THOMAS BURKE MEMORIAL WASHINGTON STATE MUSEUM

SPIDER COLLECTION:
CURATION, CONSERVATION, LABELLING, AND RECORDING
General Description of Procedures and Rationale

by Rod Crawford, January 1992
(revised Oct. 2008)

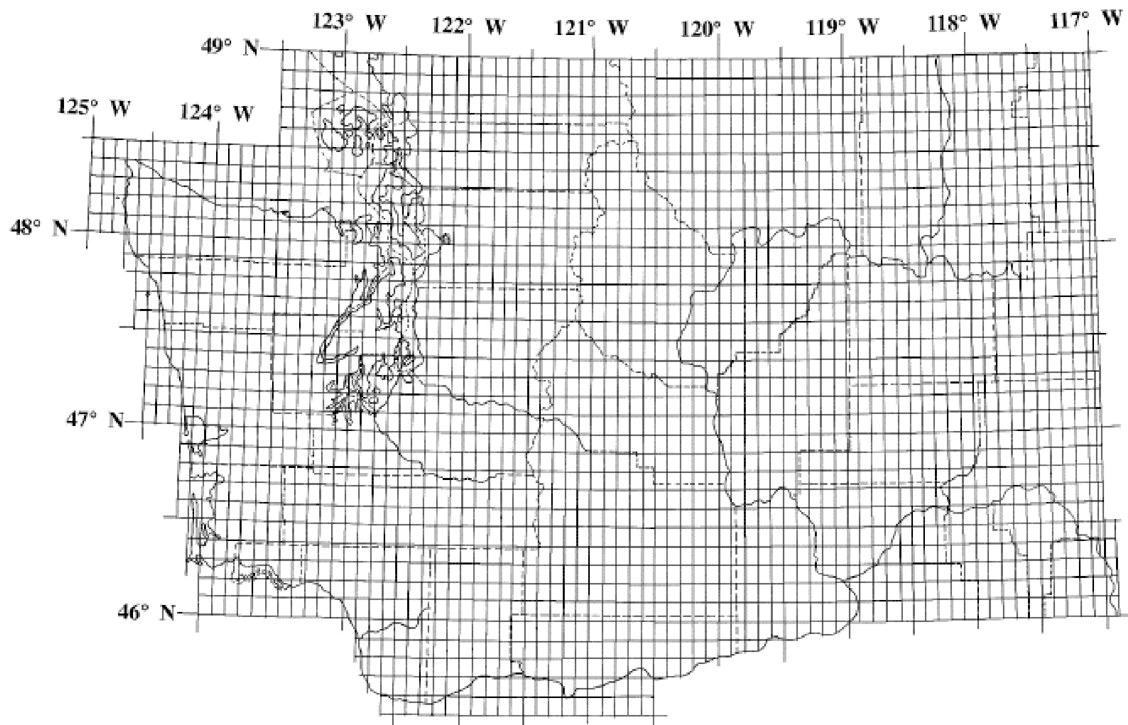
As of this writing, the Burke Museum spider collection has about 125,000 spider specimens of which 85,000 are fully curated. Ours is believed to be the largest spider collection in the Pacific Northwest and second largest on the West Coast; the next larger, that of the California Academy of Sciences, was reported in 1992 to number 90,000 specimens; it is being actively expanded and must still be larger than ours, but I couldn't easily find a more recent figure.

ACQUIRING SPECIMENS

Most spider specimens we acquire are collected by me, my student assistants and volunteers, or university colleagues as part of their own research projects. A minor but significant number are brought in for identification by the general public. The latter are accepted or discarded on an ad hoc basis, depending on potential research value, accuracy of data, or whether they represent new information on geographic, temporal, or ecological distribution. Of specimens collected by myself or staff, most that are identifiable are kept; there is seldom any need to "thin" series since 50 or more small spiders can be stored in the same size vial as one. Condition of specimens is not normally a factor; if they are identifiable this usually means the adult genitalia are intact, and condition of other body parts is less important to research value. Excessive "duplicate" series of common species from the same or nearly the same locality, may be discarded (if juvenile or in poor condition) or set aside for exchange with other institutions, including the Smithsonian, the Denver Museum of Nature and Science, the California Academy of Sciences, and the Field Museum of Natural History. Only twice in 50 years have we acquired a major private spider collection: that of J.R. Thomson acquired in 1974, and the considerably smaller one of Walla Walla College in 1992. Most of our specimens from outside Washington are either from the Thomson collection, or from expeditions to far eastern Russia undertaken in 1994-2003. The Russian material has proven very useful in elucidating trans-Beringian faunal connections and identities of obscure taxa first described on one side of the North Pacific Ocean and later found on the other.

