategies, tools, and methods in order to process them. In general, the process boils down to whether you can separate the seed from whatever extraneous material it is contained within, connected to, or mixed in with. Material is separated via weight, size, or shape and materials can be removed by smashing or with abrasion.

Seed cleaning is incredibly time intensive, especially when lacking the expensive, heavy equipment larger-scale programs or commercial producers employ. To make up the difference in equipment, we've designed and constructed our own DIY tools to avoid spending large sums of money on outsourcing the work. A huge thank you to Jordan Schaeffer for helping with these builds. Lower tech equipment like sieves were purchased. As the amount of seed we collect increases, the necessity of efficient cleaning technology becomes more and more valuable.

Aspirator

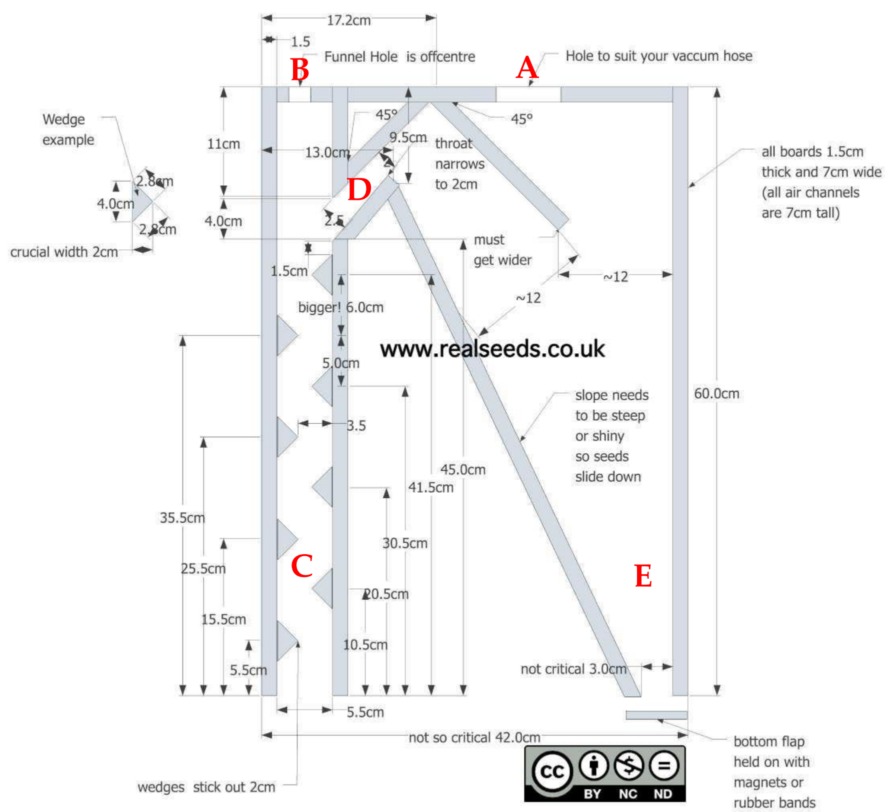
We learned from our seed cleaning experience in 2023 that in order to achieve a higher level of purity, we needed a way to separate material by weight. With this in mind, we considered purchasing equipment to meet this need but decided to go a DIY route instead. We came across these designs while attending the National Native Seed Conference and built two iterations.

A vacuum is attached with a nozzle at **A** to apply suction to the system.

Seed is dropped into a funnel at **B** and exposed to airflow coming up through **C** as it tumbles through the zigzags.

Lighter material is pulled through the thin channel at **D,** then deposited in the larger chamber at **E**.

Heavier material falls through the bottom of the chamber coming out at **C**.



- We varied our design from the original by adding sturdy legs and attachments to keep the vacuum hose in place.
- We learned quickly that static electricity was going to be a significant problem; in our second iteration, we swapped out the plastic front with a glass one and used silicone adhesive to fill the gaps. This kept the static build up down and stopped small seeds from getting lodged in the aspirator.
- Next, we needed a way to modulate the strength of the suction. We purchased two vacuums of different strength to vary the overall power of the airflow and the hole with the cover in the upper right corner works as a more nuanced regulator of suction strength.
- The device works excellently to remove both lighter materials and empty seeds.

Drum Thresher

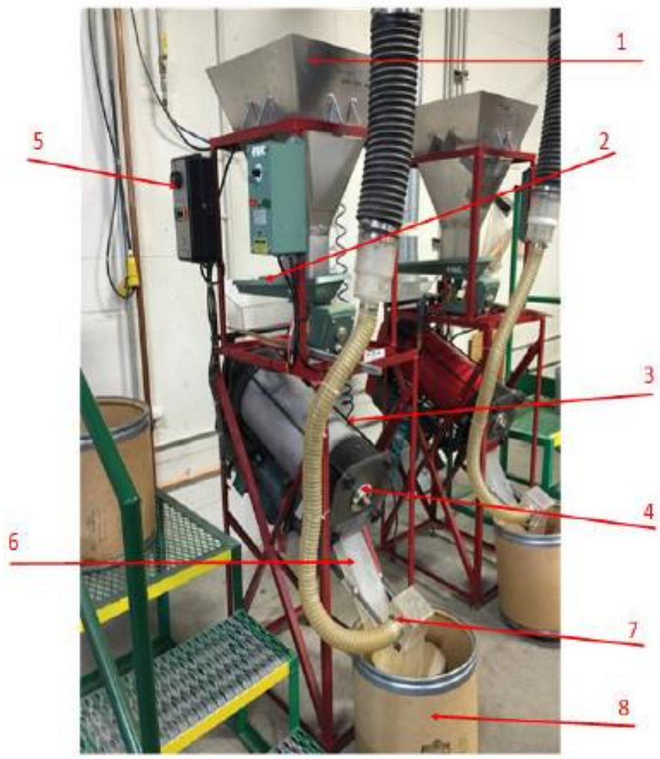
Built in 2023 with suggestions by Jessica McAleese from Swift River Farms; chains approximately the same radius as the drum are welded to a long drill bit and fed through holes cut into the lid. The contraption smashes pods and breaks down materials quickly and efficiently. As the drum is only 5 gallons, it's limited in the amount it can process at a time.

Inside the drum thresher: chains are welded in rows to the bottom of the drill bit.

Katie Baumann using the drum thresher to break open *Astragalus* seed pods.

Dewinger

In the search to find an automated way to remove pappus from aster seeds, we were directed to plans for the Missoula Dewinger by Kayla Herriman, the National Native Seed Specialist for the USDA. Developed by the Research & Development branch of the US Forest Service, it was originally designed for dewinging conifer seeds but works perfectly for asters too.



This machine allows for seed extraction that can be tailored to the physical properties of the seed based on speed (of the feeder and the flaps), angle, and roughness of the liner. A larger hopper (1) feeds material along a vibrating channel into the machine (2). There is a lined cylinder (3), which we have various textures to choose from, that encloses a central shaft with gum rubber flaps (4). A variable speed motor powers the shaft (5). The angle of the cylinder can also be adjusted so that material flows quickly or is held in longer. The steeper the tilt the shorter the processing time. The dewinged seed flows out the end of the cylinder (6) and is exposed to an overhead vacuum system (7) that allows fine inert material to be removed before it falls into the container (8).

We landed on a simplified design based on our drum thresher which employs a rubber lined drum and exchanges the welded chain drill bit for one with a large rubber flap rivetted in place. Our version does not allow for continuous processing and can only manage a single batch at a time, but its simplicity of construction and use far outweighs this loss in efficiency.

