

CARNIVOROUS PLANT NEWSLETTER

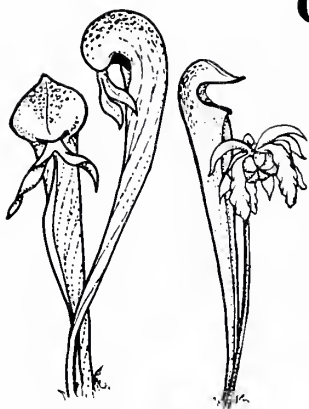
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Front Cover: *U. humboldtii* seeds, photograph by Thomas Carow.

Back Cover: Flowers of *U. nelumbifolia*. Photograph by Fernando Rivadavia.

Carnivorous Plant Newsletter is dedicated to spreading knowledge and news related to carnivorous plants. Reader contributions are essential for this mission to be successful. Do not hesitate to contact the editors with information about your plants, conservation projects, field trips, or noteworthy events. Contributors should review the "Instructions to Authors" printed in the March issue of each year. Advertisers should contact the editors. Views expressed in this publication are those of the authors, not the editorial staff.

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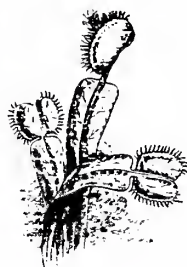
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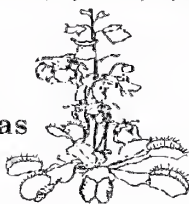
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UTRICULARIA NELUMBIFOLIA GARDN. AT LAST!

FERNANDO RIVADAVIA • Rua Inacio Pedroso 230 • Sao Paulo, S.P. 05612-050 • Brazil • fe_riva@uol.com.br

Keywords: observations: *Utricularia humboldtii*, *Utricularia nelumbifolia*.

Utricularia nelumbifolia Gardn. is one of the largest species in the genus. It is closely related to the other large species *U. reniformis* St.Hil. and *U. humboldtii* Schomb.—they are all from section *Iperua* P. Taylor. These three species are the only ones known to grow inside the water-filled leaf axils of bromeliads. While *U. nelumbifolia* thrives exclusively inside bromeliads, *U. reniformis* is mostly a terrestrial and is only very rarely found growing among the dead leaves at the base of bromeliads. *U. humboldtii* is most often found in bromeliads, but can also be observed in boggy soils surrounding the host bromeliads, and occasionally even inside pitchers of *Heliamphora*.

Although widespread on highlands of eastern Brazil (Taylor, 1989), *U. nelumbifolia* is elusive and it took me many years of intense searching before I finally found it in early 1996. For a while I even doubted it existed at all, thinking it was an artifact, the result of a few mutant leaves of *U. reniformis* growing in an exceptional habitat inside bromeliads. Ironically, I saw my first live *U. nelumbifolia* in December 1995, growing inside bromeliads cultivated at the Rio de Janeiro Botanic Garden. Although overwhelmingly happy to finally see *U. nelumbifolia* alive, I was nonetheless frustrated that they were in cultivation and in such poor health, barely alive in the hot coastal plains after having accidentally hitchhiked from their highland habitats along with their host bromeliads when these were collected.

In February/March 1996 I drove approximately 3400km all around southeastern Brazil, during a ten-day carnivorous plant marathon with two friends: Fábio Pinheiro from São Paulo, Brazil, and Joe Mullins, visiting from Ireland. While exploring highlands in eastern Minas Gerais state, we were lucky to meet Lúcio Leoni, who maintains an herbarium in the town of Carangola and knew much of the region's native flora. To our surprise, he knew a few carnivorous plant locations in the area, including a *U. nelumbifolia* population.

Lúcio took us to a beautiful mountaintop on the Serra da Araponga where we saw *Drosera villosa* St.Hil., *Genlisea lobata* Fromm-Trinta, and *U. longifolia* Gardn. This was a new recording for *G. lobata*, which until then was only known from the type location, the Serra do Caparaó, approximately 80 km to the north-east. Yet *U. longifolia* stole our attention from *G. lobata*. Not only was the whole grassy mountain top covered with millions of *U. longifolia* leaves, but there were also countless flower scapes. Each of these scapes was highly branched, with more flowers than I had ever seen on any other specimens of this species. But even more exciting was the fact that about half the *U. longifolia* lacked purple pigment and had white flowers! What a magnificent place that turned out to be!

On our way back to Carangola, Lúcio pointed to a group of mountains next to the terribly bumpy dirt road we were driving along. He claimed that the granite cliffs were absolutely covered with the bromeliad *Vriesia extensa* (L.B.Smith) J.R.Grant, and that these were in turn packed tight with *U. nelumbifolia*. We immediately decided to go there on the following day, of course!

We woke up early the next day to a beautiful cloudless morning, gobbled down

breakfast, and jumped into the car with all our gear. We could not wait to see *U. nelumbifolia*! After driving for about an hour (partly along a horrible dirt road full of holes and rocks which left numerous scrapes underneath my car), I parked at the entrance of a small ranch. We hiked up an increasingly steep hillside, passing through cow pastures, under barbed-wire fences, and over streams. At one point, stopping for a short rest, huffing and puffing with exhaustion, wiping the streams of sweat pouring down my forehead, I glanced upwards towards our destination. Squinting in the brightness of the daylight, I could see that the smooth rock face above us was polka-dotted with bromeliads by the thousands—no, millions! Would this be it? Would I finally see *U. nelumbifolia* in the wild?

That February 18 will forever remain deeply engraved in my mind as one of the most memorable days of my carnivorous plant travels, and I believe Fábio and Joe feel the same way. At about 1200m altitude, there was a brief transition between the short trees growing in brick-red lateritic soil and the bare rock surface covered with large bromeliads. The mountainside had an inclination of about 45 degrees at this point, but higher above the smooth rock curved upwards increasingly, becoming a vertical cliff. My lungs were burning, and I was getting more and more excited with each step. By the time I reached the transition zone, I felt like my heart was beating in my throat with the anticipation!

I stopped at the first bromeliad I came upon and began lifting up and looking under each of its long dark-green leaves in quick succession, like a monkey looking for ticks on its companion. Nothing. I went to the next bromeliad and proceeded to browse through it as well. Once again: nothing! I began to frantically walk from one bromeliad to another, sticking my face into each like a dog sniffing out other dogs. Joe and Fábio soon caught up with me and joined my frenzy. And still we could find no *U. nelumbifolia*! Starting to feel a terrible emptiness at the pit of my stomach, I continued moving slowly uphill, zigzagging from one bromeliad to another, my eyes radar dishes wandering from side to side.

Suddenly I spotted something and froze in mid-step. Was it wishful thinking or was that a circular leaf on a long petiole sticking out of a bromeliad? In silence, afraid to utter what I was already screaming in my mind, I cautiously approached the bromeliad in a few quick nervous jerks, knelt down on the rock next to it as if in prayer. Was that truly a *U. nelumbifolia* leaf, or maybe a heat-induced hallucination? My hand unexpectedly acted on its own, zapping out and plucking the suspicious-looking leaf. A fraction of a second was enough to confirm that it was not a product of my imagination but truly a round peltate *Utricularia* leaf (see Figures 1, 2). I immediately shouted out to Joe and Fábio, “NE-LUM-BI-FO-LIAAAAA!!” They quickly clambered up the steep mountainside towards me as whoops of joy escaped my throat and I jumped up and down, commemorating. I could not believe that I had finally found *U. nelumbifolia* in nature after so many years!

As soon as the initial explosion of joy abated slightly, we continued exploring further uphill and kept on finding more and more *U. nelumbifolia*—there were tons of it! Some bromeliads were absolutely infested with this *Utricularia*, all the way from the outermost dead and deteriorated leaf axils right up to the central water tank. We were even fortunate enough to find a few open *U. nelumbifolia* flowers. The inflorescences were similar to those of *U. reniformis* in shape and size, reaching between 50 and 80cm in length (see Back Cover). The flowers were around 3-4 cm long and wide, colored in a beautiful bright pink-lilac with two vertical yellow stripes on an inflated bulge at the base of the lower lip, surrounded by a dark pink-purple patch. Strangely, none of the inflorescences showed any signs of ovaries swollen with seeds, nor of old spent seed capsules.

According to Taylor (1989), the traps of *U. nelumbifolia* are 1.5-2.5 mm long;



Figure 1: *U. nelumbifolia* inhabiting *Vriesia extensa* at Serra da Araponga.

larger than those of *U. reniformis* (0.7-1.5 mm), but tiny in comparison to those of *U. humboldtii* (5-12 mm). The leaf petioles of *U. nelumbifolia* may reach 45 cm in length and the peltate circular lamina may be up to 10 cm in diameter (Taylor, 1989). Most of the *U. nelumbifolia* leaves arise from stolons tightly packed within the bromeliad leaf axils, but we found occasional small (often reniform) leaves with short petioles on the so-called “aerial” stolons.