While I do not rule out acquisition of additional "exotic" material, my first priority is to cover the state of Washington as thoroughly as possible; my second priority is Alaskan material (currently about 4,000 curated specimens). To facilitate even geographic coverage within Washington (desirable as a basis for meaningful distribution mapping) the state has been divided by a grid of 0.1° of latitude and longitude (Fig. 1). See Crawford (1983) for discussion of why I use this grid instead of others available.

Figure 1.



Each 0.1° area is called a gridspace; the state contains 2195 of these with some land area. A principal goal has been to collect over 20 specimens from each of 10% of these gridspaces. That has been achieved (389, 17.7%, sampled to date). Other goals are to have at least one such collection from each 0.5° area (there are 99) and one 40+ species collection from each county (there are 39). We are still working on these goals, and to date have sampled 91 0.5° areas and 29 counties. When all these goals are achieved we should have a good picture of the distribution of most species known from multiple localities. In the 1990s, I started a supplementary program to collect from as many rare habitat types as possible. These are being identified using information from numerous sources, such as the state natural heritage program, national natural landmarks program, state natural resources publications, ecological literature, conservation organizations, and personal communications.

Actual collecting techniques used in the field vary depending on the habitat type, but are aimed at sampling as many microhabitats as are present at a specific site. Specimens are swept or beaten from vegetation, some of the more productive types including grassy or herbaceous fields and meadows; wetland flora; field and forest-edge shrubs; forest understory shrubs; coniferous tree foliage; sagebrush, rabbitbrush, and greasewood. Moss and leaf litter are sifted onto a neutral colored cloth using a box with a bottom of 1/2" mesh screen, spiders being picked up with a wet brush; some faunal differences are noted with each different type of leaf litter, and sphagnum (no matter how wet) from bogs usually produces unusual species; moss and litter must be at least damp to yield many spiders. Material too fine to sift may be brought to the lab and processed in a Berlese funnel. Rocks, bark, wood, and boards on the ground, and loose bark on logs, stumps, and standing dead trees are searched under, and large pine cones tapped over a

net, all of those materials being returned to their original positions as far as possible. Horizontal and vertical surfaces (especially near water) are visually searched for active spiders and webs; the small webs of dictynids and some araneids and theridiids are searched for on the leafless tips of dead portions of field plants. Pitfall trapping for spiders uses small plastic cups containing fluid; large can traps favored by vertebrate workers are relatively useless for spider collecting. The fluid employed is propylene glycol for periods of a week or more; water for periods of up to 2 days. Short-term pitfalling works best at warmer times of the year at sites without closed canopy tree cover; in ideal conditions a 5-hour, 5-trap sample may yield 50 spiders, but other times one gets nothing. [Note: formerly we used automotive antifreeze for pitfall preservative, but the propylene glycol antifreeze made for RV water systems has proven far superior]. Buildings (from barns and outhouses to dwelling houses), trash dumps, and other manmade habitats are not neglected, since synanthropic species must also be documented, and many native species take advantage of such ideal habitats as the folds of discarded carpeting and the channels of corrugated cardboard.

When collected, spiders are preserved in 70% isopropanol, which is a better killing agent and fixative than ethanol. They may be placed directly into preservative or immobilized first with a standard entomological cyanide tube. An exception is specimens for which some potential DNA study is in prospect. These are kept alive until they can be preserved in 95% ethanol, then a leg (with larger spiders) or an entire spider from a series if very small, is kept permanently in 95% ethanol in a freezer while the rest of the spider or series goes through the standard isopropanol to 70% ethanol sequence. Live immatures that look interesting enough to be worth some time investment are brought back to the lab to rear to adulthood.