In order to modulate the speed of the device, we purchased a Milwaukee mud mixer with a variable speed dial. In future years we have discussed building a tilted stand but as it is now, the device easily shaved off hundreds of hours of rubbing seed by hand. We could not be happier to have it in our arsenal.

Other Equipment

- Set of sieves from Seedburo of different shapes and sizes. Sizes include:
    - Wire Mesh – 30x30 and 40x40
    - Special Round Perforations – 2.5/64", 8/64", 12/64", and 1/12"
- Set of 8 square-holed sieves from Strictly Medicinal Seeds, numbered 1-8.
- Rubber mallet and Ziploc bags for smashing seed pods.
- Rubber welcome mats and trowels for rubbing seed.
- Wooden/sandpaper box for rubbing seed with more abrasion.
- Box fans for winnowing (air screening) seed.

Seed Cleaning Methods

The following lists the methods we employed to clean our various seed collections. Species with very low volume collections are not listed below. Methods will be uploaded to the Reforestation, Nurseries, & Genetic Resources (RNGR) plant propagation database. The site provides cleaning methods for different species and has been extremely helpful in the past for providing guidance. Immediately following collections, seed is treated with a low-grade insecticide (Hotshot Pest Strips) as per our risk management strategy. This kills any harmful insects that can cause infections to handlers during the seed cleaning process.

Achillea millefolium – Drum thresh seed heads then sift with 1/12". Sift out dust with 30x30 mesh sieve then with square #7 to remove large chaff. Aspirate at 40% to remove empty seeds and light chaff.

Achnatherum hymenoides – Thresh collected materials. Winnow in place while rubbing material in hands with gloves on to break down stems and dislodge seed. Sieve repeatedly with square #1 and 12/64" to remove large sticks. Aspirate low to remove dust then pick out remaining large sticks. Aspirate at 90% to remove empty seeds.

Artemisia dracunculus – Shake seedheads in sealed box to dislodge seeds. Sieve w/1/12 to remove large stems/chaff. Aspirate at 5-10% to remove fine dust and unripe seed. Sieve with 30x30 to remove remaining large chaff.

Asclepias speciosa – Remove seed from pods by hand. Run through the dewinger to separate seed from its pappus. Pick through fluff to dislodge any captured seed and remove any large chaff by hand. Aspirate at 25% to remove empty seed.

Astragalus aquilonius – Drum thresh pods then sift with 12/64" to remove seed. Place remaining pods in Ziploc and smash with rubber mallet. Sift with 8/64" to remove chaff, then with 30x30 to remove dust. Aspirate at 50% to remove empty seed and any remaining chaff.

Astragalus atropubescens – Sieve with 8/64" to remove loose seed. Drum thresh remaining pods then place in Ziploc and smash with rubber mallet to dislodge any remaining seed Sift with 8/64" then repeat the previous steps until satisfied that all seed has been removed from pods. Sieve with

40x40 to remove dust then aspirate at 30%. Aspirate again at 50% to remove empty seeds and chaff. Sift with square #6 to remove smaller material then square #4 repeatedly to remove additional chaff.

Astragalus purshii – Drum thresh pods to dislodge sees these place in Ziploc and smash with rubber mallet to remove any stragglers. Sift with 8/64″ and 1/12 to remove most fluff. Large chunks can be removed by hand easily. Aspirate at 60% to remove chaff and empty seeds.

Balsamorhiza sagittata – Drum thresh seed heads. Sift with 12/64″ to remove large chaff. Sift with 30x30 mesh to remove most fluff. Aspirate at 50% to remove lighter material then again at 70% to remove large material and empty seed.

Camassia quamash – Dislodged seed from seedheads with drum thresher. Sieved with 12/64″ to remove large chaff then aspirated at 50% to remove lighter chaff. Aspirated again at 70% to remove empty seeds.

Chaenactis douglasii – Remove small amounts of cheatgrass by hand before processing. Pappus on this species is stiff and chaffy; it resists removal by rubbing but the dewinging removes it moderately well. Sieve with 1/12″ to remove any large sticks. Aspirate on low to remove fine dust then again at 70% to separate empty seeds.

Elymus elymoides – Dewinger removes awns moderately well but some still resist fully breaking off seeds. Seed is then aspirated at 50% to remove unwanted material.

Eremogone kingii – Drum thresh pods, sift with square #6 to sort out loose seed. Smash remaining pods w/ rubber mallet. Abrade remaining pods w/ sandpaper box to open/dislodge any stuck seed. Sift with square #6 repeatedly. Sift with 40x40 wire mesh to remove any dust. Sift with square #3 to remove sticks then aspirate at 25% to remove small chaff and unripe seed. If this does not meet desired cleanliness, material can be rubbed repeatedly with rubber mat to break down chaff, then run through the aspirator again.

Ericameria nauseosa – Run through the dewinger to remove pappus and break down chaff. Aspirate at 25% then sift with #4 to remove chaff.

Erigeron compositus – Run through dewinger to remove pappus. Rub on rubber mat to remove any remaining pappus. Aspirate at 30% to remove dust and small chaff.

Erigeron pumilus – Run through dewinger to remove pappus. Winnow in place using box fans to remove fine dust material. Sieve with 8/64″ to remove larger inert matter. Aspirate at 50%. Must be run 3-5 additional times as more seed keeps falling out. Sieve with square #7 to remove smaller contaminants.

Grindelia squarrosa – Seed is extremely sticky, allow proper time for material to dry out as much as possible. Once dried sufficiently, shake clipped heads in a sealed box to dislodge seed. Sieve with 1/12″ to separate seed and chaff from the stems and heads. Remove heads and repeat above steps x3. Pick through heads using metal prong to dislodge any remaining seed. Sieve heads with 1/12″ again, forcing clumped seeds through sieve. Aspirate at 70% to remove further chaff.

Helianthus annuus – Use thumbs to rub seed out of dried flowerheads. Sieve through 8/64″ then 1/12″ to remove large chaff. Aspirate at 75% to remove smaller chaff and empty seeds.

Ionactis alpina – Run seed through dewinger then aspirate at 50% to remove dust and chaff.

Machaeranthera canescens – Run seed through dewinger then aspirate at 50% to remove dust and chaff.

Oenothera villosa – Run seedpods through drum thresher to dislodge seed. Sieve with 8/64″ to remove large chaff. Aspirate at 50% to pull out smaller chaff.

Oreocarya glomerata – Rub seed to break apart seed still attached to each other. Sieve with 12/64″ to remove large chaff. Aspirate at 65% then sieve again with 8/64″.

Oxytropis lagopus – Smash pods with rubber mallet. Sift with 40x40 wire mesh to remove dust. Aspirate at 40% to remove chaff.

Panicum capillare – Strip any remaining seeds on stems by hand. Sieve with 8/64″ to remove stems. Aspirate at 50% to remove chaff and unripe seeds.

Penstemon aridus – Drum threshed pods and stems to dislodge seed. Sift with 1/12″ to remove stems and other inert matter. Rub remaining material to further break down chaff. Sieve with 40x40 wire mesh to remove dust, then aspirate at 25%.

Penstemon deustus – Drum thresh pods and stems. Sieve with 12/64″ to remove seed pods. A high percentage of seed pods will remain closed for years even after seed ripens. Smash the removed pods in Ziploc bags with the rubber mallet to break them open and dislodge seed. Sieve with 30x30 wire mesh to remove large chaff then again at 40x40 to remove dust. Aspirate at 30% then higher if needed to remove small chaff from seed.