Aerial stolons are probably the most amazing feature of *U. nelumbifolia*. These organs are also present in *U. humboldtii* and are a fantastic adaptation to life in bromeliads. I had the chance to study *U. humboldtii* in the wild between December 1998 and January 1999 while botanizing Mt. Neblina, on the Brazil-Venezuela border and made some interesting comparisons with *U. nelumbifolia*.

Typical mature stolons of *U. nelumbifolia* and *U. humboldtii* are thick and white in color, although occasionally slightly greenish—possibly because some sunlight may get past the younger, translucent bromeliad leaves. Aerial stolons are atypical in that they are very thin and dark-green and most importantly because they initially grow vertically like a flower scape before suddenly curving back downwards into an inverted U-shape. If it encounters a bromeliad leaf axil on its descending route, the tip of the aerial stolon “germinates,” branching numerous times just below the surface of the water.

In *U. nelumbifolia* these aerial stolons reach deep into the axils of the tightly-layered bromeliad leaves, like a grasping claw, acquiring thus a strong foothold (see Figure 2). At the Serra da Araponga we quickly discovered how well ensconced *U. nelumbifolia* was, as our initial attempts to obtain stolons from within the bromeliad leaf axils by pulling on the long petioles and aerial stolons of this *Utricularia* resulted only in broken bits and pieces of plant parts. Yet at Mt. Neblina it was quite the opposite—I had to be careful not to pull whole plants out of the bromeliads by yanking on a single leaf! I think this difference was mostly due to the different anatomy of the host *Brocchinia* species at Mt. Neblina which had more loose and open leaf rosettes.

As for the function of these aerial stolons, Taylor (1989) claims that they are a means of spreading from one bromeliad into another. But after studying both *U. nelumbifolia* and *U. humboldtii* in the wild and in cultivation, I have a different hypothesis based on the following four observations.



Figure 2: The outer leaves of *V. extensa* removed to reveal *U. nelumbifolia*.

1)Although aerial stolons may reach more than a meter in length (Taylor, 1989), I noticed that each one usually lands only 5-20cm away from where it originally emerged. Furthermore, I observed that the host bromeliads were usually located too far apart from each other to be within the reach of *U. nelumbifolia* aerial stolons arising from neighboring plants. Therefore the aerial stolons almost always grew out of and back into the same bromeliad.

2)Because the younger leaves of bromeliads stick out more horizontally than the older leaves (which are packed tightly around the base of the bromeliad), the aerial stolons of *U. nelumbifolia* and *U. humboldtii* mostly descend into the axils of younger leaves closer to the center of the bromeliad rosettes.

3)The water-tight seals between the leaves of each concentric ring in bromeliad rosettes are very smoothly-fit, possibly too perfect to allow the passage of the *Utricularia* stolons. If this is true, then the continuous growth of a host bromeliad would slowly but continuously push *U. nelumbifolia* or *U. humboldtii* outwards from the center, eventually leading it to certain death among the old bromeliad leaves as these dried and decomposed.

4)The aerial stolons of *U. nelumbifolia* and *U. humboldtii* appear to emerge most frequently from outermost leaf axils in a bromeliad rosette, where the water pools have already dried up or been squeezed out as the leaves become more tightly-packed.

Based on these observations, I believe that it is likely that the main function of the aerial stolons of *U. nelumbifolia* and *U. humboldtii* is not to colonize new bromeliads—which are usually too far away to be reached by the stolons—but rather is to continuously recolonize the central parts of the same host bromeliad. If the function of the aerial stolons was to colonize bromeliads other than the host, they would have to be able to reach farther than they actually do. Based on my fourth observation above, I wonder if the production of aerial stolons is triggered by a lack-of-water-related stress?



Figure 3: Fruit and flowers of *U. nelumbifolia* at Serra dos Órgãos.

Anyway, back to the Serra da Araponga, where after a few hours of studying, photographing, herborizing, collecting, and simply drooling over *U. nelumbifolia*, we finally agreed to trudge back downhill towards the car. Our feet ached tremendously from the strain of attempting to keep our balance for so long on that steep diagonal incline where the bromeliads grew. The heat and intense sunlight on that treeless terrain had been a bit of a problem too, but I am sure it would have been much worse if it had rained. I would not like to find out how slippery that smooth bromeliad-covered rock surface becomes when wet!

We collected *U. nelumbifolia* stolons for cultivation, but we did not know if they would survive inside the species of bromeliads we had back home, and certainly did not know if they would accept a home of sphagnum moss. Therefore, we also lugged down the mountain two or three small—but heavy and cumbersome—specimens of the bromeliad *Vriesia extensa* which contained *U. nelumbifolia*. It was difficult to pry these bromeliads off the rock, they are more tightly rooted than may seem possible!

Surprisingly, *U. nelumbifolia* survived very well the following week or so, bouncing around in the hot and stuffy trunk of my car before we arrived back to São Paulo. We even found more *U. nelumbifolia* towards the end of that trip in a similar habitat at approximately 1100m of altitude the Serra dos Órgãos highlands in Rio de Janeiro state, where they grew in two different (but unidentified) species of bromeliads. I revisited this site in October 1999 and found several flowering specimens, including one which had six open flowers on a single scape! (See Figure 3)

I am now very happy to say that *U. nelumbifolia* is still thriving in cultivation, growing much better for us here in Brazil than *U. humboldtii* ever did. It is even being successfully cultivated outside bromeliads, in live *Sphagnum* moss, by my friend Marcelo Fontana. In fact Joe has helped us introduce *U. nelumbifolia* to cultivation in Europe and hopefully this wonderful species will soon be commonly traded among carnivorous plant growers.

Although *U. nelumbifolia* has flowered in cultivation several times here in São Paulo and produced plenty of seeds, these are unfortunately short-lived and very fragile. The seeds of *U. nelumbifolia* are small seedlings surrounded by a thin papery sheath, and are viable only while green, much like the seeds of *U. humboldtii* (see Front Cover). On contact with water, each seed sheath breaks open and releases the three-dimensional snowflake-shaped plantlet. Those of *U. nelumbifolia* appear to have an indefinite number of leaf segments, each with a widely-spreading forked tip. The seeds should be treated like the gemmae of pygmy sundews, and must not be allowed to dry out. It should be possible to transport *U. nelumbifolia* by keeping the seeds or seedlings wrapped in moist tissue paper or live *Sphagnum*. Hopefully this way this magnificent species will soon be more widely cultivated in carnivorous plant collections around the world.

I was most surprised one day in early 2000 to discover *U. nelumbifolia* growing in my hometown of São Paulo! Walking around one of the busiest street corners in the city, I happened to pass by a huge bank with a beautiful bromeliad garden full of *Vriesia imperialis* Carrière specimens. I could not help stopping to admire it from across the tall fence, and nearly fell over backwards as I suddenly noticed that several of the plants were packed with *U. nelumbifolia* leaves! Although momentarily happy with this surprise, I nonetheless immediately felt a hard kick to my stomach as I realised what the presence of *U. nelumbifolia* in those bromeliads actually meant: that they were illegally wild-collected, and that some beautiful cliffside had been stripped clean of those huge bromeliads, which must take decades to reach their full size. And the worse part is that bromeliads have become increasingly popular in landscaping around the city...

Acknowledgements

I would like to thank Joe Mullins and Fábio Pinheiro for their company on that memorable trip, congratulations to Fábio, Marcelo Fontana, and Maurício Piliackas for keeping *U. nelumbifolia* alive in cultivation here in São Paulo, and a big thank you to Lúcio Leoni who pointed out the exact place where we finally saw wild *U. nelumbifolia*.

Reference:

Taylor, P. 1989, The Genus *Utricularia*—a taxonomic monograph, Kew Bull. Additional Series XIV, HMSO, London.

Looking Back: CPN 25 years ago

Susan Verhoek-Williams wrote a bizarre article about ancient and modern medical beliefs and uses of carnivorous plants: "In France, sorcerers...used *Drosera* in their potions....one who searched for a sundew and rubbed its leaves over his skin on St. John's Eve would become indefatigable. *Searching* was necessary; a person who simply blundered upon the plants would be confounded and never find the spot again.

"It was easy to tell if *Drosera* were nearby, at least so it was believed in the Bourbon region of France. There, it was said, the plants glowed at night and by day green woodpeckers marked the spot; they could be seen flying strangely as they maneuvered to pluck the sundews, which were used to harden their beaks.

