SPECIES FACT SHEET

<u>Common Name</u>: a caddisfly
<u>Scientific Name</u>: *Namamyia plutonis* (Banks 1905)
Phylum: Mandibulata
Class: Insecta
Order: Trichoptera
Family: Odontoceridae
Subfamily: Odontocerinae

Conservation Status:
Global Status (2005): G3
Rounded Global Status: G3- Vulnerable
National Status (United States): N3
State Statuses: California: SNR, Oregon: S3 (Vulnerable in the state due to a restricted range, relatively few populations, recent and widespread declines, or other factors making it vulnerable to extirpation).
(NatureServe 2008).

<u>Technical Description</u>:
Adult: A small, dull-colored moth-like insect. Adults in this family lack ocelli, and have antennae which are usually longer than the fore wings. The family is defined more precisely by wing venation (Wiggins 2004). Schmid (1968) provides a complete description for adults, including diagnostic characters for adult male and female, a photograph of the female, and figures of male and female wings, male genitalia (lateral, face view and aedeagus), and female genitalia (ventral) (Wisseman, *pers. comm.*). The species, originally described by Banks (1905), is the only species in the *Namamyia* genus.

Immature: The larvae of this species can be distinguished from other odontocerids by the heavily setate abdominal segment I (both dorsally and ventrally), and by the lack of ventral gills. The dorsum of the head is pebbled in texture and bears a ridge along each side. Larvae can reach up to 2 mm (0.08 in.) in length (Wiggins 1996). Larval cases in this family are made of large and small rock fragments held together by silken mortar joints, and are remarkably strong and resistant to crushing, The case of this species is curved and somewhat tapered, and up to 30 mm (1.2 in.) in length (Anderson 1976). It is coarser in texture than those of the closely related *Nerophilus* genus (Wiggins 1996). For additional descriptive information, see Wiggins (1996).

<u>Life History</u>:
Most trichopterans in temperate latitudes are univoltine (Wiggins 1996), developing from the egg through five larval instars, pupating and emerging as adults within a single year. Larvae in this family have a burrowing life-style mwhich is accommodated by the unique architecture of their case. The diet of

this species is unclear, but gut content analyses of other North American odontocerid genera reflect both predacious feeding (e.g. insect parts) and scavenging (e.g. detritus) (Wiggins 1996). Larvae of this species have been collected from May to September and the flight period of adults is from May to July (Anderson 1976).

Range, Distribution, and Abundance:
This species is restricted to the Coastal and Cascade Ranges of Oregon and California, occurring as far south as Kern Co., CA. In Oregon it is known from Benton, Curry, Jackson, Josephine, Lane, and Marion counties. Populations appear to be patchily distributed and exceedingly localized. Fewer than 30 total locations are currently known and it is not abundant at any location (Wisseman 2008, *pers. comm.*).

Forest Service/BLM Lands: Documented occurrences are from the Rogue River, Siskiyou, Siuslaw, and Willamette National Forests (Anderson 1976), including a recent (1999) occurrence in Siskiyou National Forest (Borgias and Wisseman 1999).

Habitat Associations:
This species is known from small, cool, densely forested streams in old-growth or mature forest watersheds (Wisseman pers. comm., Wiggins 1996). Odontocerid larvae generally burrow under gravel, sand, or silt (Wiggins 1996); this species has been found in core samples taken from areas of coarse gravel intermixed with silt and organic sediments (Anderson 1976).

Threats:
Most trichopteran species have highly specific preferences with regard to water temperature, velocity, dissolved-oxygen levels, and substrate characteristics, and are therefore sensitive to a wide array of habitat alterations. The loss of trees through timber harvest poses serious threats, since this species occupies mature forested habitats, and trees provide shade that maintains appropriate water levels and temperatures for larval and pupal development. Continued global climate change may further threaten this species. Projected changes due to this phenomenon include increased frequency and severity of seasonal droughts and flooding, reduced snowpack to feed river flow, increased siltation, and increased air and water temperatures (Field *et al.* 2007), all of which could unfavorably impact this species' habitat and long-term survival. Sedimentation, eutrophication, and chemical pollution caused by development and road construction could also impact this species.