Penstemon eriantherus – Drum threshed pods and stems to dislodge seed. Sift with 1/12″ then 8/64″ repeatedly to remove stems and other inert matter. Aspirate at 50% then again at 75%. Sifted again with 1/12″.

Phacelia heterophylla – Drum threshed to break open seed pods. Sieve with 1/12″ to remove unopened pods and stems. Pods that will not break open during the threshing process may need to be rubbed on rubber mat. Winnow in place to remove large quantities of dust then aspirate at 30% to remove small chaff. Sieve with 1/12 again to remove any sticks and cheatgrass seed. Aspirate again at 50% to reach desired cleanliness.

Plantago patagonica – Thresh all stems. Sieve with 1/12″ to separate unbroken stems and pods from seed, fluff, and small chaff. Remove smaller material and set it aside in bin#1. Sieve the remaining stems/pods/chaff with 12/64″ to separate unopened pods from intact stems. Move unbroken pods to bin #2. Repeat this process to continue threshing stems, dislodging seeds, then separating unopened pods until all stems are broken down. Rub the unopened pods from bin #2 to dislodge seed, once all pods are opened, sieve material with 1/12″ and add to bin #1. Aspirate material from bin #1. This requires multiple attempts to continue removing seed from the inert matter. Repeatedly thresh, sieve, aspirate, sieve, rub and aspirate.

Sporobolus cryptandrus – Sieve with 1/12″ then 30x30 to remove large chaff. Winnow in place to remove fluff. Aspirate on very low to achieve desired cleanliness.

Stenotus acaulis – Run seed through dewinger to remove pappus. Aspirate on low to remove dust and fine material. Sieve with 12/64″ repeatedly to remove large chaff. Rub material to break down chaff then aspirate again on low to remove dust, chaff, and unripe seed.

Thelypodium laciniatum – Sift with 1/12″ to remove chaff and pods. Sift with 40x40 wire mesh to remove dust and smaller unripe seed. Seed is extremely clean and does not need to be aspirated.

Verbena bracteata – Remove pods/stems/leaves from stems by hand. Sieve through square #7 to separate seeds and small chaff from leaves, chaff, unopened pods, and stuck together seed. Rub the latter with rubber mat to open pods and break apart clumped seed. Sieve with 40x40 wire mesh to remove dust. Aspirate to remove chaff and empty seed. Sieve with square #7 to remove any remaining unseparated seed and sticks. Crush in Ziploc with rubber mallet. Aspirate again to finish. Any large chaff remaining can be easily picked out by hand.

Vulpia octoflora – Removed small amounts of cheatgrass seed by hand. Sieved with 1/12″ to remove chaff.

Any further seed cleaning strategies missing from this list can be provided upon request. Additionally, we would be more than happy discuss our equipment designs or methods. Seed cleaning requires us to constantly shift and pivot in order to meet programmatic needs.

Seed Cleaning Results/Measurements

After seed is cleaned, we calculate the percent purity of the lot. This is done by counting out three sets of 100 seeds. While counting, any inert matter or unwanted material like chaff or unripe seed that is mixed into the seed lot is included and each sample is weighed. The unwanted material is then removed from each sample and weighed again. With three sets, we have a large enough sample size to average our calculations and make an assumption about the entirety of the seed lot. The following is an example of the calculation:

Species	Set #	Mass w/ chaff (g)	Mass w/o chaff (g)	% Purity
Balsamorhiza sagittata	#1	0.530 g	0.489 g	0.489/0.530 = 0.923 = 92.3%
	#2	0.526 g	0.430 g	0.430/0.526 = 0.817 = 81.7%
	#3	0.500 g	0.472 g	0.472/0.500 = 0.944 = 94.4%
				(92.3+81.7+94.4)/3 = **89.5%**

This information allows us to determine % Purity but also give us the mass of a single seed and from there, the number of seeds within a specific volume. This is extremely useful information in designing seed mixes. For most commercially available seed, this information is readily available, however, most of our collections are niche enough that we are unable to find this information through our many sources. The following table lists relevant data:

Cleaning Measurements

Species Name	Total Mass Cleaned		Purity	Mass of 1 Seed	Seed/lb.	Seed/tsp	Seed/cup
Achillea millefolium	80.295 g	0.177 lb.	99.9%	0.000083 g	5,443,115	17,436	836,928
Achnatherum hymenoides	58.150 g	0.128 lb.	97.8%	0.001803 g	251,530	5,030	241,431
Artemisia dracunculus	5.729 g	0.013 lb.	65.6%	0.000067 g	6,803,894	46,110	2,213,280
Asclepias speciosa	836.965 g	1.845 lb.	98.6%	0.005827 g	77,848	195	93,64
Astragalus aquilonius	561.606 g	1.238 lb.	93.7%	0.002970 g	152,725	1,254	60,170
Astragalus atropubescens	275.916 g	0.608 lb.	89.6%	0.002377 g	190,853	1,279	61,404
Astragalus beckwithii	0.556 g	0.001 lb.	99.9%	0.004484 g	101,161	441	21,153
Astragalus purshii	52.154 g	0.115 lb.	92.5%	0.002440 g	185,899	1,537	73,784
Balsamorhiza sagittata	593.782 g	1.309 lb.	89.5%	0.004637 g	97,827	276	13,251
Camassia quamash	108.752 g	0.240 lb.	99.4%	0.002363 g	191,929	1,243	59,672
Chaenactis douglasii	121.714 g	0.268 lb.	98.0%	0.000930 g	487,734	902	43,303
Eremogone kingii	3.871 g	0.008 lb.	21.1%	0.000487 g	--	2,821	135,386
Erigeron compositus	18.361 g	0.040 lb.	95.4%	0.000237 g	1,916,590	5,024	241,149
Erigeron linearis	0.315 g	0.001 lb.	99.9%	0.000477 g	951,590	2,199	105,533
Erigeron pumilus	218.701 g	0.482 lb.	95.2%	0.000063 g	7,161,993	16,405	787,453
Grindelia squarrosa	499.738 g	1.102 lb.	93.5%	0.000860 g	527,434	2,938	141,042
Helianthus annuus	185.438 g	0.409 lb.	94.7%	0.005923 g	76,577	434	20,853
Ionactis alpina	0.877 g	0.001 lb.	83.5%	0.000540 g	839,987	1,660	79,704
Oenothera villosa	7.021 g	0.015 lb.	100%	0.000407 g	1,115,392	4,375	209,980
Oreocarya glomerata	48.634 g	0.107 lb.	91.6%	0.001320 g	343,631	1,448	69,527
Oxytropis lagopus	4.025 g	0.009 lb.	99.8%	0.001647 g	275,461	2,421	116,230
Panicum capillare	53.423 g	0.118 lb.	99.9%	0.000607 g	747,681	3,462	166,180
Penstemon aridus	30.855 g	0.068 lb.	71.1%	0.000120 g	3,779,941	20,381	978,267
Penstemon deustus	474.902 g	1.047 lb.	76.7%	0.000060 g	7,559,882	41,167	1,976,000
Penstemon eriantherus	83.043 g	0.183 lb.	87.9%	0.000967 g	469,234	2,090	100,320
Phacelia heterophylla	455.731g	1.005 lb.	99.3%	0.000520 g	872,294	6,466	310,369
Plantago patagonica	812.827g	1.792 lb.	91.9%	0.000720 g	629,990	4,203	201,733
Sporobolus cryptandrus	28.721 g	0.063 lb.	95.9%	0.000120 g	3,779,941	--	--
Stenotus acaulis	15.715 g	0.035 lb.	86.1%	0.000833 g	544,311	1,152	55,296
Thelypodium laciniatum	158.193 g	0.349 lb.	83.4%	0.000133 g	3,401,947	19,983	959,160
Townsendia parryi	1.111 g	0.002 lb.	97.8%	0.001450 g	312,823	679	32,596
Verbena bracteata	43.428 g	0.096 lb.	98.4%	0.000407 g	115,392	6,914	331,869
Vulpia octoflora	154.104 g	0.340 lb.	95.9%	0.000313 g	1,447,637	3,913	187,813