"Sorcerers and laymen alike collected the plant on St. John's Eve (midsummer eve), midnight being considered the time to gather the most effective plants. The gathering was perilous; the collector had to do his collecting walking backwards to avoid being followed by the devil. Devil or no, walking backwards in a bog at midnight would give a collector an uneasy feeling."

NEW CULTIVARS

Keywords: cultivar: *Pinguicula* 'Titan', *Sarracenia leucophylla* 'Schnell's Ghost'.

Pinguicula 'Titan'

Received: 20 October 1999

This hybrid *Pinguicula* was created by me on 17 July 1987. The seed parent was a fragrant clone of *P. agnata* we grow at California State University at Fullerton. This is the only fragrant *Pinguicula* we have, and its taxonomic status is still uncertain. The pollen parent was an unidentified *Pinguicula* collected by David Verity near Guanajuato, Mexico in 1975. The pollen parent forms a hibernaculum below the surface of the ground, sometimes at a depth of over 1.5 cm (1/2 inch), and has flowers similar in form to *P. gypsicola*. It may be *P. macrophylla*.

At its largest, the leaves of *Pinguicula* 'Titan' can exceed the confines of a 15 cm (six inch) pot! The leaves resemble the pollen parent's, in having a longer and more pronounced petiole than the leaves of the seed parent. However, the flower shape definitely favors the seed parent, but the blue edges have been replaced with an even magenta glow. A slight fragrance is also evident. It forms a large subterranean hibernaculum, but has a relatively short dormant period.

Pinguicula 'Titan' is very vigorous and easy to grow (Figure 1). We use a general mix for carnivorous plants (2 parts coco peat, 2 parts peat moss, 1 part fine orchid bark, 3 parts #20 quartz sand, 1-1.5 parts coarse perlite) to which we add a bit of dolomite and gypsum (1 part to 800 parts potting mix). A 5 cm (2 inch) layer of perlite is placed at the bottom of the pot for added drainage and enhanced aeration. We grow it under lights and natural lighting. It is being sold by a number of stores, such as Booman Floral of Vista California. It survives on store shelves longer than all other carnivorous plants, flowering there even after Venus Flytraps, sundews, and *Sarracenia* have died.

The cultivar name 'Titan' was chosen both to indicate the plants large size, but also to commemorate the nickname for California State University, Fullerton. I nominated this name some time in 1998, and it was sent (by Barry Meyers-Rice) for registration on 20 October 1999. *Pinguicula* 'Titan' should be propagated by vegetative means only in order to preserve its distinctive characters.

—LEO SONG, JR. • Biological Sciences Greenhouse Complex • California State University • Fullerton, CA 92834-6850 • USA • leo@carnivorousplants.org

Sarracenia leucophylla 'Schnell's Ghost'

Received: 4 February 2000

One of the peculiarities found in many of the red-flowered species of *Sarracenia* are the so-called aberrant colour forms. In addition to the range of pink and red colours found in many individuals of these species, plants are occasionally discovered which have pure yellow flowers. The genetics governing this effect are well documented (Sheridan & Scholl, 1996; Sheridan, 1997) and are known to affect red colour production in either the flower or the entire plant.

Of particular merit is the yellow-flowered clone of *Sarracenia leucophylla*. This plant has, in addition to the pure yellow flower, a complete absence of red colouring in the upper-pitcher tube and lid. The almost pure white lid is innervated with nar-



Figure 1: *Pinguicula* 'Titan' photographed by John Brittnacher.



Figure 2: *Sarracenia leucophylla* 'Schnell's Ghost', photo by Phil Wilson.



Figure 3: *Sarracenia leucophylla* 'Schnell's Ghost', photo by John Constable.

row green veins; the white colour includes the inner lid-surface and column, and extends to the upper section of the pitcher tube. The only red colour in the plant is found in the growing points of emerging pitchers and in aging pitchers as they start to senesce. Because of this, the plant cannot be described as an anthocyanin-free plant (or green mutant, as erroneously reported by D'Amato (1998, page 82).

The yellow-flowered *Sarracenia leucophylla* was originally collected by Donald Schnell during the summer of 1972. At the time of this collection the plant was not in flower; presumably the intense white colouration of the plant's leaves attracted his attention. Schnell sent a piece of the plant to Steven Clemesha in Australia, who adjusted its growth habit to southern hemisphere seasons, and grew the plant to maturity. It was not until the plant flowered in September 1974 that Clemesha discovered that the plant also produced a pure yellow flower (Clemesha, 1999, personal communication).

Some years later Martin Cheek obtained plants from Clemesha, propagated them, and offered specimens for sale with the unregistered name of "Schnell's Ghost". At the time Martin produced a catalogue of plants which contained a full description of the cultivar (Cheek, 1990, page 2). Although references to this plant's "very pale ghost-like qualities" were made in private correspondence as early as 1972 (Clemesha, 1999, personal communication), the first printed reference to its "ghost-like" qualities was in Schnell (1989):

"The pitcher top is so pale and the lack of red venation gives the plant an almost ghost-like appearance and it stands out readily in a stand of typical plants, even when not in yellow flower. This plant bore a yellow flower the following spring in cultivation...."

In the early 1990s Alan Hindle, a grower and collector of *Sarracenia* forms in the UK, began selling a yellow-flowered *S. leucophylla*. Alan Hindle received his original stock from Bruce Bednar in the USA, so this plant subsequently became known among UK growers as the "Bednar clone". Bednar reported that he obtained his plant from Clemesha in Australia, so the "Bednar clone" is the same plant as the "Schnell's Ghost" plant (Bednar, 1999, personal communication). Other unestablished names that have been used to label this *S. leucophylla* plant include "Alba" and "Yellow Flower".

Several other distinct clones of the species with yellow flowers have subsequently been found. For instance, there is at least one clone from the Citronelle region in southern Alabama. The plants are again characterised by having predominantly white colouration in the lid and upper pitcher, and a yellow flower. I am registering the cultivar name *Sarracenia leucophylla* 'Schnell's Ghost', which should be applied to all clones of the species with yellow flowers and predominantly white coloured lids and upper pitchers (Figures 2 and 3). Since seed from self-pollinated individuals of this clone breed true (and presumably between different clones of this cultivar), *Sarracenia leucophylla* 'Schnell's Ghost' may be propagated both asexually from cuttings and sexually from seed, as long as the cultivar characters are maintained.

As mentioned above, *Sarracenia leucophylla* 'Schnell's Ghost' does have some red pigmentation in the growing points. In contrast, collections of *Sarracenia leucophylla* plants completely lacking anthocyanin have been reported (Sheridan & Scholl, 1996). The cultivar description of *Sarracenia leucophylla* 'Schnell's Ghost' does not include these plants. I am happy to report that, despite fears that *Sarracenia* 'Schnell's Ghost' had become extinct (Meyers-Rice, 2000), it is quite alive both in England and in the USA.

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<i>Dionaea muscipula</i>	<i>D. nidiformis</i>
<i>Drosera aliciae</i>	<i>D. stenopetala</i>
<i>D. anglica</i> —Oregon	<i>Nepenthes stenophylla</i>
<i>D. auriculata</i>	<i>N. gymnamphora</i> ?
<i>D. burmannii</i>	<i>N. ventricosa</i> ?
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<i>D. capensis</i> —purple flower, narrow leaf	<i>S. flava</i>
<i>D. capensis</i> —white flower	<i>S. leucophylla</i>
<i>D. capensis</i> —typical/wide leaf	<i>S. minor</i>
<i>D. capillaris</i>	<i>S. psittacina</i>
<i>D. dielsiana</i>	<i>S. purpurea</i> subsp. <i>purpurea</i> —Quebec, Canada
<i>D. filiformis</i> var. <i>filiformis</i>	<i>S. (flava</i> ? <i>leucophylla)</i> ?
<i>D. filiformis</i> var. <i>tracyi</i>	<i>Utricularia multifida</i>
<i>D. intermedia</i>	<i>U. violacea</i>
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UPTAKE OF THE AMINO ACID ALANINE BY DIGESTIVE LEAVES: PROOF OF CARNIVORY IN THE TROPICAL LIANA *TRIPHYOPHYLLUM PELTATUM* (DIONCOPHYLLACEAE)¹

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Keywords: carnivory: prey, *Triphyophyllum peltatum*.

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This paper is dedicated to Prof. Wittko Francke, on the occasion of his 60th birthday.