Conservation Considerations:
Inventory: Re-evaluation of this species' status at the known sites (last surveyed between 1950 and 1999) is critical in identifying both its current distribution and its conservation needs. Abundance estimates for this species

at new and known sites would also assist future conservation efforts, since population size is important in evaluating the stability of a species at a given locality. Small, cool, densely forested streams in old-growth or mature forest watersheds are good candidates for new population sites (Anderson 1976, Wiggins 1996, Wisseman 2008, *pers. comm.*).

Management: Protect all new and known sites and their associated watersheds from practices that would adversely affect any aspect of this species' life cycle. Riparian habitat protection, including maintenance of water quality, substrate conditions, and canopy cover, would likely benefit and help maintain this species.

Version 2:
Prepared by: Sarah Foltz
Xerces Society for Invertebrate Conservation
Date: December 2008

Edited by: Celeste Mazzacano, Sarina Jepsen
Xerces Society for Invertebrate Conservation
Date: December 2008

Version 1:
Prepared by: Eric Scheuering
Date: January 2006

Edited by: Rob Huff
Date: June 2007

ATTACHMENTS:
(1) References
(2) List of pertinent or knowledgeable contacts
(3) Trichoptera Survey Protocol, including specifics for this species

ATTACHMENT 1: References:

Anderson, N.H. 1976. The distribution and biology of the Oregon Trichoptera. Oregon Agricultural Experiment Station Technical Bulletin, 134:1-152.

Banks, N. 1905. Descriptions of new Nearctic neuropteroid insects. Transactions of the American. Entomological Society 32: 1-20.

Borgias, D. and Wisseman R.W. 1999. Report on the 1998 and 1999 survey for *Rhyacophila colonus*, in forested torrents near O'Brien, Oregon. The Nature Conservancy of Oregon. Prepared for Diane Perez, Siskiyou National Forest.

Field, C.B., Mortsch, L.D., Brklacich, M., Forbes, D.L., Kovacs, P., Patz, J.A., Running, S.W. and M.J. Scott. 2007. Chapter 14: North America. *In:* Climate Change 2007: Impacts, Adaptation and Vulnerability. Contribution of Working Group II to the Fourth Assessment Report of the Intergovernmental Panel on Climate Change (Parry, M.L., Canziani, O.F., Palutikof, J.P., van der Linden, P.J. and Hanson, C.E., eds.). Cambridge University Press, Cambridge, UK. Available at: www.ipcc.ch/pdf/assessment-report/ar4/wg2/ar4-wg2-chapter14.pdf.

NatureServe. 2008. "*Namamyia plutonis*." *NatureServe Explorer: An online encyclopedia of life [web application]*. Feb. 2008. Version 7.0. NatureServe, Arlington, Virginia. 21 Oct. 2008. <http://www.natureserve.org/explorer/>

Schmid, F. 1968. Quelques Trichopteres nearctiques nouveaux ou peu connus. Naturaliste Canadien 95 (3): 673-98.

Wiggins, G.B. 1996. Larvae of the North American Caddisfly Genera. 2nd ed. University of Toronto Press, Toronto. 424pp.

Wiggins, G.B. 2004. Caddisflies: the underwater architects. University of Toronto Press, Toronto. 292pp.

Wisseman, R.W. 2008. Personal communication with Sarah Foltz.

ATTACHMENT 2: List of pertinent, knowledgeable contacts:

Robert Wisseman, Aquatic Biology Associates, Inc. Corvallis, OR.

ATTACHMENT 3: Trichoptera Survey Protocol, including specifics for this species:
Survey Protocol

Taxonomic group:
Trichoptera

Species:
Agapetus denningi
Farula constricta
Farula davisi
Namamyia plutonis
Rhyacophila colonus

Where:

Trichopterans utilize a diversity of fresh water aquatic habitats, including headwater springs, streams, rivers, lakes, marshes, seepage areas, ponds, hot springs, and temporary pools. Most species have highly specific preferences with regard to water temperature, velocity, dissolved-oxygen levels, and substrate characteristics. Since the case-making larvae generally specialize in certain types of building material, the size and composition of available organic and inorganic materials can largely limit species' distributions. Construction materials include sand, pebbles, small rocks, mollusk shells, algae, duck-weed, plant stems, pine-needles, bark, grasses, and dead leaves. Some species are more selective than others and a few even exhibit life-stage-specific specialization, changing the case material and design partway through their aquatic life. Additionally, trichopteran larvae are often highly specialized in their dietary preferences and in the manner and location in which food is obtained. For species-specific construction material, feeding behavior, and habitat information, see the section at the end of this protocol.