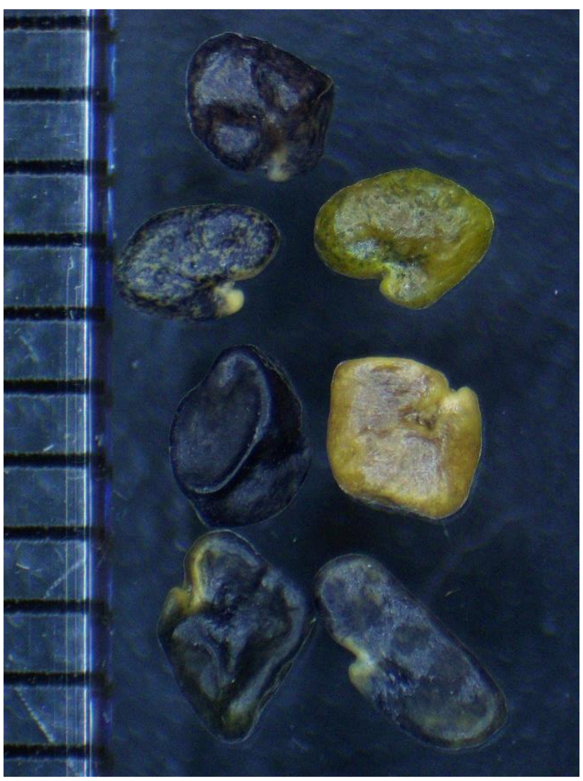
The seed per pound column illustrates how widely seed mass differs between species.

Hi-Res Seed Imagery

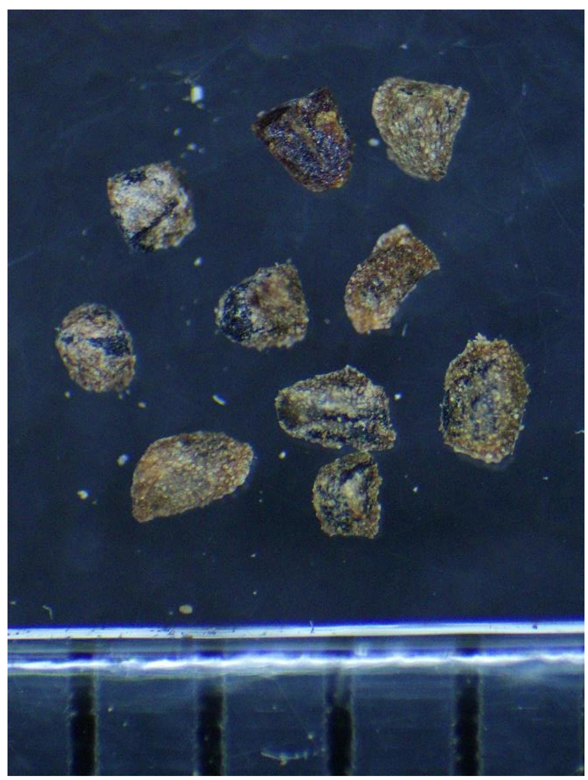
These photos were captured using a stereomicroscope:

***Phacelia heterophylla* seed.**

***Erigeron pumilus* seed.**



***Astragalus atropubescens* seed.**



***Penstemon deustus* seed.**

***Helianthus annuus* seed.**

***Achillea millefolium* seed.**

***Crepis acuminata* seed.**

***Achnatherum hymenoides* seed.**

Seed Mix Summary

Significant work was put into designing our seed mixes. With the hillsides of the Salmon River Corridor as our primary restoration site, we tailored our mixes to this location. Our goal this year was to limit the number of species within each mix, selecting ones that have been observed to cohabitate well or growing within the same substrate. We first selected species that would be suitable for every mix, to give a base to build from, then designed 10 different mixes, some with only a single primary species selected. 33 out of 37 species collected this year were utilized in our mixes. Mixes are listed below:

- Base Mix – *Vulpia octoflora*, *Phacelia linearis*, *Plantago patagonica*
- Mix 1 – **"GRSQ"** – *Grindelia squarrosa*
- Mix 2 – **"Rocky1"** – *Stenotus acaulis*, *Ionactis alpina*, *Penstemon aridus*, *Astragalus atropubescens*
- Mix 3 – **"Rocky2"** – *Chaenactis douglasii*, *Eriogonum strictum* var. *proliferum*, *Artemisia dracunculus*, *Achillea millefolium*
- Mix 4 – **"Rocky3"** – *Thelypodium laciniatum*, *Penstemon deustus*, *Phacelia heterophylla*
- Mix 5 – **"Rocky4"** – *Oreocarya glomerata*, *Penstemon eriantherus*, *Astragalus purshii*, *Eremogone kingii*, *Oxytropis lagopus*
- Mix 6 – **"Rocky5"** – *Erigeron pumilus*, *Erigeron compositus*, *Helianthus annuus*
- Mix 7 – **"ERNA"** – *Ericameria nauseosa*, *Elymus elymoides*, *Achnatherum hymenoides*
- Mix 8 – **"BASA"** – *Balsamorhiza sagittata*, *Elymus elymoides*, *Achnatherum hymenoides*
- Mix 9 – **"ASSP"** – *Asclepias speciosa* (does not include base mix)
- Mix 10 – **"Road"** – *Oenothera villosa*, *Achillea millefolium*, *Helianthus annuus*, *Verbena bracteata*, *Grindelia squarrosa*, *Elymus elymoides*, *Panicum capillare*, *Achnatherum hymenoides*, *Sporobolus cryptandrus*, *Pseudoroegneria spicata*