Summary

The uptake of organic matter by the insect-trapping glandular leaves of the Western tropical African liana *Triphyophyllum peltatum* (Dioncophyllaceae) is demonstrated for the first time. After feeding carbon-13 labelled L-alanine to the trapping leaves, the label is detected in apical shoot parts and normal (non-trapping) leaves within 2 d of application. With this proof of resorption and transport, the carnivorous syndrome of *T. peltatum* is demonstrated to be complete. The prey composition reveals the glandular leaves of *T. peltatum* to be traps for flying insects, predominantly.

Introduction

The glandular leaves produced in juvenile stages of the rare West African liana *Triphyophyllum peltatum* (Dioncophyllaceae, see Figure 1) trap considerable amounts of arthropods (Green *et al.*, 1979). The homology of the stalked and sessile glands on these organs to those of *Drosophyllum lusitanicum* (Drosophyllaceae) was very early recognized by plant anatomists (Metcalf, 1951). From these data and the detection of endogenous proteolytic enzymes in the secretions of these glands, the hypothesis of carnivory in *T. peltatum* was derived (Marburger, 1979; Green *et al.*, 1979). The resorption of organic matter by the trapping leaves has, however, not been demonstrated.

Recent genetic studies (Fay *et al.*, 1997; Meimberg *et al.*, 2000) indicate a close phylogenetic relationship of the Dioncophyllaceae to the carnivorous families Drosophyllaceae (with sticky leaves), Nepenthaceae (with pitfalls), and Droseraceae (with sticky leaves or spring traps), as well as the non-carnivorous families Ancistrocladaceae, Plumbaginaceae, and Polygonaceae. Phytochemical (Bringmann

¹Part 144 in the series 'acetogenic isoquinoline alkaloids'. For part 143, see Bringmann, G., Mühlbacher, J., Repges, C., and Fleischhauer, J. 2000. MD-based CD calculations on the naphthylisoquinoline alkaloid dioncophylline A, for the assignment of the absolute axial configuration. *J. Comp. Chem.* (submitted).

& Pokorny, 1995; Bringmann *et al.*, 1998) and anatomical (Schlauer, 1997) similarities support grouping these families together in a caryophyllid clade (a redefined order Nepenthales).

First data on carnivorous properties of the glandular leaves formed at certain developmental stages of *T. peltatum* (Dioncophyllaceae) were obtained earlier (Green *et al.*, 1979; Marburger, 1979). Here we describe experiments that demonstrate the ability of *T. peltatum* to absorb the amino acid alanine applied to the glandular leaves, which completes the knowledge of the carnivorous syndrome in this species. Furthermore, the fauna trapped by these leaves has been analyzed in order to further characterize carnivory in *T. peltatum*.

Materials and Methods

Plant material: 24 specimens of *T. peltatum* (Hutch. & Dalz.) Airy Shaw bearing glandular leaves were fed with 500 mg [2,3-¹³C]-labelled L-alanine (Promochem, Wesel, Germany) at the Parc de Taï (Ivory Coast) in April 1996, just before the beginning of the rainy season (Figure 2), and harvested after 2 d incubation (Bringmann *et al.*, 1996). The material was air dried at the Centre National de Floristique, Abidjan (Ivory Coast) and stored at 4°C in Würzburg, Germany. The prey animals attached to the traps were removed and determined taxonomically. All work and collection of material for research was performed in accordance with the official permit conditions of Ivory Coast. Voucher specimens of *T. peltatum* are deposited at the Centre National de Floristique, Abidjan (UCJ) and at Herb. Bringmann, Institute of Organic Chemistry, Würzburg.

Extraction and analysis of plant material: Dried *T. peltatum* plants were divided into roots, basal stems, apical stems, normal leaves, and trapping leaves, then washed with distilled water (3-5 times) until alanine was no longer detected in the supernatant. The material was then lyophilized and homogenized. The resulting finely powdered fractions were extracted three times with the tenfold (w/w) amount of distilled water, each with ultrasonification for 2 d at 22°C and subsequent filtration. 5 ml each of the aqueous extracts were filtered through preconditioned RP-18 columns (Waters, Eschborn, Germany) and the columns were washed with 1 ml of distilled water each. The procedure was repeated with preconditioned anion exchange columns. The solvent was removed by lyophilization. The residues were dissolved in 1 ml of distilled water, neutralized with 1N NaOH, and applied on preconditioned cation exchange columns. The columns were washed with 1 ml of distilled water each and vacuum-dried for 1 min. The cations (including alanine) were eluted by 1.5 ml of 1N HCl each, the eluates were dried at 22°C, redissolved in a solution of 0.108 ml (1.5 mmol) thionyl chloride in 1 ml of isopropanol each, refluxed for 1 h and subsequently deprived of the solvent by evaporation. The residues were treated with 50 µl of trifluoroacetic acid anhydride in 400 µl of dichloromethane each and stirred for 1 h at 22°C. The solvent was removed by superfusion with nitrogen, the residues obtained were redissolved in 300 µl of toluene and analyzed by GC-MS.

GC-MS: A gas chromatograph HP 5890 Series II with on-column injector (Hewlett-Packard, Avondale, USA) was coupled directly with a quadrupole mass spectrometer MSD 5971 A (Hewlett-Packard, Avondale, USA). The temperature of the transfer line was 280°C, resulting in an ion source temperature of 180°C. A DB-17 column (J&W Scientific, 10 m × 0.16 mm, film thickness 0.18 µm) with helium as the carrier at a pre-column pressure of 100 kPa was applied. Temperature program: 50°C (4 min), 6°C/min, 80°C, 60°C/min, 210°C (3 min). Alanine was analyzed as its *N*-trifluoroacetyl-L-alanine isopropyl ester. The label (fragment weights increased by two units) was detected by the shift in the ratio between the *m/z* 142

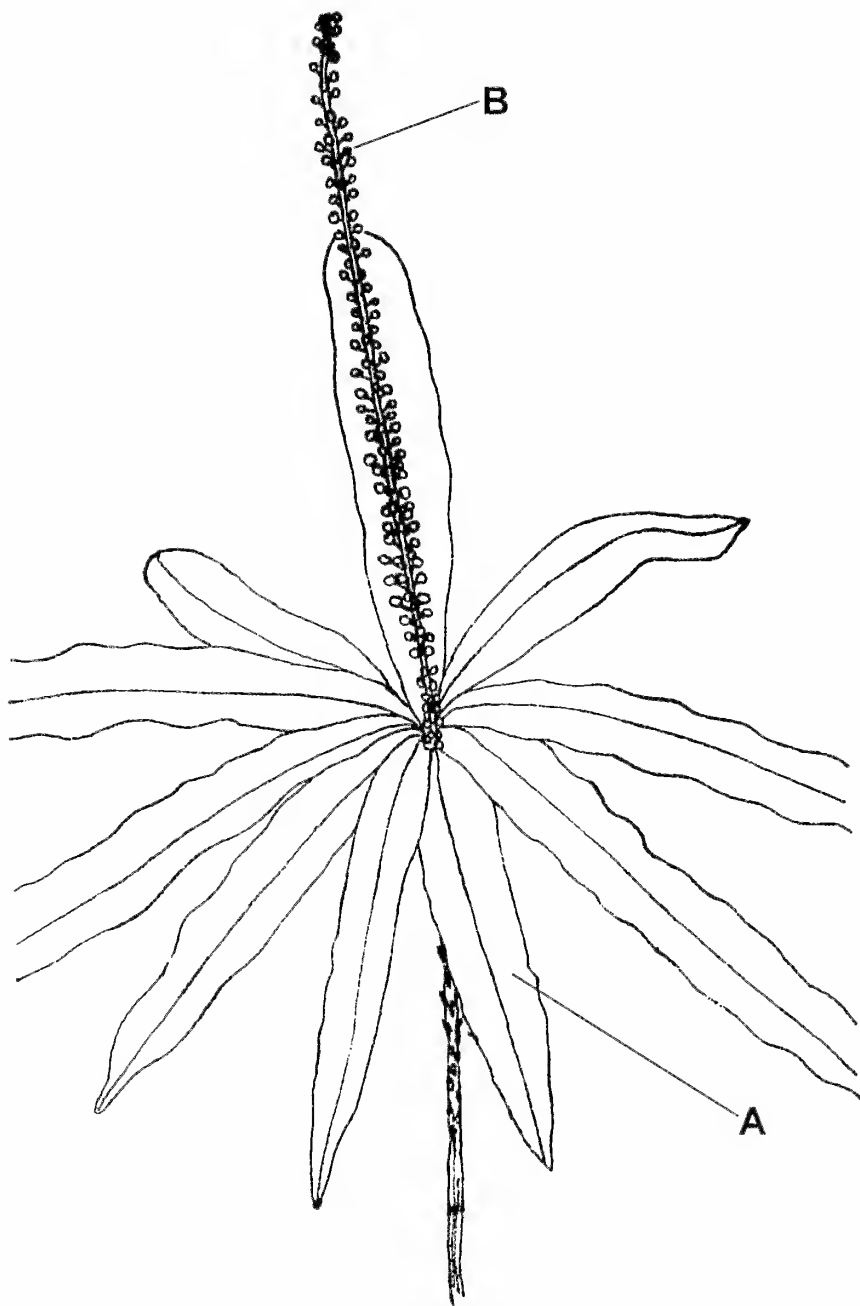


Figure 1: Juvenile plant of *T. peltatum* with rosette of normal leaves (A) and trapping leaf (B). Drawn by J. Schlauer

and 140 [M⁺- COO⁻iPr] peaks, which correspond to characteristic fragment ions of the respective derivatives. Unlabelled leaf tissue was extracted and derivatized in the same way and analyzed as a control. All values were obtained from triplicate measurements (from three separate chromatograms each).