All specimens with the same data can be put in the same container if there is room (up to half the liquid volume). Before leaving a site, each container is given a pencil label giving locality, date, and microhabitat plus any field observations. N.B. the notation "hand collected" is never employed, as it tell nothing about the specimen. Instead, "active on ground" or "under rock" or "web on bare branch, " etc. are preferred. Putting code or catalogue numbers in vials, and data in a separate record, is usually tried by beginning collectors but invariably results in the data becoming separated from the specimens, the latter being rendered worthless.

Spiders in from the field or other sources are stored in our standard field vials, 7-ml all-plastic HDPE scintillation vials (Fisher Scientific 03-337-1); larger samples, in 4-dram patent lip vials with neoprene stoppers, until they can be processed. They are kept in isopropanol a minimum of one week to ensure proper fixation (spiders not properly fixed may be easily punctured and the colors fade more); up to several months in isopropanol does no harm. Permanent preservation in isopropanol (as recommended by Levi 1966) makes specimens excessively stiff and brittle, but this can be reversed by transfer to ethanol.

IDENTIFICATION

Spiders are rough sorted (to remove unidentifiable juveniles and debris) in small dishes of ethanol (fumes less irritating than isopropanol), and definitively identified in dishes containing sand; washed white sand for large spiders and chromatography beads

for tiny ones, the whole being submerged in ethanol, under the strongest illumination obtainable. The purpose of the sand is to orient the specimens and/or their dissected genitalia in precise positions, since even slightly different angles can make them look and measure "different." Genitalia are compared with drawings made from other specimens using a camera lucida, so that the images of the specimen and drawing can be precisely superimposed; this removes the subjective factor in comparing specimens with drawings (Crawford 1988c). All specimens of one species from the same precise locality and date are put in the same vial, with a tally of the number of males, females, and juveniles from different microhabitats if collected from more than one. Dissected parts are put in homemade microvials (cut sections of microcapillary tubing stoppered with cotton). Commercial microvials are too heavy, and there is a risk of their crushing specimens they fall against.

LABELLING

1. Materials. Formerly, it was necessary to produce spider labels with an old-fashioned stencil mimeograph or a now almost as old-fashioned daisy-wheel printer with ink ribbon. This was because laser-type printers are wholly unsuitable for labeling of alcoholic specimens. Fortunately, the discovery (by the Field Museum invertebrate department) of the Lexmark Z52 inkjet printer, which uses permanent ink, has now made the production of spider labels easy. Where possible, paper of at least 50% rag content is used, although wood pulp paper seems to be permanent in alcohol if the fluid is changed at least once to remove leached acid. Card stocks and thick "biology paper" tend to disintegrate eventually in fluid. Many other methods have been tried and found wanting. Entirely handwritten labels cannot be read quickly enough and interpretation becomes even more difficult with time and distance (Levi 1966). **Xerographic printing, including computer laser printing, is wholly unsuitable for liquid storage labels!** Such labels seem excellent initially, but the print is merely heat-glued onto the surface of the paper, and in about 5-10 years begins to fall off. Ultimately, all that is left of the data is black dust at the bottom of the vial. Photo-offset printing is usable, but offset inks seem to contain some of the purple component of typewriter inks. Inks from film (as opposed to cloth) printer ribbons may be 100% alcohol soluble and are to be avoided, as are temporary labels written in ball-point or felt-tip. Old-fashioned mimeograph ink (NOT "ditto" or duplicator) was wholly inert in alcohol, but this ink and the machinery to use it are now almost unobtainable; thus, the Lexmark permanent black ink cartridges are a tremendous boon to arachnology. For handwritten portions of labels, only India ink (including the non-clogging type for technical pens) and the ink in the new Sakura "Micron Pigma" disposable pens, is permanent in alcohol, and only if it is first thoroughly dried under a lamp.