Seedballs

Seedball Technology

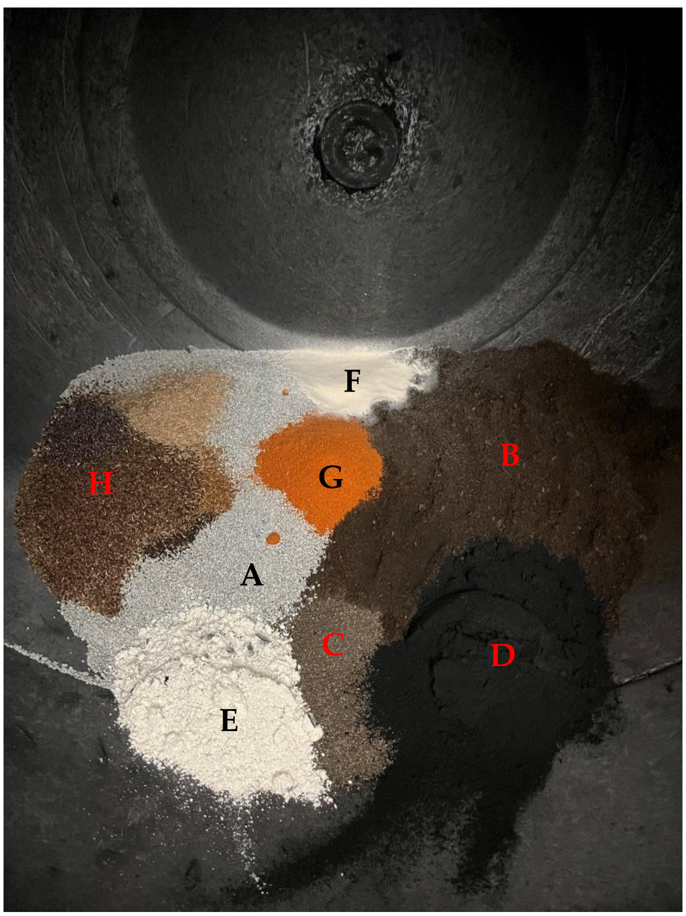
Seedballs are an ancient technology used throughout history for arid land plantings. First developed in ancient Egypt, and rediscovered in Japan, they have been recently adapted for use in restoration and are widely used across West Africa. Seedballs are a conglomeration of clay, soil, compost, and seed. They are designed to time seed dispersion with a germination event, protecting seed from the elements and predation while providing valuable nutrients to new seedlings. They are also ideal for seed mixes with a variety of planting depth requirements. The Salmon-Challis National Forest and surrounding country consists of extremely dry and steep hillsides with a long history of anthropogenic disturbance resulting in shallow soil and unbalanced plant communities. Seedballs were selected over other revegetation strategies due to these challenges to give our seed and any potential seedlings their best chance of survival.

With the guidance of Dr. Elise Gornish from University of Arizona, we developed our own seedball protocol and mix. In 2023, we followed her base design of 5 parts clay, 3 parts compost, and 1 part seed. This year, we researched and identified new technologies that would give our seedballs an extra boost and altered our recipe accordingly.

- **Health Soil** – Soil dug at the same locations seed was collected. Ideally contains symbiotic microbiota and fungi important for nutrient exchange via plant roots.
- **Cayenne Pepper** – Commonly used to ward of animals and insects.
- **Activated Carbon** – Capable of absorbing and deactivating any residual herbicides in the soil and its extreme porosity is good water retention.
- **Diatomaceous Earth** – This highly abrasive powder is another effective method for warding off insects and adds additional porosity for more water retention.
- **Microalgae Soil Amendment** – Mixed into the water used for our seedballs, it stimulates microorganisms within the soil, priming them for nutrient exchange.
- **Agar-Agar** – An excellent binder and a good substrate for microorganisms to flourish.

Seedball Production

In order to increase our total output to levels required to scale the program up to meet our goals, we transitioned away from hand rolling to mass production using a cement mixer. With a one part equaling one cup, this recipe produces approximately 2000 seedballs ranging from ¼ - 2 cm in diameter. Lastly, we learned that using 1 part seed was not necessary. Since most of our species have relatively small seed, using the original recipe would result in far too much seed per ball, creating an undesirable level of competition between seedlings. Instead, we focused on controlling the amount of seeds per ball to between 40-50.



Seedball ingredients combined in our cement mixer.

A) 20 parts clay
B) 12 parts compost
C) 4 parts health soil
D) 4 parts activated carbon
E) 2 parts diatomaceous earth
F) ½ part agar-agar
G) ½ part cayenne pepper
H) Seed mix with ≈ 200,000 seeds.
≈ 2 gallons of water mixed with 16 oz of microalgae soil amendment per gallon.

Producing seedballs with a cement mixer proved to be a learning experience. The following steps represent the best strategy we came up with:

1. Combine clay, compost, seed, and soil in the cement mixer then turn it on.
2. Slowly add water. Use a mist setting on your nozzle to ensure you are evenly applying moisture.
3. Once the mixture becomes lightly wet, begin to add in the other ingredients one by one, adding more water in between. This ensures the finer materials don't spit particulates from the drum. We followed the order: Agar-agar → Activated carbon → Cayenne pepper → Diatomaceous earth.
4. Continue slowly adding water, pausing every so often to allow the moisture to spread evenly.
5. Once sufficiently wet, balls will begin to form. Anything too large should be broken up to prevent grapefruit sized balls from forming.
6. Repeatedly add small amounts of water and break up the too-large balls. The key here is to move slowly.
7. Once all loose material has been incorporated and balls are a sufficient size, add 1-2 cups of a mix of activated carbon and soil to reduce any excess stickiness, giving the balls a good coating of material.
8. Once the batch is completed, seedballs should be laid out on tarps for at least two days to dry. Ensure they are moved occasionally as mold can develop if they remain moist for too long.

- PPE (respirator, eyewear, gloves) must be worn. The cement mixer will throw out a lot of fine particulate matter and diatomaceous earth is not safe to handle without gloves and eye protection.
- Soil and compost must be sieved to remove rocks and bark from the mix.
- If parts of the mixture become too wet, they will stick to the sides of the drum and will need to be scraped off which can be both time intensive and exhausting. Adding more dry material to the mixture as needed will reduce the stickiness.
- Each batch can take about 45 minutes to an hour to properly complete.
- This method often creates oblong seedballs which don't easily roll down a hillside, ideal for our steep terrain.

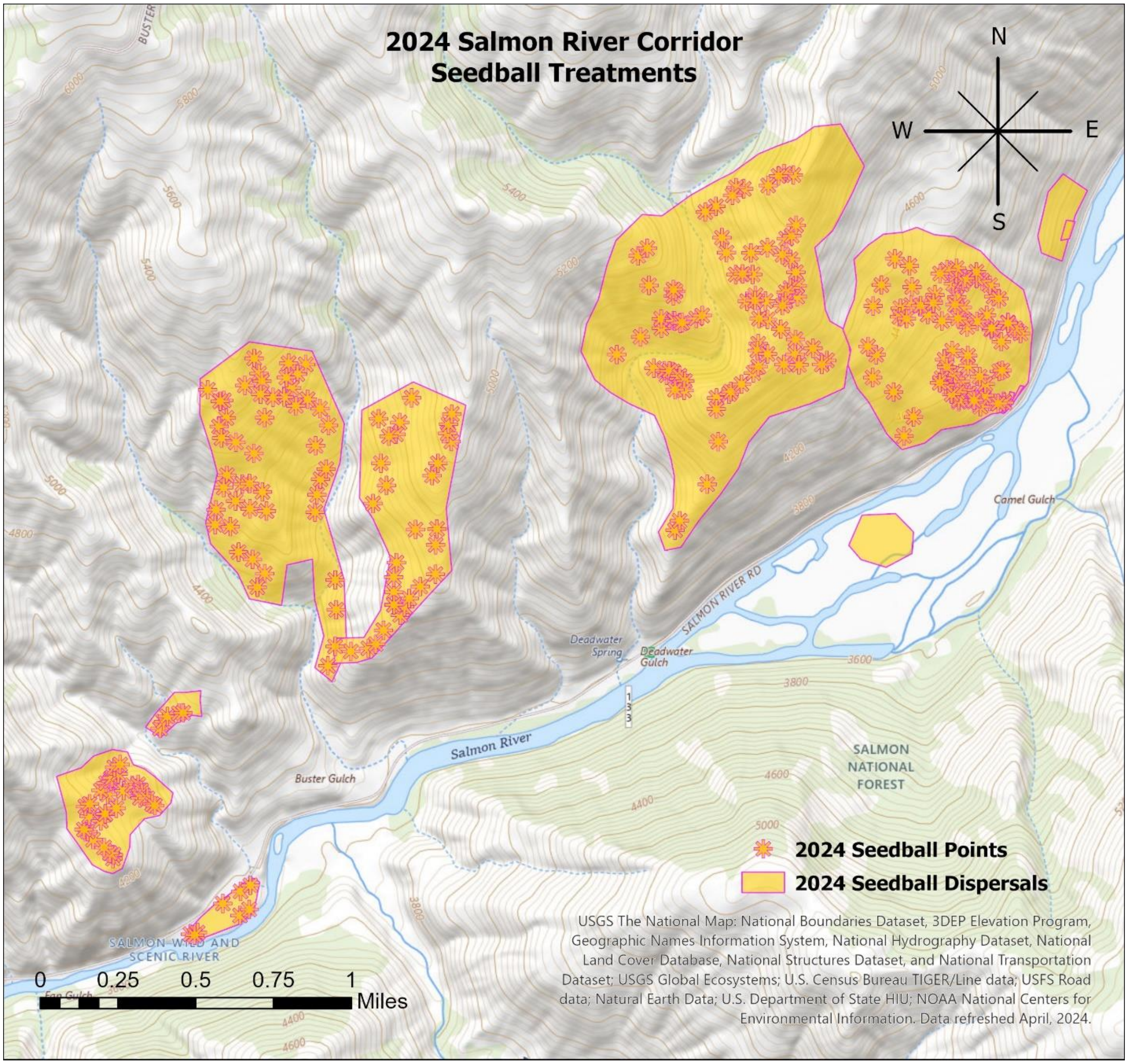
We made three batches of each seed mix using these instructions producing approximately 6,000 seedballs for each mix and approximately 60,000 seedballs in total.