Results

Alanine uptake and redistribution: After uptake, labelled alanine was found predominantly in the trapping leaves and the adjacent stem by GC-MS analysis. A somewhat smaller content of labelled alanine was detected in the normal (i.e. non-carnivorous) leaves of fed plants of *T. peltatum* (Table 1). No alanine (either labelled or unlabelled) was detected in the roots by the analytic method employed.

	Tissue				
	Control	Roots	Stem	Leaves	Traps
142/140	0.048±0.004	no alanine detected	0.538±0.031	0.101±0.046	1.163±0.188
Label/control	1	-	11.2	2.1	24.2

Table 1: Distribution of labelled alanine after feeding to *T. peltatum*. Relative abundance (mean, SE calculated from three independent experiments) of characteristic fragment ions of labelled (m/z 142) vs. Unlabelled (m/z 140) alanine in different parts of *T. peltatum* fed with ¹³C₂ labelled alanine and unfed control plants (likewise with glandular leaves) of the same species.



Figure 2: Feeding labelled alanine to a carnivorous plant of *T. peltatum* in the tropical rain forest in the Parc de Taï (Ivory Coast); interestingly this specimen bears two glandular leaves. Photo by H. Bringmann

Fauna trapped by T. peltatum: The invertebrates collected from the glandular leaves of 24 specimens of *T. peltatum* were determined to their order (Table 2). A total of 197 specimens was obtained. The mouthparts of the prey and the presence/absence of wings were investigated in order to estimate relative abundances of different prey types.

Members of following suborders, families, or subfamilies were found:

Diptera:

Brachycera, Nematocera, Sciaridae, Mycetophilidae, Anisopodidae, Phoridae, Ceratopogonidae, Scatopsidae, Macroceridae, Bolitophilidae, Dixidae

Coleoptera:

Chrysomelidae, Lycidae, Scolytidae, Curculionidae, Staphylinidae (predator), Elateridae, Malachiidae

Hymenoptera:

Formicidae, Myrmecinae (winged sexuals and few workers)

Aranea:

Theridiidae, Salticidae, Linyphiidae

Prey Type	no.	% of total	% winged	mouthparts
Insecta				
Isoptera	13	6.5	100	biting
Auchenorrhyncha	1	0.5		sucking
Heteroptera	2	1.0		sucking
Thysanoptera	1	0.5		sucking
Hymenoptera	43	21.8	93	biting
Coleoptera	41	20.8	100 ¹	biting
Diptera	80	40.6	100	licking, licking-sucking
Lepidoptera	2	1.0	50	sucking, biting (one larva)
Arachnida				
Aranea	12	6.0	0	
Acari	2	1.0	0	
Total	197			

Table 2: Prey trapped by *T. peltatum*. Twenty-four specimens of *T. peltatum* with glandular leaves were investigated. ¹Wings not always visible through elytra.

Discussion

These results demonstrate that *T. peltatum* takes up and redistributes alanine applied to the digestive glands. The relative concentration of incorporated alanine was highest at the trapping leaves, which is not unexpected since the plants were harvested only a brief time after application of the label (2 d). Since the material had been washed repeatedly, the high signal found was apparently caused by alanine that had really been taken up. A considerable proportion of the label (almost half of the relative concentration found in the trapping leaves) was detected in the stems, which had not been in any external contact with labelled alanine. This region should be the first one to be reached by compounds taken up by the glandular leaves. As the normal (not trapping) leaves showed significant (although lower) label concentrations, while the roots were devoid of label, the leaves might be a sink for nutrients derived from animal prey. This would also accord with previous results obtained for pitcher plants (Schulze *et al.*, 1997), in which non-trapping leaves were

found to be a sink for probably insect-derived nitrogen. The fact that no alanine was detected in the roots could be due to very low steady state concentrations of free (i.e. not protein-bound or otherwise fixed) alanine in these organs.

Previous to this investigation, the resorption of amino acids by the glandular leaves and the distribution of these metabolites to other tissues had been the missing evidence required to show that *T. peltatum* is able to perform the entire series of carnivorous activity—to attract, capture, kill, decompose and absorb animal prey to a nutritional benefit. Our findings complete the “carnivorous syndrome” (Juniper *et al.*, 1989): In the growth phase marked by the formation of glandular leaves, *T. peltatum* is, beyond doubt, a carnivorous plant.

The major groups of animals found attached to the glandular leaves of *T. peltatum* in Ivory Coast differ only slightly from those previously reported to be captured by the same species in Sierra Leone (Green *et al.*, 1979), while the trapping efficiency (197 identified carcasses from 24 plants) was apparently lower than in that study (164 captured arthropods from 8 plants). Like in Sierra Leone, the trapping season in Ivory Coast coincides with periods of peak activity of the prey (many winged, sexual specimens) at the onset of the rainy season. Most of the identified arthropods trapped by *T. peltatum* in this study were winged. Only few other animals were found to be trapped, although they appeared to be abundant everywhere, so that those species seem not to be specific prey of *T. peltatum*. Most specimens caught had biting or licking-sucking mouthparts. Surprisingly few Lepidoptera were captured, which could imply that the traps are not attractive to them.

Acknowledgements

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Writings from the Readership

REFINING THE TERRARIUM: ALTERNATIVE TECHNIQUES FOR THE INDOOR GARDENER

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The quantity of literature pertaining to the indoor cultivation of *Nepenthes* is far from overwhelming. The two essays I have found most useful appeared in the pages of this journal: "A Rainforest In The Basement: *Nepenthes* Cultivation Under Lights" (Butler, 1987), and "The Potted Terrarium" (D'Amato, 1996). Butler's piece described the construction of a basement grow-chamber for his "intermediate" *Nepenthes*. D'Amato's article, which is more important for our purposes, described two styles of terraria which were well-suited for a general carnivorous plant collection. Both of the foregoing works were instrumental in my early attempts to grow carnivorous plants, and especially *Nepenthes*, indoors. Nonetheless, neither article provides an optimal method for growing lowland *Nepenthes* indoors. In this article I will describe and advocate two modifications to D'Amato's "potted greenhouse style terrarium" (henceforth "potted terrarium"). These modifications yield improved growing conditions for lowland *Nepenthes* without any sacrifice in convenience. Furthermore, the "refined" potted terrarium has a few other useful applications. Before getting on to this, I will review D'Amato's article.

At the time of publication D'Amato's article was perhaps the most important discussion of carnivorous plant culture in terraria. I remember thanking him personally at a Bay Area Carnivorous Plant Society meeting for sharing such useful information. (The piece reappeared in his book, much supplemented.) In contrast with the paradigmatic terrarium containing a planted soil bed, D'Amato advocated simply placing potted plants directly on the terrarium floor. As he pointed out, this simple change allowed one to grow plants requiring a variety of different soils and soil moisture levels in the same terrarium. This versatility, I think, is the chief benefit of the potted terrarium; it constitutes a dramatic improvement over the planted tank.