2. Size, number, and placement. Labels must be placed in vials with the alcohol and specimens; otherwise, they are subject to falling off, abrasion, fading, and insect damage. To make the labels easy to read without removing them, I make their width exactly the inside length of the wide part of the vial (examples below are actual size), so they can be wrapped around the inside of the vial in contact with the glass. Only one label is used per vial. This is a matter of taste, but I find that with the common practice of putting identification on a separate label, the two labels end up facing each other far too

often. If identifications change, the new i.d. label is put **between** the glass and the i.d. portion of the original label. I never put extra labels into the main body of the vial, as this can result in squashing specimens between labels. We also take care than no specimens are between the original label and the glass. All labels are oriented the same way, left edge of the print at bottom of vial.

3. Format. The example below shows our spider label format.

Coreorgonal monoceros (S.)	Cicurina pusilla (Simon)	Zygottus corvallis Chamb.
det. R.Crawford 2006	det. R.Crawford 2006	det. R.Crawford 2006
1♂	1♀	3♀48♂
Rockport State Park 540':	Rockport State Park 540':	Rockport State Park 500-700'
69 pitfalls, old growth forest	77 pitfalls, old growth forest	77 pitfalls, old growth forest
48.490°N 121.611°W	48.490°N 121.613°W	48.489-493°N 121.609-617°W
WASH.: Skagit Co.	WASH.: Skagit Co.	WASH.: Skagit Co.
10 I-13 III 1992 James Bergdahl	13 III-7 V 1992 James Bergdahl	13 III-7 V 1992 James Bergdahl

The first three lines need little explanation. Line 4 gives a locality name as a convenient tag for speech and memory, and an elevation in all cases (regardless of terrain) so that all data will be comparable. Line 5 gives microhabitat data, handwritten if there is too much to fit, and itemized with specimen numbers if multiple. Line 6 is the most important; it gives the precise location in decimal latitude/longitude to 0.001° if possible; this makes the site relocatable with roughly 100-m accuracy, unaffected by name changes, road changes, and the great, sometimes unexpected, inaccuracy of most other types of locality designation, as well as readily mappable on a gridded map such as in Fig. 1. With practice, grid locality data can be determined quickly and easily; see Crawford (1983, 1988b) for full information and the drawbacks of other methods. It is essential to include the degree sign ° which can be a raised lower-case "o" because otherwise the numbers can be mistaken for degrees/minutes/seconds. It would seem that no excuse is needed for improving the accuracy of scientific data, but it is amazing how many collectors will defend to the death their right to put "Columbus, Ohio" or such as the entire locality description! Line 7 gives the state and county, line 8 the date and collector. It is extremely important to give dates in this or an equivalent form. **Never use Arabic numerals for both day and month,** as "4-8-89" will be April 8 to one person and August 4 to another, and many collections (including ours) go back to **18**89! N.B. With multi-day trapped specimens, **it is essential to put both the starting and end day of trapping on labels!** For reasons that escape me, most pitfall trappers tend to put only the trap pick-up date on their labels. This amounts to falsification of data, as many of the specimens were not trapped on that exact day. The collector's name is not used for reasons of ego, but to provide a way for later users to evaluate accuracy of the data.

When I started using decimal degrees, the only available coordinate system for North America was according to the N.A. 1927 geodetic datum (a datum, in this sense, is a coordinate system based on a locatable reference point and standardized set of dimensions for our somewhat irregularly spheroid-shaped planet that provide a "best fit" for a given area). Today most GPS receivers and online map applications come pre-set with the WGS 1984 or N.A. 1983 datum, which gives slightly but significantly different coordinates. My North American labels will, for consistency, continue to use the earlier datum for the foreseeable future. For the same reason my Russian collection uses coordinates of the Pulkovo 1942 datum.

4. Treatment of older locality data. All vials are given an "outer" label of the type described above. Where there is an older label in a previous format, that is also kept,

inside the vial if it is small, elsewhere if it is so large as to risk crushing specimens (old I.D. labels are not kept unless they have special research value). Where possible, the new label includes a grid interpretation of the locality data on the old label (which, since it is kept, remains available for back-checking). In most cases, old locality data can only be interpreted to the nearest 0.1° (if that), but this is still a useful exercise because it allows consistency in specimen storage and data retrieval.