Seedball Strategy

Though seedballs offer a viable alternative to other revegetation methods such as aerial seeding or plug planting, we still want to ensure we are distributing them wisely, placing them strategically in prime locations for seedling survival. We focused on a few different strategies in order provide our seedballs with the greatest chance of success.

- **Site Selection** – Despite having an idea of where we want to focus revegetation efforts, actually selecting the proper location for planting is impossible without visiting the site in person. We are aiming for swales in the hillsides that show where water is accumulating and flowing. We're looking for spots with deeper soil and locations where seedlings may be offered some shade. We're following the signs of existing vegetation to indicate where on the landscape plants are surviving and thriving.
- **Microtopography** – Studies have shown that a site's microtopography can have massive impacts on where plants are able to successfully establish. Irregularities in the soil like small divots, furrows, or channels, collect more water and litter which in turn provides shade from the sun, cooler temperatures, decreased wind abrasion, and increased nutrients and moisture. This effect is so strong that plantings just inches apart from each other may result in drastically different results. We utilized this science by digging small divots or ditches before placing our seedballs. This also has the added benefit of getting our seed deeper into the ground.
- **Nurse Plants –** Research clearly indicates that plantings made in the presence of existing vegetation have greater chances of survival. Seedlings benefit from their larger neighbors by utilizing their litter build up for nutrients, the shade they provide from the sun, and the added moisture retention that shade provides.
- **Restoration Islands** – Dense, focused clusters of plantings are a proven method in revegetation. Conducting restoration activities across an entire landscape is a huge and insurmountable challenge. However, focusing your efforts on ideal locations allows nature to do the work for you. If we can establish dense "islands" of biodiverse plant communities, they will hopefully survive and spread across the landscape on their own with little to no help needed.
- **Pollinator Habitat** – With one of our primary goals being geared towards supporting pollinators, establishing healthy plant communities means nothing if they aren't considered. Most of the plant species we seed absolutely must be pollinated by the proper insect in order to reproduce. However, many pollinator species like bumblebees have extremely limited flight distances, meaning if our plantings are too far apart or too far from existing pollinators, they'll never be visited by our buzzing friends. With this in mind, we avoided isolated plantings, ensuring we kept them well within the proper distance from each other, ideally leading pollinators from one planting to another.

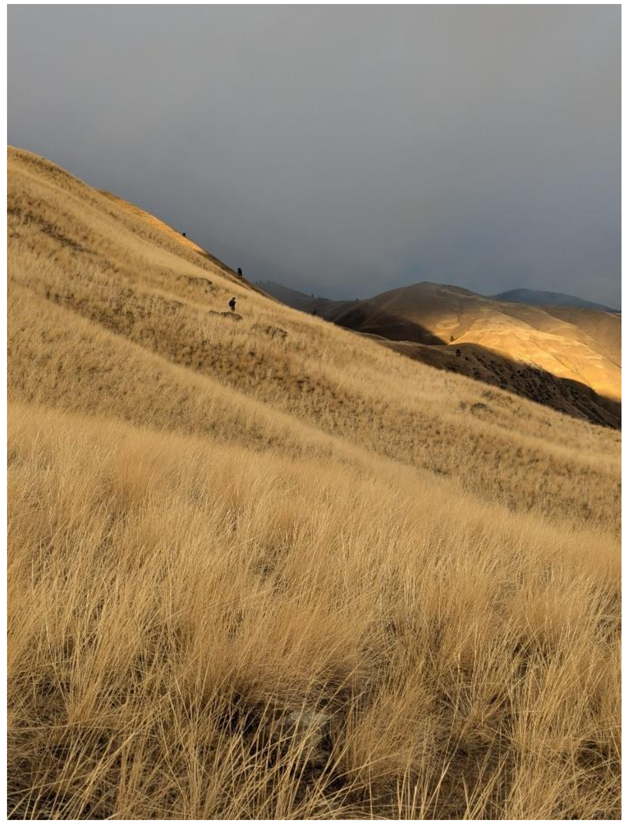
Seedball Treatments



Restoration islands were installed in ideal locations by placing seedballs in small divots or ditches. Each island includes approximately 20-60 plantings. Seedball points were placed where an island was installed. Additionally, we did significant distribution using slingshots which is why our polygons encompass and go beyond just the islands. We believe this strategy was worth testing as it allowed us to cover additional ground and distribute seedballs to areas unreachable on foot.

We seeded a total of 828 acres in 2024, more than 100 times what we accomplished in 2023. 105 of those acres were seeded with *Asclepias Speciosa* to improve Monarch habitat. We still have thousands of seedballs but unfortunately, early snowfall hindered us from accomplishing any more acreage this year. The possibility of spring plantings is being pursued.

Bins showing seedballs produced in our workshop containing different seed mixes.



Forest Service technicians dispersing seedballs on the hillsides of the Salmon River Corridor.

An installation of a small restoration island in a relatively bare patch of soil. Islands consist of 20-60 dense plantings in which small divots are dug with a mini-pick and seedballs are placed within.

Seed Storage

After seed is collected and processed, it must be adequately stored in order to retain its overall viability for future use. We learned from the National Native Seed Specialist, Kayla Herriman, that if seed is properly stored it can last for decades. She taught us that seed should be dried to below 30% equilibrium relative humidity (eRH), placed in sealed moisture proof containers, then stored in a dark, dry space between 35-50°F. With these guidelines in mind, we purchased a refrigerator for storage, a desiccating chamber for drying, and a hygrometer for testing seed moisture content.

Desiccator

Drying seed prior to storage is of the utmost importance because in most cases, each 10% reduction in eRH can extend a seed's shelf life by years. For short term storage of up to 18 months, below 30% moisture content is recommended, however for longer term storage, seed is ideally dried between 5-10%. In order to ensure we were drying our seed to the proper metrics; we decided against trusting the relatively arid environment of the Salmon region and instead looked for a more reliable method.