While acknowledging the great utility of the potted terrarium, it proves deficient for the lowland *Nepenthes* grower in two areas. The first is watering. Unlike many carnivorous plants, *Nepenthes* will not tolerate standing in water. Hence, one must remove the plants, water them, let them drain, and then return them to the terrarium. This is incredibly inconvenient. The alternative D'Amato suggested is to sit each potted *Nepenthes* in "a shallow saucer and water overhead as soon as the water in the saucer evaporates" (D'Amato, 1998, p 277). Though he cautioned the

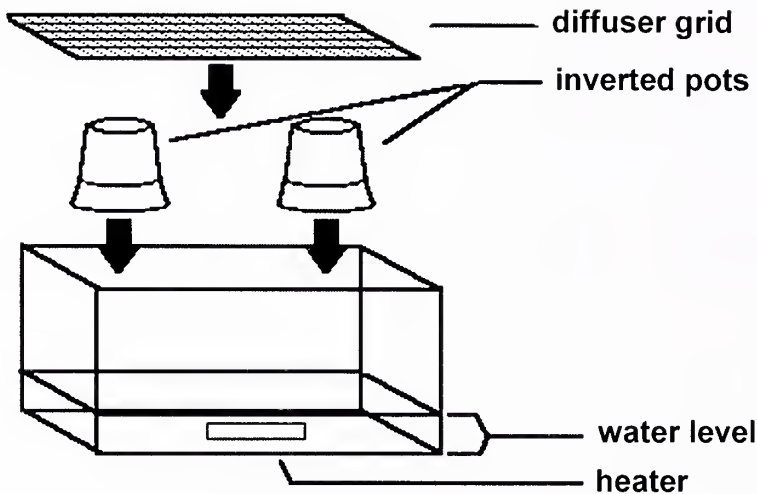


Figure 1: Setting up the refined terrarium.

reader not to let plants sit in deep water for extended periods, I find this method of watering unacceptably risky. Furthermore, Barry Meyers-Rice reports that he has had unsatisfactory results letting *Nepenthes* sit in water (personal communication, 1999).

The second area of difficulty is heating. Though some lowlanders will tolerate consistent temperatures around 18°C (approximately 65°F), most will not. Indeed, my clone of *N. ventricosa*, a highland species, completely ceased growth one winter when the room temperature never rose above 18°C (65°F). Even if the plants tolerate these lower temperatures, growth is very slow. A heater would help. Although many manufacturers produce electric heating mats, the mats tend to have a surface area far smaller than most terraria. Furthermore, most are designed to heat a seed-flat, not the large air/soil volume of a 120-200 liter (30-50 gallon) terrarium. Hence, these mats are of minimal utility (especially considering their cost). Again, D'Amato proposed an alternative. He suggested that one submerge an aquarium heater in a jar of water placed in the terrarium. Though aquarium heaters are cheaper than the aforementioned mats, they have their own shortcomings. The small volume of water in the jar evaporates rapidly, and while this does help increase humidity, the water must be continually topped-off. (Submersible aquarium heaters exposed to air may crack, creating a potentially hazardous situation.) Furthermore, these heaters do not produce enough heat to do the job.

In the next section, I will describe how to set up a “refined” potted terrarium, which incorporates my two modifications to the standard potted terrarium.

In addition to an empty aquarium, you will require a submersible aquarium heater and a sheet of plastic lighting diffuser grid. In essence, the bottom of the aquarium is used as a heated water reservoir, and the plastic grid acts as a raised platform or staging for the plants (see Figure 1). Submersible aquarium heaters are available in several power ranges, from 50 to 300 watts. Generally, the wattage desired is determined by the volume of water to be heated; 1.25 watts per liter (5 watts per gallon) is the aquarist’s rule of thumb. Assuming that one begins with a 220 liter (55 gallon) aquarium, a 7.5 cm (3 inch) deep water reservoir would be roughly 40 liters (10 gallons). The rule of thumb dictates that a 50 watt heater should be sufficient for a tank this size or smaller. However, it is important to remember that this heuristic was designed for the fishkeeper, not the *Nepenthes*



Figure 2: A 160 liter (40 gallon) terrarium containing *Nepenthes* and *Cephalotus*.



Figure 3: Young plants of *D. derbyensis* growing in heated water.

grower. Although we want to heat the water, our true object is to heat the much larger volume of air and soil in the tank. Hence, I recommend doubling the figure to 2.5 watts per liter (10 watts per gallon) of water. Following this new rule, I therefore use a 100 watt heater for tanks with volumes of 120-220 liters (30-55 gallons), and a 50 watt heater for smaller tanks. The heater is placed horizontally on the floor of the aquarium. Its thermostat should be set to 30°C (85°F) or its highest temperature setting, whichever is lower. The tank should then be filled with 7.5 cm (3 inches) of water, ensuring that the heater is completely submerged. Those who grow only highland plants may wish to consider putting the heater on the same timer as the lights, so that temperatures rise and fall each day. With my collection, however, I have found this unnecessary.

In the USA, lighting diffuser grid is available in 60 × 120 cm (2 × 4 foot) sheets, and is made of an inert plastic. In the USA, it sells at large hardware stores for about \$6 per sheet. Cut it to fit your terrarium floor. The grid will become the platform for your plants. Use overturned flower pots as stilts to elevate the grid off the terrarium floor. Make sure the grid will be above the water level in the terrarium, and will also provide plenty of room for your pots and the growing room your plants will require. The grid is rigid but not especially strong, so make certain that it is well supported at the center and edges. Once the flower pot supports, grid platform, and water are all in place, turn on the heater and arrange the *Nepenthes*.

The refined potted terrarium has many benefits. The ambient room temperature where I have my *Nepenthes* terraria (in a basement) is a constant 15-18°C (60-65°F), and the humidity is well below 50%. I grow my seedlings, unrooted cuttings and smaller rosetted plants in a 120 liter (30 gallon) terrarium. The platform is 18 cm (7 inches) above the aquarium floor. My larger plants grow in a 160 liter (40 gallon) tank, and its platform is only 10 cm (4 inches) above the floor (Figure 2). The plants can be watered in place; they drain freely, and the runoff keeps the reservoirs full. The plants sit 5-15 cm (2-6 inches) away from the light fixture. I have found that two fluorescent bulbs (the same length as the terraria) provide ample light when plants are this close. The air temperature inside both sealed terraria remains a satisfying 27°C (80°F), and the humidity stays constant at 90%. Given that I am using measly 100 watt heaters, this is a substantial temperature increase at a very low cost. Furthermore, air convection currents circulate warm moist air around the plants.

I have used this method quite successfully over the last few years with many plants, including *N. rafflesiana*, *N. truncata*, *N. gracilis*, *N. × hookeriana*, *N. ventricosa*, *N. × wrigleyana*, *N. × ventrata*, *N. × ventrata* × species, *N. veitchii* × *lowii*, and other unlabeled hybrids. (The inclusion of highland *Nepenthes* on this list may raise some eyebrows, but they grow well in these conditions!) *Cephalotus follicularis* has also grown quite vigorously in this environment; the 2 cm (3/4 inch) cutting I started with now requires a 15 cm (6 inch) diameter pot! My *N. truncata* provides the most dramatic demonstration of the refined terrarium's utility. When I obtained it, it was a sorry specimen with small, sickly leaves and dinky, poorly colored 3.75 cm (1.5 inch) pitchers. Just six months later, the plant began producing substantial 18 cm (7 inch) pitchers with rich coloration.

The refined terrarium can be modified for special applications. I particularly enjoy growing carnivorous plants from seed. After experimenting with several methods to heat seedlings, I achieved the best results by slightly modifying the method presented above. Instead of raising the platform above the water reservoir, I raise the water level to 2.5 cm (1 inch) above the platform. The plants sit in 25-27°C (78-80°F) water. Germination and growth of tropical *Drosera* and *Pinguicula* is quite rapid in such conditions. Also, the heat significantly reduces the occurrence of fungal attacks on newborns, and those that do occur are less severe. Many growers today are extremely interested in *Drosera* that are in section *Lasiocephala* (i.e.

related to *D. petiolaris*). I have found that these potent Aussies (e.g. *D. ordensis*, *D. derbyensis*, and *D. paradoxa*) really perk up when grown in heated water (Figure 3).

The method of terrarium culture presented here has provided excellent growing conditions for several genera of carnivorous plants. I sincerely hope that other growers will find this method useful, especially those who are not blessed with a greenhouse. Apartment dwellers, students, and others who crave choice rainforest plants can enjoy a lush indoor garden. Even greenhouse owners may find this method ideal for pampering a newly imported *N. clipeata*, or for experimenting with the exciting perennial tropical *Drosera*.

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NEWS AND VIEWS

Kenneth Skau (3176 Sunny Hollow Lane, Cincinnati OH 45239, ken.skau@uc.edu) wrote: The Krohn Conservatory in Cincinnati, Ohio, USA sponsored a "Join a Plant Society Weekend" February 26-27, and I made a display so the ICPS was represented. Barry Meyers-Rice (who was the person contacted by Krohn) provided some back issues of Carnivorous Plant Newsletter and membership application forms. Meanwhile, I made up a poster with some information about the society (shamelessly plagiarized from the web site) and some pictures of carnivorous plants. Unfortunately, I was out of town over that weekend and did not feel comfortable providing any of my plants for the event. There were over 3,000 visitors, which has encouraged the conservatory to make this a yearly event. The conservatory will be sponsoring a "Bug Fest" in the fall in which I hope to participate and display some live bug eaters.