STORAGE

1. Materials. Our permanent preservative is 70% ethanol, 30% distilled water. Use of distilled water seems to help prevent yellowing of the alcohol. Isopropanol is not the best for permanent preservation, but denatured ethanol can be used if your institution cannot buy tax-free pure ethanol. Never preserve spiders in formalin or methanol, as they become unworkably brittle and may disintegrate. Many authorities recommend addition of glycerin to help prevent desiccation if the alcohol dries. I find that specimens to which this happens may be in worse condition than those completely dried, as when the glycerin concentrates it clears tissues and sometimes promotes fungal growth; also, it "greases" the stoppers which may then pop out in warm weather! On the other hand, mites can be preserved successfully in pure glycerin, although acarology texts do not mention this as an option. Our standard vial for permanent storage is the 2-dram patent lip vial, varied with occasional 3 or 4 dram vials for very large specimens. The "standard museum method" of cotton-stoppered vials inverted in a larger jar of alcohol is safer for long-term protection from drying, but we have found it impracticable for a collection where vials are added and retrieved on a daily basis. We therefore keep vials as separate units stoppered with neoprene. The stopper material is very important. Screw caps eventually loosen (the new type containing polyethylene cones are better, but expensive); natural corks may leak tannic acid into alcohol which softens spider abdomens to the consistency of cheese; natural (black) rubber and some neoprene contains too much sulfur, which dissolves and recrystallizes on specimens, so when ordering neoprene, specify a low-sulfur formulation. Most neoprene stoppers are green or rose colored; similar-appearing buff or amber stoppers are made of natural gum rubber which eventually hardens, cracks, and disintegrates.

2. Sources. Size 00 solid stoppers fit 2-dram and 3-dram vials; size 0 fits 4-dram vials. Our suppliers:

Acme Vial and Glass Co., Inc.
1601 Commerce Way
Paso Robles, CA 93446
(805) 239-2666
http://www.acmevial.com/
2-dram patent lip vials #S-900

Plasticoid Co., Inc.
249 W. High St.
Elkton, MD 21921
(301) 398-2800
http://www.plasticoid.com/
00 solid neoprene lab. stoppers #M350

The stoppers were formerly made by Rhoades Rubber Co. and The West Co.; both are now out of the stopper business. Both vials and stoppers can also be purchased from BioQuip Products, 17803 LaSalle Ave., Gardena CA 90248, (213) 324-0620, but it is 30-40% cheaper to buy them in bulk from the manufacturers. BioQuip is a good source

of collecting equipment and other supplies, and their "heavy duty sweep net bag" is excellent and practically indestructible (not so their "heavy duty aerial net bag!").

3. Vial filling and leakage. As implied above, the use of individual stoppered vials is not without hazard, and rarely individual vials dry, either because the vial spontaneously cracks (this seems to happen mainly or entirely with vials from other manufacturers than Acme; VWR vials are especially prone to a circular crack around the bottom) or because the stopper was previously used in a vial with a narrower neck and is therefore loose. Barring such problems, these vials lose about 15% of their alcohol in 20 years in Seattle; the problem would be worse in warmer climates. Frequent monitoring helps (with all types of alcohol storage!) and dried spiders can be restored to second-rate but usable quality by a few minutes in boiling distilled water. We fill our vials as full as practicable, with as little air as possible. To do this, it is necessary to insert a flexible wire with the stopper so that the air can escape, then remove the wire, being careful to leave no broken bits of wire. Complete filling is important for 3 reasons: a) air compressed into the vial by the stopper will tend to force the stopper back out; b) if the vial is not full, specimens can be stranded above the alcohol; c) if the vial was initially full, it is easier to tell if it is leaking. Type specimens are protected in individual very small stoppered vials (from BioQuip) within regular vials.