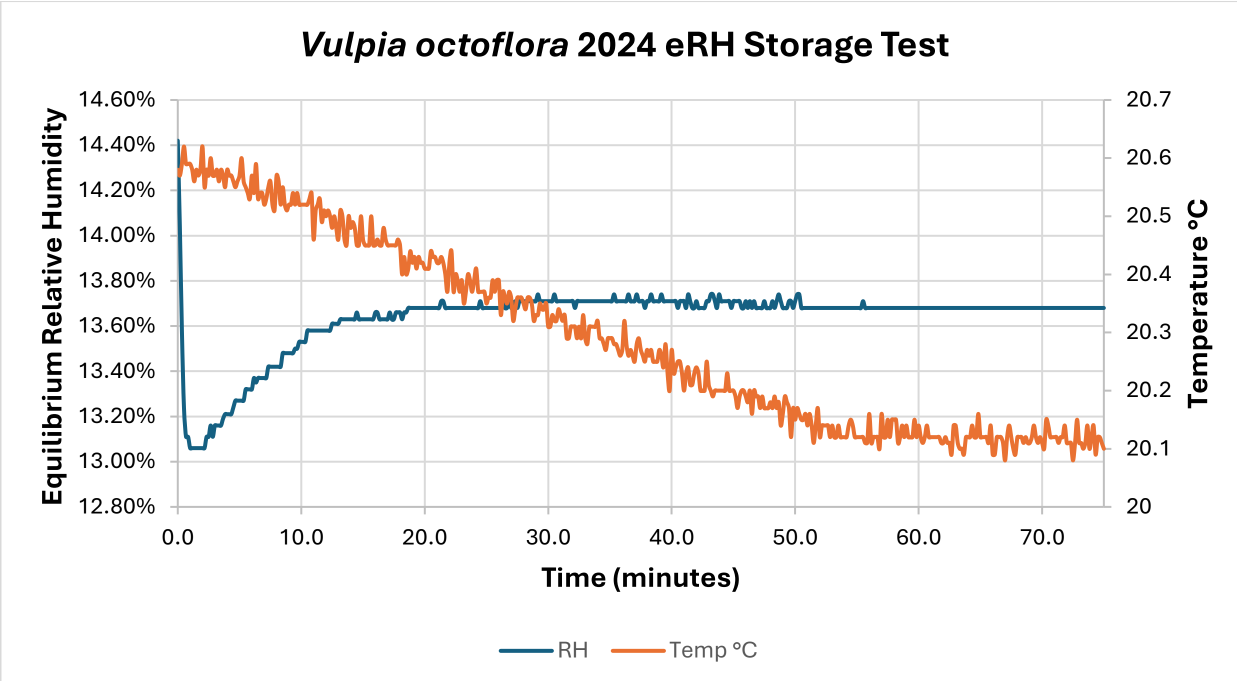
The solution we landed on was an electronic drying cabinet, designed for storing sensitive electronics in humid environments. It is essentially a sealed cabinet with multiple dehumidifiers that can dry the air inside to a set level. We selected a model capable of maintaining a relative humidity of 1-50%, giving us the range to meet any target moisture levels we may want. We set the device to 12.5% and all seed was stored inside in open containers following processing to give adequate time to dry.

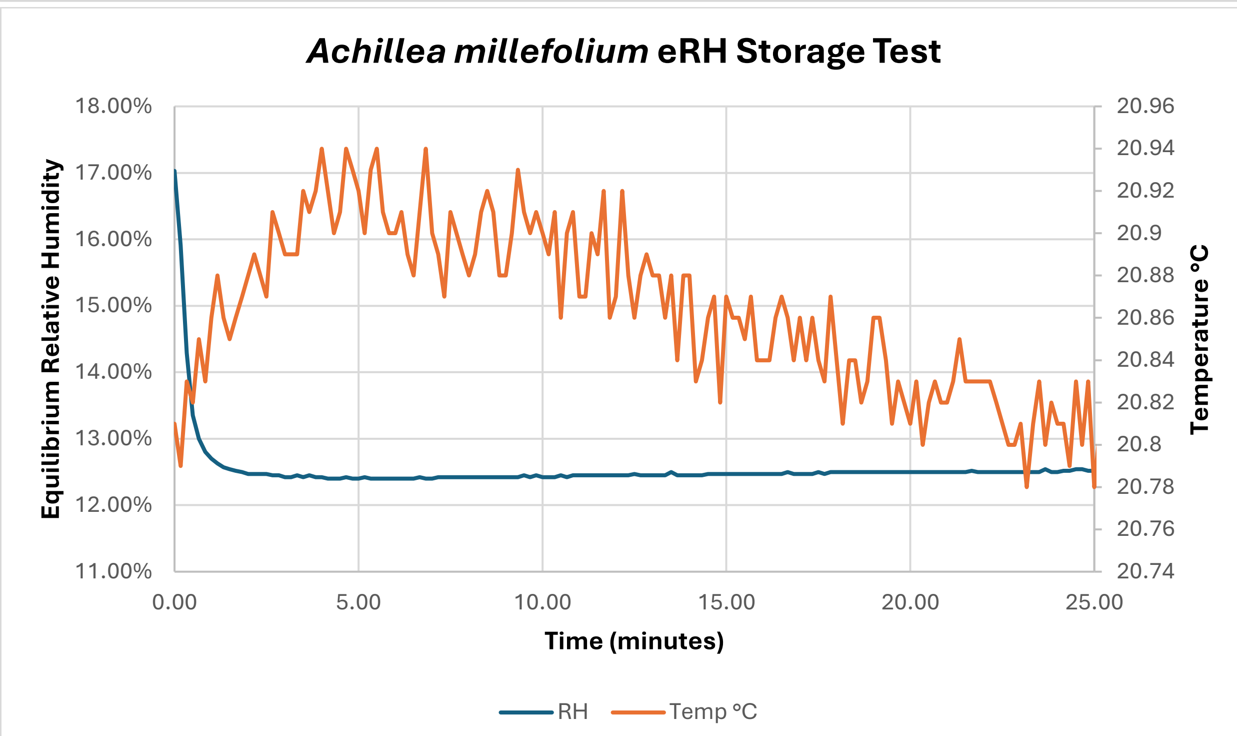
Hygrometer Testing

Hygrometers are capable of measuring moisture content in objects by calculating eRH which is equal to the relative humidity of the sample once it has stabilized. All living organisms and other objects which contain water are constantly attempting to reach equilibrium with the surrounding air. In this case, if your seed is moister than the surrounding air, it will give off moisture until the water content of the seed and the air is equal. The less air present in the chamber where the measurement is being taken reduces the time necessary for the readings to reach equilibrium. Our hygrometer works by measuring the relative humidity of a small amount of air trapped in a chamber with our seed sample and eRH is typically reached within 30-60 minutes.

We tested 25 different seed samples before preparing our seed for storage and all eRH values ranged between 12-19%. The following graphs display data from these tests showing how RH values stabilize over time. More data can be made available upon request.

eRH Storage Tests





Seed moisture content and eRH are analogs for one another and in many agricultural grains and seeds, research revealing this relationship's isotherms have been studied and quantified. Though, in regard to our niche native species, limited data is available. A rule of thumb is that an eRH of 10-20% equates to approximately 3-6% moisture content rising to 8% at around 30% eRH. Lower moisture content is ideal for species adapted to dry conditions.