David Parker (13516 Parker Ave., Grandview, MO 64030 USA, david.a.parker@mail.sprint.com) writes: I have been growing Venus flytraps and other carnivorous plants for the past eight years. The pride of my collection are twelve Venus flytraps that are over twenty years old. These plants produce very large traps. This summer I measured traps that were just over 5 cm (2 inches) long. I remove the flower stalks as soon as they appear in spring, and feed the plants constantly during the growing season. I feel these factors and the plants' ages are responsible for their large size. These plants continue to get bigger every year and it would not surprise me if they produce 6 cm (2 1/4 inches) traps next summer. I feed them wax worms that I get at the local bait or pet care stores. I find it is easier to handle them than crickets or other insects. They also seem to be very easy for the plants to digest—they do not rot and kill the trap leaves as long as one worm is fed to a trap. Just before I feed wax worms to the plants I place them in the refrigerator for 10-15 minutes. This keeps them immobile. After I place them in the traps, and the traps close, the wax worms warm and slowly start to move again. This triggers the traps into their digestive phase. The wax worms are not that strong and rarely escape or damage the trap.

Barry Meyers-Rice (P.O. Box 72741, Davis, CA 95617 USA) writes: The US postal service has announced that in 2001 it will publish four carnivorous plant stamps. *Dionaea muscipula*, *Sarracenia flava*, *Darlingtonia californica*, and *Drosera anglica* will be featured on the new 34 cent stamps.

BLACK VINE WEEVIL: A *SARRACENIA* AND *DARLINGTONIA* PEST

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Keywords: cultivation: *Darlingtonia*, pesticides, *Sarracenia*.

In the last few years I have heard from a few growers who have had their plants attacked by a pest relatively new to carnivorous plant growers—the black vine weevil, *Otiorhynchus sulcatus*. This plant can devastate *Sarracenia* and *Darlingtonia*.

The black vine weevil is a black, hard beetle slightly less than 2 cm long. It is peppered with tiny patches of short yellow bristles which make its identification easy (Figure 1). This weevil cannot fly, but it can crawl and climb great distances. It is nocturnal, so if you have them nibbling your plants you may only be able to find them if you examine your plants at night. Although these adults can cause some damage to your plants, the main danger is from the larvae.

Weevil eggs hatch in the summer, and the larvae begin their lives by gnawing on plant roots. The larvae are approximately 1.5 cm long (Figure 2), and are generalist pests capable of living on many different plants. They relish *Sarracenia* and *Darlingtonia*. As they mature they travel towards the rhizome, and the real dining begins. They chew their way deeply into the rhizomes, and the damage from just 1-3 larvae can kill a plant. Even if the larvae are removed, the damaged rhizome may succumb to rot. The larvae

overwinter as pupae, and emerge as adults in the spring. The adults forage for food and lay eggs all summer long.

Kevin Snively (who first informed me about this pest) has told me the first symptom of an infestation is the feeling that the rhizome has become loose in the pot (because of the loss of roots). Furthermore, even though the planting mix is moist, the plant may wilt in hot conditions—this is because the damaged root system cannot supply the plant with sufficient water.

If you discover weevil larvae, the best thing to do is isolate the infested plants, then clean them thoroughly. This involves lifting the plants out of the soil, washing the roots, and cutting out infested rhizome sections. Dig the larvae out of the cavities that they have eaten into the rhizomes. You may wish to dust the cuts with sulphur fungicide. Look for larvae in your discarded soil and wash-water—kill all that you find. Repot the plant in fresh mix. This draconian treatment will certainly result in the death of some of your plants, but untreated plants are nearly certainly doomed. (It is particularly frustrating that autumn searches for weevil larvae traumatize the root



Figure 1: *Otiorhynchus sulcatus* weevils.



Figure 2: *Otiorhynchus sulcatus* larvae.

system precisely when pitcher plants should be left undisturbed.)

Black vine weevil has been reported on *Sarracenia* on both the east and west coasts of the USA, as well as in England. This pest seems to prefer coastal areas, but everyone growing *Sarracenia* or *Darlingtonia* should be vigilant.

I contacted entomologist TunyaLee Martin (University of California at Davis), who is studying the black vine weevil's biology. Ms. Martin told me that while a nematode bio-control exists, it is not the instant-kill, silver bullet that carnivorous plant enthusiasts would consider acceptable. (In the time it takes to affect the larvae, significant *Sarracenia* or *Darlingtonia* fatality could occur.) A pesticide like Dursban or Orthene can be effective if it comes in contact with the larvae, but a simple soil drench would probably not work well since the larvae (ensconced in rhizome cavities) are effectively shielded. These pesticides might be more effective if mixed with the soil as a preventative measure. Find out from your local supplier of pesticides if you can buy the appropriate pesticide in a granular form for this strategy.

Since biocontrols and pesticides have such major problems and mechanical treatment (i.e., uprooting and cleaning the plants) is so taxing on the plants, preventing infestations is the best policy. Since the weevils cannot fly and can only crawl, it would be wise to devise physical barriers the weevils cannot defeat. If your pots are in trays on tables, setting the table legs in cups of soap solution may discourage prowling adults. Antifreeze (ethylene glycol) is certainly effective, but since wildlife and pets are attracted to it, drink it, and are poisoned, it should be used with great caution. Plastic trays with steep walls may not provide sufficient protection—black vine weevils are incredible climbers. Even if the trays are difficult to climb when clean, the weevils may be able to scale them if they are even a little dirty.

Be proactive in your preventative measures. Watch for overhanging vegetation that weevils may drop from—they are particularly fond of *Euonymus* bushes. If you are having trouble with weevils, separate all the infested plants. Finally, keep your plants tidy. Pitchers flopping over to other pots or to the ground can allow weevils to move from plant to plant. Experiment. And good luck!

Acknowledgments:

I wish to thank Ms. Martin for useful discussions, and for providing live specimens to pose for my camera.

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Species/Hybrid Name	Native Area, Description Leaf Span in Inches, Highland/Lowland	US \$
<i>Brocchinia reducta</i>	Guyana Highlands, S.A. (5" tall pitchers) L	30
<i>Cephalotus follicularis</i>	Australia (1.5") (3 plants) cool L	10
<i>Heliamphora ionasi</i>	Ilu Tepui., Venezuela (1.5") H	45
<i>H. minor</i>	AuyanTepui, Venezuela (4") H	80
<i>H. tatei</i>	C. Huachamachari, Venezuela (2") H	55
<i>N. alata</i>	Palawan, Philippines (5") L	15
<i>N. albo-marginata</i>	Penang, Malaysia (all red, 5") L	35
<i>N. ampullaria</i>	Sarawak, Malaysia (green, 3") L	15
<i>N. bicalcarata</i>	Brunei, Borneo (3') L	20
<i>N. bongso</i>	Indonesia (4") H	50
<i>N. burbridgeae</i>	G. Kinabalu, Sabah (4") H	55
<i>N. burkei</i>	Philippines (4") L	25
<i>N. danseri</i>	Waigeo Island, Irian Jaya (3") L	40
<i>N. distillatoria</i>	Sri Lanka (yellow/green, 5") L	10
<i>N. ephippiata</i>	G. Raya, Borneo (4") H	40
<i>N. eymae</i>	G. Lumut, Sulawesi (3") H	50
<i>N. faizaliana</i>	Borneo (3") L	35
<i>N. fusca</i>	Sabah (3") L	15
<i>N. gracilis</i>	Talangka Rajah, Borneo (3") L	15
<i>N. gracillima</i>	Genting Highlands, Malaysia (3") H	35
<i>N. gymnamphora</i>	G. Singgalang, Sumatra (3") H	30
<i>N. hirsuta</i>	Sarawak, Malaysia (3") H/L	25
<i>N. lamii</i>	Irian Jaya (3") H	50
<i>N. lowii</i>	G. Trusmadi, Sabah (3") H	40
<i>N. macfarlanei</i>	Genting Highlands, Malaysia (3") H	25
<i>N. macrophylla</i>	G. Trusmadi, Borneo (2") H/H	70
<i>N. madagascariensis</i>	Madagascar (3") L	7
<i>N. maesolensis</i>	Madagascar (2") L	30
<i>N. maxima</i>	Rantepao, Sulawesi (3") L	30