4. Storage facilities. The spider collection is stored in an increasing number of free-standing metal cabinets, 6.5' high, 3' wide, and 18" deep, each with 11 shelf levels. Individual storage trays were custom made to fit these cabinets, which were adapted from previous use in vertebrate collections. Each tray (presently of wood) is as long as the depth of a shelf and as wide as 1, 2 or 3 vials plus wood thickness, and contains 1-3 longitudinal channels into which 2 and 3 dram vials can fit (snug for the 3-dram) separated by a longitudinal median partition. The "egg carton" type of tray with cross-hatched cardboard partitions could also be used, but would make it much more trouble to insert a vial into the middle of a series of vials. The existing trays employ spacers for the ends of vial rows, cut from polyurethane foam sheets.

5. Collection arrangement. Currently specimens from different states and regions (Washington, Alaska, Oregon, Russia and other areas) are kept separately to facilitate a more detailed taxonomic-geographic arrangement within the Washington collection. The latter is kept in taxonomic arrangement of the "Annotated Checklist of the Spiders of Washington" (Crawford 1988a), which is also available online:

http://www.tardigrade.org/natives/crawford/index.html

Within each species, vials are stored in the numerical order of the grid locality designations, which are written in a 4-digit abbreviated format on the stopper (described by Crawford 1988a, p.3). Thus, a particular vial of a common species can be retrieved without pulling out and reading the labels of all the vials of that species.

RECORDING

The paper catalogue of the spider collections is kept on 4x6" cards adjacent to the first cabinet of the collection. Spider data are kept by locality, not species, because the collection is organized in such a way that species-by-species data can be easily retrieved from the original labels. Having a file by locality adds the capability of easily checking data on a geographic basis, frequently needed in my research. All spider data for a

locality, as well as data about the locality itself, are recorded on pre-printed cards such as the one reproduced below.

1199

ABOVE MAYFIELD DAM 520 ' (158 m) LEWIS Co.

46.506 °N 122.586 °W Roadside in seral Douglas fir-hemlock-maple-alder forest

Metellina mimetoides (C+I.) 1♀	ex forest under story	12 III 1988	R Crawford
M. curtisi McC. 1♀5juv	" "	" "	"
Tetragnatha laboriosa H. 1juv	" "	" "	"
" " 1juv	beaten from conifers	" "	"
T. versicolor W. 1juv	" "	" "	"
Cyclosa conica (Pallas) 1♂penult	ex forest understory	" "	"
Theridion tinctum W. 2juv	beaten from conifers	" "	"
Wubana pacifica (Bks) 1♂	ex forest understory	" "	"
Microlinyphia dana (C+I) 7♀3♂2j	" "	" "	"
" " 3juv	beaten from conifers	" "	"
M. mandibulata (Em) 1juv	ex forest understory	" "	"
Neriene digna (Keys) 2juv	beaten from conifers	" "	"
Pityohyphantes rubrofasciatus K.	4small j. " "	" "	"
" " 3juv	(♂penult) ex forest understory	" "	"
Philodromus rufus W. 1juv	" "	" "	"
" " 4juv	beaten from conifers	" "	"

Each different locality is given a unique serial number (upper left corner) and the grid coordinates are also unique; either could furnish the basis for formal "catalogue number" should such ever seem desirable. It is not my intention ever to assign numbers to individual specimens, since numerous specimens may be kept in a single vial and there is no way to tag them.

All the data in the card file are also kept in a computer database. The spider collection was the first zoology collection at this museum to be databased. Therefore, unfortunately, it was done on a computer platform that is no longer supported. Transfer to a newer, custom-designed platform is in progress as of this writing.

The card file is currently fully up to date with all curated Washington specimens. There is a backlog for database entry, but it is actively being reduced. Most Russian spiders are catalogued in a wholly different system based on computers at the UW College of Fisheries which sponsored my Russian trips, but these too will be transferred to the new platform. Cataloguing of specimens from Alaska is also in progress; those from other states, as well as harvestmen and other arachnids, are a project for the future.

LOAN PROCEDURE

Specimens from the Burke Museum spider collection are available on short-term loan for scholarly research. It is our policy to loan a reasonable number of specimens on request to researchers with institutional affiliation, unless the individual has a reputation for not returning loans. Loans to unaffiliated individuals are only made in exceptional

circumstances, since in case of need an institution may be able to return specimens not returned by the individual. Loans are not made to artists or photographers. If a loan request specifies a group for which we have a large number of specimens (e.g., 100 vials), the loan may be made in installments. Detailed regulations for spider loans will be found on the back of the attached sample loan form (next page).