Refrigerator

Once eRH testing confirmed our seed was adequately dried, we began preparing them for storage in our fridge. All seed was placed in sealed mason jars containing silica desiccant packets, labeled, then tightly wrapped with multiple layers of saran wrap to keep moisture out. Jars are then covered in foil to keep seed in a dark environment, then organized in the refrigerator's shelves according to collection year and source. The refrigerator is set to 35°F.

It's important to note that sealed containers should not be opened until they have been allowed to warm back up to the surrounding air temperature. Otherwise, the cold seed now exposed to the air can quickly build up excess moisture through condensation causing molding or rot.

Electronic drying cabinet repurposed as a seed desiccator.

Jars containing seed ready to be wrapped in tin foil for storage.

Volunteer Events

Volunteers assisted us at every turn in our process. Everything from seed collecting to seedball making and distributing. At the beginning of the field season, we created a Google form for locals to sign up for notices about our events and by the end of the year had over 60 people on the list. We held two volunteer seed collection days and a two-part event where volunteers helped hand roll milkweed seedballs then came out with us the following week to disperse them on the landscape.

With over 50 individuals having attended our events, volunteers helped us to collect seed from thousands of individual plants, helped roll over 2000 seedballs, and dispersed those seedballs across 69.8 acres. We are extremely proud of the community support we received this year and hope to continue engaging the public in what we do in the years to come.

Volunteers collecting seed from *Plantago patagonica* at Diamond Creek.

Volunteers hand rolling milkweed seedballs at the Sacajawea Center.

Volunteers using slingshots to shoot milkweed seedballs across the Salmon River near Newland Gulch.

Budget Spending

Overview

Our program primarily operates via a five-year challenge cost-share agreement with the Salmon-Challis National Forest under the designation: 22-CS-11041300-032. A new modification to that agreement signed on June 3rd, 2024, increased our total funding from $125,000 to $378,200 with money received from the Bipartisan Infrastructure Law specifically for our program. Approximately $25,000 of the BIL funding remained in a Forest Service owned pool for the purchasing of larger equipment and paying for fleet vehicle use. Additional funds in small amounts have been allocated to materials purchasing from other members of the Salmon-Challis Restoration Working Group, mainly the Bureau of Land Management and the Idaho Fish & Game. Lastly, SVS received the NEEF Biodiversity Grant in June of 2024, some of which is intended to pay for materials and staff time related to this program through June 2025.

SVS invoices quarterly and at this time has received reimbursement for all 2024 expenses as per our agreement. A total of $244,389.28 remains in the agreement and is intended to be used by the end of September 2027.

Materials & Equipment

The following tables show in what areas we spent funding and how each of our funding sources contributed overall. It is worth noting that Forest Service spending was much higher this year than other funding sources because we made multiple large, one-time purchases using the BIL money left with the forest for simplicity's sake. Anything purchased by SVS is technically owned by SVS, even if paid for by agreement funding, and we deemed these larger purchases were more appropriately acquired on the forest's side.

Total Cost per Category	**$24,264.28**
Seed Collection Materials	$2,087.38
Seed Storage Materials	$13,055.60
Seedball Materials	$1,821.12
Seed Cleaning Materials	$2,648.81
Office Supplies	$36.84
Lodging/Travel/Vehicle	$4,614.53

Total Contribution	**$24,264.28**	**100%**
SVS Agreement	$2,196.18	9.05%
FS Funding	$16,126.68	66.46%
BLM Funding	$2,460.52	10.14%
IDFG Funding	$3,211.17	13.23%
NEEF Grant	$269.73	1.11%

Match

SVS is committed to a minimum of 20% match of total funding in our cost-share agreements in the form of both non-cash and in-kind contributions. SVS has committed to matching a total of $103,161.50 by the end of our five-year agreement and as of the end of 2024, SVS has contributed $63,369.50. We are well on our way to meeting the remainder of our commitment, $39,792.00, by our agreement's end date in 2027.

Looking to the Future

SVS's Native Plants Restoration Program's goal on a year-to-year basis is to increase its output and its outreach, producing better results and engaging more of the community. The program has a quick turnaround time and an extremely busy field season, as the length of this report may indicate. But we have a lot of ideas for how to fill gaps in our workflow and increase the program's overall success:

- **Limited availability of native seed** – This is an ongoing problem across the United States. There simply is not enough appropriate seed being produced on a large enough scale to meet the plans and goals of those involved in landscape restoration. Though commercial growers are becoming more common, there is often a disconnect between what they grow and what is needed. One of our primary goals for 2025 is to apply some of our funding to support local growers in developing the infrastructure and technical expertise to produce seed for us on a larger scale, minimizing our dependency on wildland seed collections.
- **Seed testing** – Without proper germination or fill tests, we simply do not actually know the true viability of the seed we put in our seedballs. Whether we need to find the time to do it ourselves or work with a laboratory able to operate on our accelerated timeline, having our seed properly tested would help us better formulate our seed mixes and focus our collection efforts.
- **Biological Soil Crusts** – Cryptobiotic soil plays an important role in arid ecosystems improving soil stability, nutrient availability, and water retention. Soil crust restoration is becoming an interesting new sector of the conservation world, and our program is strongly interested in adopting some of these techniques and strategies in our future operations.
- **New technologies** – We are always looking for ways to give our seed an extra push towards successful establishment and we think there are ways we can further innovate on seedball technology.
  - **Activated carbon** – As mentioned previously in the report, this is an immensely helpful additive to our seedball recipe that absorbs residual herbicide in the soil and increases moisture retention for our seeds. Research into its restoration applications is still ongoing, but we know that further tweaking of its utilization in our seedballs is needed.
  - **Gibberellic acid** – The hormone which stimulates germination and growth in seeds, new research into seed coatings is revealing it is excellent at breaking dormancy in species that exhibit it, allowing for higher rates of germination and establishment. As many of the species we use exhibit high levels of dormancy, we are excited about the prospect of integrating this technology into the program.
- **New Staff** – We are currently on the hunt for a full-time coordinator who will help with the day-to-day management and coordination of our operations, allowing the program manager to focus more on growing the program as a whole.

Monitoring for Success

We have our work cut out for us in the coming spring. With 828 acres seeded, revisiting our seedball sites is of the utmost importance. We are excited to see what grows, learn from what we've accomplished, and pivot to find better success in the coming year. We can't wait to share with you what we find growing out of our seedballs in spring 2025!

Contacts

A huge thank you to everyone who assisted us this year: Dr. Elise Gornish, Kayla Herriman, Jordan Schaeffer, Leslie Hamilton, Teresa Hawley, Gina Knudson, Becca Aceto, Andy Klimek, Courtney Frost, Sara Windsor, Tova Spector, Kristi Mingus, Marisa Anderson, SCNF NZ BIP Staff: Diane Schuldt, Katie Baumann, Chris Stenlund, Arianne Pieszchala, Maggie Wertheimer, SVS Staff: Maggie Seaberg, Jenny Gonyer, Kate Yeater, Cameron Rolle, Garret Stahl, Meg Super, Laura Hollingshead, SVS Technicians/Interns: Max, Gus, Chayenne, David, Stephanie, Ezra, everyone involved in our Native Plant Communities Restoration Working Group, everyone who signed up for our mailing list, and the countless volunteers who came out and joined us this year. We absolutely could not have accomplished what we did without the support and cooperation of everyone involved.

Please feel free to reach out if you have any further questions and keep your ears to the ground for future events and information from us in 2025.

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