Species/Hybrid Name	Native Area, Description Leaf Span in Inches Highland/Lowland	US \$
<i>N. maxima</i>	Indonesia (3")	35
<i>N. merrilliana</i>	Mindanao, Philippines (3")	10
<i>N. muluensis</i>	G. Mulu, Sarawak (3")	35
<i>N. murudensis</i>	G. Murud, 7000 ft. (3")	35
<i>N. neogunieensis</i>	Angkasa, Irian Jaya (3") L	45
<i>N. northiana</i>	Bau, Sarawak (3") L	30
<i>N. pilosa</i>	G. Batu Buli, Sarawak (2") H	30
<i>N. rafflesiana</i>	Bau, Sarawak (giant form, 3") L	30
<i>N. rafflesiana</i>	Brunei (typical, 3")	12
<i>N. rafflesiana</i>	Brunei (elongata, 3") L	30
<i>N. rafflesiana</i>	Brunei (gigantea, 3") L	30
<i>N. rajah</i>	G. Kinabalu, Sabah (2") H/H	25
<i>N. reinwardtiana</i>	Borneo (green trap, 3") L	20
<i>N. reinwardtiana</i>	Telupid, Sarawak (red trap, 3") L	30
<i>N. reinwardtiana</i>	G. Murud, 6900 ft. (2") L	35
<i>N. sanguinea</i>	Genting Highlands, Malaysia (4") H/L	15
<i>N. sibuyanensis</i>	Sibuyan, Philippines (4") L	30
<i>N. spathulata</i>	Sumatra (4") H	40
<i>N. stenophylla</i>	Sarawak (4") H	45
<i>N. sumatrana</i>	Sibolga, Sumatra (4") L	30
<i>N. talangensis</i>	G. Talang, Sumatra (4") H	35
<i>N. tentaculata</i>	G. Murud (4") H	30
<i>N. thorelli</i>	Phuk Radung, E. Thailand (3") L	20
<i>N. tobaica</i>	Sumatra (red pitcher) (4") H/L	35
<i>N. treubiana</i>	Sibolga, Sumatra (4") L	35
<i>N. truncata</i>	Philippines (4") L	35
<i>N. veitchii</i> (highland)	Batu Lawi, Sarawak (4") L	25
<i>N. veitchii</i> (lowland)	Sungai Samba, Borneo (4") L	25
<i>N. ventricosa</i>	Philippines (4") L	10
<i>N. vieillardii</i>	New Caledonia (3") L	25
<i>N. villosa</i>	G. Kinabalu, Sabah (2") H/H	50

GOOD NEWS: *DROSERA LONGIFOLIA* L. REJECTED, *SARRACENIA PURPUREA* L. CONSERVED WITH A NEW TYPE

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Keywords: taxonomy: *Drosera anglica*, *Drosera intermedia*, *Sarracenia purpurea*.

Drosera longifolia L. versus
Drosera intermedia Hayne and *Drosera anglica* Hudson

The Committee for Spermatophyta (see for example Brummitt, 1999) has voted by 11: 0 to reject the name *Drosera longifolia* L. (Brummitt *pers. comm.*), as proposed by Cheek (Cheek, 1998). This is good news for those who wish to keep applying the names *Drosera anglica* Huds. and *Drosera intermedia* Hayne in the sense in which they have been used in recent decades in most books on carnivorous plants (e.g. Schnell, 1976; Slack, 1979) and in most Floras (e.g. Tutin *et al.*, 1993). These two broadly similar circumboreal species were treated by Linnaeus as one species, *Drosera longifolia*, in 1753 and were not separated until some years later. Plant nomenclature is ruled by the law of priority so *Drosera longifolia*, being published earlier, has priority over both *Drosera intermedia* and *Drosera anglica*. This has caused confusion over nearly two centuries. Eventually an informal consensus emerged among botanists to ignore the name *Drosera longifolia*, as reflected in national and regional flora accounts. The exception has been in French Floras where, in the main, *Drosera longifolia* has been preferred to *Drosera anglica*. A few years ago I was alerted by Don Schnell to the danger that *Drosera longifolia* L. was edging towards common currency again in various European works. Fortunately the International Code of Botanical Nomenclature now allows the possibility of protecting against name changes of well-known species brought about by observing the principle of priority. Protection is achieved by publishing a case for the protection of the existing usage of a name in Taxon, the Journal of the International Association of Plant Taxonomists, and awaiting the judgement of the Committee of Spermatophyta. This judgement has now been made: only eight votes in favour from the committee are required and eleven have cast in favour. *Drosera longifolia* L. can now be officially and eternally ignored as a threat to *D. anglica* Huds. and *D. intermedia* Hayne. We can all afford a sigh of relief.

Sarracenia purpurea L. subsp. *purpurea* is the northern subspecies or the southern?

Of course, the northern subspecies of *Sarracenia purpurea* is *S. purpurea* subsp. *purpurea*, and the southern one *Sarracenia purpurea* subsp. *venosa* (Raf.) Wherry (e.g. Schnell, 1976; Slack, 1979). However, as pointed out by Reveal (1993) this application of names is contrary to the Code. Because McDaniell (1976) lectotypified *Sarracenia purpurea* on a southern element instead of a northern one, the southern subspecies and not the northern should be called *S. purpurea* subsp. *purpurea*. See Cheek (1994) for full explanation. In order to oppose Reveal's reve-

lation and to maintain existing usage, another proposal was prepared for Taxon, this time to conserve *Sarracenia purpurea* with a new type of the northern subspecies, expressly so as to maintain the existing application of subspecific names in *Sarracenia purpurea* (Cheek *et al.*, 1997). The Committee for Spermatophyta has now adjudicated in favour of this proposal, again by voting 11: 0 (Brummitt *pers. comm.*). Thus, the name *Sarracenia purpurea* subsp. *purpurea* can, despite McDaniel's lectotypification, still be used in the sense that it has been in the last 20-30 years, which is for the northern, and not the southern subspecies.

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NEW CULTIVARS REGISTERED IN 2000

Name	Established by, in	Registered on
<i>Sarracenia</i> 'Abandoned Hope'	P. D'Amato, Carniv. Pl. Newslett. 29:46 (2000)	4. 7. 2000
<i>Sarracenia</i> 'Imhotep'	B. Meyers-Rice, Carniv. Pl. Newslett. 29:14 (2000)	30. 3. 2000
<i>Sarracenia</i> 'Lamentations'	P. D'Amato, Carniv. Pl. Newslett. 29: 47 (2000)	4. 7. 2000
<i>Sarracenia</i> 'Spatter Pattern'	P. D'Amato, Carniv. Pl. Newslett. 29: 47 (2000)	4. 7. 2000
<i>Dionaea</i> Dentate Traps Group	B. Meyers-Rice, Carniv. Pl. Newslett. 29:16 (2000)	30. 3. 2000
<i>Dionaea</i> 'Dentate Traps'	B. Meyers-Rice, Carniv. Pl. Newslett. 29:16 (2000)	30. 3. 2000
<i>Dionaea</i> 'Sawtooth'	B. Meyers-Rice, Carniv. Pl. Newslett. 29:16 (2000)	30. 3. 2000
<i>Nepenthes</i> 'Bruce Bednar'	A. Marshall, Carniv. Pl. Newslett. 29:87 (2000)	1. 10. 2000
<i>Nepenthes</i> 'Frau Anna Babi'	P. D'Amato, Savage Garden: 260 (1998)	1. 10. 2000
<i>Nepenthes</i> 'Île de France'	Hort.M.Lecoufle ex Kusakabe, Carniv. Pl. Newslett. 12:6 (1983)	14. 1. 2000
<i>Nepenthes</i> 'Marie'	P. D'Amato, Savage Garden: 260 (1998)	1. 10. 2000
<i>Nepenthes</i> 'Nora'	P. D'Amato, Savage Garden: 260 (1998)	1. 10. 2000
<i>Pinguicula</i> 'Hanka'	M. Studnicka, Carniv. Pl. Newslett. 29:46 (2000)	4. 7. 2000
<i>Pinguicula</i> 'Pirouette'	J. Brittnacher, B. Meyers-Rice & L. Song, Carniv. Pl. Newslett. 29:17 (2000)	30. 3. 2000
<i>Utricularia</i> 'Cthulhu'	B. Meyers-Rice, Carniv. Pl. Newslett. 29:20 (2000)	30. 3. 2000
<i>Utricularia</i> 'Lavinia Whateley'	B. Meyers-Rice, Carniv. Pl. Newslett. 29:20 (2000)	30. 3. 2000
<i>Utricularia</i> 'Mrs. Marsh'	B. Meyers-Rice, Carniv. Pl. Newslett. 29:20 (2000)	30. 3. 2000
<i>Utricularia</i> 'Yog-Sothoth'	B. Meyers-Rice, Carniv. Pl. Newslett. 29:20 (2000)	30. 3. 2000

INSTRUCTIONS TO AUTHORS

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