Vials to be mailed are individually wrapped in plastic foam sheeting and taped, then packed in a smaller box(es) which in turn is placed, surrounded by packing material, in a larger box. I send all loans to the U.S. and Canada by registered mail, return receipt requested. Mailing by library rate is false economy, since saving a few dollars in no way compensates for the potential loss of irreplaceable specimens. Boxes of books are extremely heavy, and even solid metal containers have arrived crushed when sent to me library rate. Much more care is taken of registered mail than any other type (even insured; insuring natural history specimens seems to me futile, as establishing a commercial value may be more trouble than it is worth and the specimens cannot be replaced for any money). Also, the return receipt provides an immediate and legally usable record that the specimens were received.

Referenced Cited

Crawford, R.L. 1983. Grid systems for recording specimen collection localities in North America. Systematic Zoology, 32 (4): 389-402.
Crawford, R.L. 1988a. An annotated checklist of the spiders of Washington. Burke Museum Contributions in Anthropology and Natural History, 5: 1-48.
Crawford, R.L. 1988b. Map location literacy: How well does Johnny Geologist read?: discussion. Geological Society of America Bulletin, 100: 1847-1849.
Crawford, R.L. 1988c. Comparison and measurement of camera lucida images in arthropod taxonomy. Microscopy: Journal of the Quekett Microscopical Club, 36 (1-2): 85-86.
Levi, H.W. 1966. The care of alcoholic collections of small invertebrates. Systematic Zoology 15 (3): 183-188.

BURKE MUSEUM RECEIPT OF SPECIMENS

Arachnology Box 353010, University of Washington, Seattle, WA 98195-3010, USA

To:

Date _______________ By _____________

Loan _____ Exchange _____ Other _________

How shipped _________ Insured for _______

LOAN DUE _________________________

Returned ______________________________

Condition (UWBM only) __________________

PLEASE SIGN AND RETURN ONE COPY UPON RECEPT OF MATERIAL
Undersigned agrees to return all loaned material described herein by due date stated above.

Received in good condition **X** __________________________ _______________
(signature) (date)

Description of material:

INSTRUCTIONS FOR BORROWERS

Burke Museum Arachnid Collections

1. Please sign one copy of this loan form and return it immediately on receipt of the specimens; the other copy is for your files.

2. We would greatly appreciate return of this loan by the specified due date; if an extension of the period is needed, this must be requested in writing. Overdue loans for which no extension has been requested will be recalled.

3. Please do not insert new determination labels unless your species determination differs from the existing one. If that is the case, insert the new label between the glass of the vial and the existing determination. This is because specimens are often damaged by excessive labels forced into the specimen area of the vial.

4. Dissection of genitalia is permitted unless otherwise specified. If clearing is necessary, use a temporary clearing agent such as clove oil, not a permanent macerating agent such as potassium hydroxide. Dissected genitalia should be restored to the original vial in a microvial consisting of a short cut section of microcapillary tubing, plugged at both ends with cotton, or other vials of equivalent diameter (1-1.5 mm). If small microvials are not available, we will supply some. Do not use larger microvials; when the vials are transported, these bump and damage the specimens. Genitalia circa 1 mm or more in diameter can be left loose in the vial.

5. If for any reason it is necessary to transfer specimens to a new vial, be sure to transfer all labels and dissected parts along with the specimens.

6. Please keep the same stoppers in the same vials, as received. When restoppering vials, try to avoid smearing the locality numbers; however, we realize this may sometimes be unavoidable.

7. Specimens are preserved in 70% ethanol; please refill with the same preservative. To prevent stranding of specimens above the preservative, vials must be filled completely (stoppers can be inserted in full vials with a wire).

8. When returning vials, please wrap them individually as they were when received.

9. If you abbreviate depositories in your publications, please use UWBM for our collection.

THE OBJECT OF THE ABOVE GUIDELINES IS THE SAFETY AND LONGEVITY OF THE SPECIMENS.