PRINCIPES

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PRINCIPES

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A staminate plant of *Chamaedorea stolonifera*. Photo by G. Hampfler, Longwood Gardens, courtesy of W. H. Hodge. See pp. 26–28.

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Palm Seed Storage and Germination Studies

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ABSTRACT

A series of experiments was performed to evaluate the effects of seed maturity, seed cleaning, and gibberellic acid (GA_3) or water presoaking on the percentage and speed of germination of *Chrysalidocarpus lutescens*, *Syagrus romanzoffiana*, *Phoenix roebelenii*, and *Roystonea regia* seed. Effects of temperature, cleaning, and storage container on the viability of stored *C. lutescens* seed were determined in another set of experiments. Most palm seed germinated rapidly and consistently when half-ripe to ripe seed was maintained at temperatures between 30° and 35° C. Cleaning seed is not essential if planting is done immediately for *C. lutescens*, but cleaning seed of the other 3 species enhanced germination. Presoaking seeds in 1,000 ppm GA_3 for 48 hr slightly accelerated germination speed, but caused excessive elongation of the resulting seedlings and was therefore not recommended. The best method for long term storage of palm seed was to clean half-ripe or ripe seed, air dry at 80 to 90% relative humidity, treat with a seed protectant fungicide, and store at 23° C in tightly sealed polyethylene containers. Optimum planting depth was dependent on the drying potential of the germination site.

The primary method of propagating most palms is by seed, although tissue culture techniques have made it possible to propagate asexually a few important palm species (Tisserat 1979, Startisky 1970). Germination of palm seeds can require from several weeks to over a year (McCurrach 1960, Basu and Mukherjee 1972) and methods of accelerating palm seed germination are being sought. Presoaking seeds in gibberellic acid (GA_3) has been shown to accelerate germination of *Ptychosperma macarthuri* (H. A. Wendl.) Nichols, *Archontophoenix alexandrae* (F. J. Muell.) H. A. Wendl. and Drude, and *Chrysalidocarpus lutescens* H. A. Wendl. (Nagao and Sakai 1979, Nagao et al. 1980, Schmidt and Rauch 1982), but preliminary studies by the authors indicated that GA_3 presoaking can cause excessive elongation of seedlings.

Maintaining relatively high germination temperatures (e.g. 27° C) is known to promote seed germination of *Chamaedorea elegans* Mart., *Elaeis guineensis* Jacq. and *P. macarthurii* (Poole et al. 1975, Nagao et al. 1980). Other factors such as seed maturity, pericarp removal (cleaning), and planting depth have not been critically investigated, however.

Palm seed is generally considered to be short-lived and often loses its viability after 2 weeks to 3 months of storage (DeLeon 1958). Effective seed storage methods have been determined for *E. guineensis*, but little is known about the storage of other palm species (Rees 1963). The purpose of this study was to determine the effects of various factors on seed storability and germination in several species of ornamental palms.

Materials and Methods

Freshly harvested fruit of *C. lutescens*, *Syagrus romanzoffiana* (Cham.) Becc., *Roystonea regia* (Kunth) O. F. Cook, and *Phoenix roebelenii* O'Brien was sorted into green, half-ripe, and ripe categories for determination of fruit maturity effects as well as interactive effects of seed cleaning and presoaking on speed of germination and germination percentage. Green fruit was hard, full-sized, and was collected from infructescences containing some ripening fruits. Half-ripe fruits were semi-hard and slightly green in color, while ripe fruits were soft and had the normal ripe fruit color for each species. Half the fruit in each ripeness category were cleaned by manually removing the pericarp from the seed and half were left uncleaned. A 3 × 2 × 2 factorial experiment involving 3 ripeness categories, cleaned vs. uncleaned seed, and water vs. GA_3 presoaks was set up using 3 replicate lots of 50 seeds each per treatment. Seeds were soaked in GA_3 at 1,000 ppm or deionized water for 48 hr. Following presoaking, seed was sown in 10 cm square containers filled with a Canadian peat and perlite (1:1, by vol.) medium. The containers were placed in a seed germination room in which temperatures of 30–35° C were maintained. Green *P. roebelenii* seeds were not cleaned due to the difficulty of this operation and no *R. regia* seeds were water-soaked due to a shortage of seed. Number of emerging shoots per container was recorded weekly and final germination percentage and time required for 50% of final germination percentage rate were determined from this. Data from this experiment was subjected to analysis of variance.

Optimum germination temperature for *C. lutescens* seed was determined by planting fresh cleaned seed as above and placing the containers in growth chambers set at constant temperatures of 15, 20, 25, 30, 35, or 40° C. Presoak treatments consisting of 1,000 ppm GA_3 for 48 hr, deionized water for 48 hr, or no presoak were applied to 3 replicate lots of 50 seeds each for each temperature. Emerging seedlings were counted weekly and the data used to calculate final germination and time required for 50% of final germination percentage.