<u>When:</u>
Adults are surveyed in the spring, summer, and fall, within the window of the species' documented flight period. In temperate climates, adults of various species can be collected from ice-break until the first days of heavy frost (Canton and Ward 1980). Larvae and pupae are most conveniently surveyed at the same time as adults.

<u>Adults:</u>
Adult trichopterans are predominantly encountered in the vicinity of water, close to their emergence or oviposition site. Dispersal from the emergence site appears to be negatively correlated with vegetation density along the dispersal corridor; adults disperse farther (up to around 200 m (656 ft.) in sparsely vegetated areas (Collier and Smith 1998). In general, searches will be most productive within 30 m (98 ft.) of the water edge (Collier and Smith 2004). Adults are frequently collected from riparian vegetation with an aerial sweep net; they can also be hand-picked from the undersides of bridges and culverts, and from the sides and upper-surfaces of partly-submerged logs. Additionally, adults can often be collected in large numbers in soapy-water pan traps placed under a light (e.g. a vehicle headlight) and left overnight. Specimens can also be collected at night directly from lights or an illuminated sheet using an aspirator or finger dipped in alcohol. An aspirator is especially useful for capturing small species. Some species (such as members of the *Rhyacophila*) are attracted to ultraviolet light. Emergence traps placed over habitat where the larvae are known or suspected to occur are another good method for obtaining adults (Wisseman 2005, *pers. comm.*). For emergence trap designs and sampling information, see Davies (1984). Additionally, sticky traps constructed from 5-gallon buckets lined with non-drying glue are effective at capturing adults of some species (Applegarth 1995).

Adults should be killed and preserved in 80% alcohol, or killed in cyanide and transferred to alcohol. Cyanide-killed adults may also be pinned, particularly to preserve color patterns, but pinning often damages critical aspects of the thorax and dried specimens are very difficult to identify to species (Triplehorn and Johnson 2005).

Since trichopteran identification often involves close investigation of adult male genitalia, photographs and sight records will not provide sufficient evidence of species occurrences. However, such observations may be valuable in directing further study to an area.

Larvae and pupae:

The aquatic larvae and pupae are found underwater, often creeping slowly along the substrate, or attached to rocks. In streams and springs, it is best to search for larvae and pupae on the undersurface of large rocks and in the smaller substrate underneath the rocks. Since some species pupate in clusters, it may be necessary to turn over many rocks before finding a cluster. Grazing larvae frequently occur in mosses and liverworts growing on the tops of rocks, and in the thin layers of water running over rocks. In seepage areas at the head of springs, particular attention should be given to washing and searching samples of water-saturated organic muck (Wiggins 1996). In the heavily vegetated areas of lake shores, ponds, and marshes, larvae can be found in the substrate and crawling on aquatic plants. In deeper parts of lakes, larvae occur in surface mat plants, such as *Ceratophyllum*, and in soft bottom materials (Wiggins 1996).

When surveying for larvae, care must be used to avoid disrupting stream banks, shorelines, vegetation, and habitat. Depending on the habitat, a variety of nets can be useful. D-frame nets with mesh size fine enough to retain small larvae (0.5 mm, 0.02 in.) are the most versatile, as they can be used in both lotic and lentic habitats. In stream systems, the standard kick-net technique can be applied. The net is held vertically with the opening facing upstream and the flat side pressed tightly against the bottom substrate, so that water flows neither under nor over the net. Large rocks and wood immediately upstream of the net are gently scrubbed by hand or with a soft brush and the bottom substrate is disturbed with the hands, feet, or a stick while the current carries the uncovered and dislodged insects and material into the net. The stream bottom is disturbed to a depth of 4 – 6 cm (1.2 – 2 in.) for about three minutes, following which the net is removed from the water for specimen retrieval. When lifting the net, the bottom of the frame is swept forward in a scooping motion to prevent insects from escaping. Net contents are then flipped or rinsed into shallow white trays to search for larvae more easily, as they are often quite cryptic and can be difficult to see if they are not moving. In addition to nets and shallow trays, the following equipment is also useful: fine-mesh strainers/sieves for washing mud and silt from samples, squirt bottles for rinsing the net, five-gallon buckets for holding rinsing water, and white ice-cube trays, forceps, and a hand lens for sorting insects.

Larvae and pupae should be preserved on-site in 80% alcohol, unless collection for rearing is an objective. Since most trichopteran species have not been described in their larval stage, rearing can be critical in both (1) enabling species identification and (2) providing novel associations of larvae with adults. Wiggins (1996, pages 37-38) provides a summary of the accepted methods for immature-adult associations in caddisflies. Generally, in order to maximize the amount of information that can be gained from collected specimens, as many life stages as possible should be collected and a portion of both the larval and pupal series reared to adulthood. While pupae can be reared in small, refrigerated containers containing damp moss, larvae require an aerated aquarium with isolated cages for individuals. An oxygen bubbler generally provides sufficient oxygen and current, although some species (e.g. members of the Hydropsychidae) may require unidirectional current. Detailed techniques for rearing stream-dwelling organisms in the laboratory, including transportation, aeration, current production, temperature control, food, and toxic substances, are provided by Craig (1966), and available online at http://www.nzetc.org/tm/scholarly/tei-Bio14Tuat02-t1-body-d1.html (last accessed 19 November 2008).

Although quantitative collecting of trichopterans is difficult, population-size data is important in evaluating a species' stability at a given locality and in assessing its conservation needs. Relative abundances of immature trichopterans can be estimated by using a uniform collecting effort over a given sample period at comparable habitats (Wiggins 1996). The area or volume of substrate samples can also be standardized, although the aggregated spatial distributions of many species (e.g. Schmera 2004) can complicate this approach.

While researchers are visiting sites and collecting specimens, detailed habitat data should also be acquired, including substrate type(s), water temperature, water source, water velocity, water depth, stream width, canopy cover, streamside vegetation density, and degree of human impact. Algal or cyanobacterial blooms and other signs of eutrophication should be watched for and noted.

Species-specific Survey Details:
Namamyia plutonis

The sole member of the genus *Namamyia,* this species is restricted to the Coastal and Cascade Ranges of Oregon and California. Populations appear to be patchily distributed and exceedingly localized. Fewer than 30 total locations are currently known and it is not abundant at any location. Because populations of this species may be negatively impacted by timber harvest, road construction, and other activities that degrade stream habitat, re-evaluation of this species' status at the known sites (last surveyed between 1950 and 1999) is critical in identifying both its current distribution and its conservation needs. Abundance estimates for this species at new and known sites would also assist future conservation efforts, since population size is important in evaluating the stability of a species at a given locality. Small, cool, densely forested streams in old-growth or mature forest watersheds are good candidates for new population sites (Anderson 1976, Wiggins 1996, Wisseman 2008, *pers. comm.*). The larvae of this species inhabit sand-gravel substrates in streams and channels as narrow as 30 cm (11.8 in.) across, and at elevations of approximately 1200 to 1500 m (4000 to 5000 ft.) (Wisseman 2008, *pers. comm.*). Aquatic vegetation may also be an important habitat characteristic, since the gut contents of three examined *Farula* larvae comprised mainly vascular plant pieces with some fine particulate organic matter (Wiggins 1996).

Surveys should be conducted from May to July in order to optimize encounters with both adults and larvae (adults have been collected from May to July; larvae have been collected from May to September). Hand-picking or sweep-netting riparian areas seem to be the most effective methods for sampling adults, as only one adult has been collected at an ultraviolet light (Wisseman 2008, *pers. comm.*). Since the larvae of this family generally burrow under gravel, sand, or silt, they are typically not encountered unless the substrate is searched (Wiggins 1996). Core sampling may be an effective way to sample the aquatic stages; larvae have been found in core samples taken from areas of coarse gravel intermixed with silt and organic sediments (Anderson 1976). Since this species has low abundances and patchy distribution patterns, multiple areas of suitable habitat at each site should be carefully searched for individuals.

Schmid (1968) provides a description of both male and female adults. The larval case is described as being made of small rock fragments, curved and somewhat tapered, and up to 30 mm (1.2 in.) long. Because the larval stage of this species has not yet been definitively associated with the adult (Wiggins 1996), successful rearing of larvae to adults would be especially informative for our understanding of this species.

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References (survey protocol only):

Anderson, N.H. 1976. Distribution and Ecology of Oregon Trichoptera. Technical Bulletin #134. Agricultural Experiment Station, Oregon State University, Corvallis, Oregon, 152 pp.

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