Effects of storage temperature on germination of *C. lutescens* seed were determined by storing lots of 50 seeds each in sealed polyethylene bags placed in chambers maintained at 0, 5, 10, 15, and 23° C. Fresh cleaned ripe seed was air-dried at 80–90% relative humidity and dusted with thiram (a seed protectant fungicide) prior to storage. Three replicate lots of 50 seeds each were removed monthly (hourly for 0° C and every 8 hr for 5° C stored seed) from each storage chamber and planted as in the seed maturity experiment. Number of seeds germinating each week was counted for each treatment and storage was continued for a maximum of 600 days. Seed storage was discontinued for a given temperature 4 months after such seed ceased germinating. Similar lots of *P. roebelenii*, *R. regia*, and *S. romanzoffiana* seed were stored only at 23° C since preliminary studies showed this to be the optimum temperature for these species.

Cleaned and uncleaned *C. lutescens* seeds were air-dried at 80–90% relative humidity and treated with thiram prior to storage to determine if dry or humid storage was best. Seeds were stored in lots of 50 in either paper bags (permeable to moisture) or sealed polyethylene bags (impervious to water) at 23° C. Three replicate lots of 50 seeds each were removed from each storage treatment every month and planted as in the seed maturity experiment. Number of seeds germinating weekly was recorded and final germination percentage was calculated. Data from germination temperature and seed storage experiments were analyzed by analysis of covariance.

Interaction of planting depth and germination environment on germination time and percentage was determined by performing an additional experiment involving direct seeding of cleaned *C. lutescens* seed in 3-liter containers. One hundred fresh cleaned seeds were planted in each of 10 replicate containers per treatment using a well drained potting medium. Treatments consisted of factorial combinations of various planting depths (surface planting, barely covered, or 1, 2, 4, or 6 cm deep) and germination environments (full sun, full sun but containers covered with 2 layers of cheesecloth, or shadehouse having 63% shade). Number of seedlings was counted weekly for each container and from this, germination rate and time were calculated.

Table 1. Effects of seed maturity, seed cleaning, and GA_3 presoaking on Chrysalidocarpus *seed germination percentage and time.*

Fruit Maturity	Cleaned	Presoak	Final Germ. %	Germination Time (Days)
Green	yes	H_2O	28.0	61.3
Green	no	H_2O	74.7	58.0
Green	yes	GA_3	4.7	45.3
Green	no	GA_3	64.7	53.0
Half-ripe	yes	H_2O	86.7	43.0
Half-ripe	no	H_2O	88.7	57.0
Half-ripe	yes	GA_3	84.0	33.7
Half-ripe	no	GA_3	72.0	46.3
Ripe	yes	H_2O	86.0	40.0
Ripe	no	H_2O	82.7	53.0
Ripe	yes	GA_3	83.3	30.3
Ripe	no	GA_3	76.0	50.3
Significant Effects				
Fruit maturity			**z	***
Cleaning			***	***
Presoak			**	***
Maturity × Presoak			NS	NS
Maturity × Cleaning			***	***
Presoak × Cleaning			NS	*
Maturity × Presoak × Cleaning			NS	NS

z NS, *, **, and *** indicate not significant, or significant at 5%, 1%, or 0.1% levels, respectively.

Table 2. Effects of fruit maturity, seed cleaning, and presoaking on Syagrus romanzoffiana *seed germination.*

Fruit Maturity	Presoak	Cleaned	Germination Time (Days)	Final Germ. (%)
Green	None	no	78.3	14.0
Green	None	yes	41.7	45.3
Green	H_2O	no	77.5	5.3
Green	H_2O	yes	33.7	57.3
Green	GA_3	no	87.7	2.7
Green	GA_3	yes	48.0	27.3
Half-ripe	None	no	81.3	4.7
Half-ripe	None	yes	75.3	38.7
Half-ripe	H_2O	no	—	0.0
Half-ripe	H_2O	yes	77.7	73.3
Half-ripe	GA_3	no	81.5	6.0
Half-ripe	GA_3	yes	99.0	10.0
Ripe	None	no	78.0	4.7
Ripe	None	yes	86.5	20.0
Ripe	H_2O	no	85.0	1.3
Ripe	H_2O	yes	99.3	23.3
Ripe	GA_3	no	—	0.0
Ripe	GA_3	yes	102.7	27.3
Significant Effects				
Fruit Maturity			***z	NS
Presoak			NS	NS
Cleaning			NS	***
Maturity × Presoak			NS	NS
Maturity × Cleaning			***	NS
Presoak × Cleaning			NS	NS
Maturity × Presoak × Cleaning			NS	NS

z NS, and *** indicate not significant, or significant at 0.1% level, respectively.

Results and Discussion

Fruit Maturity. Fruit maturity, cleaning, and presoaking effects were highly significant in improving final germination percentage of *C. lutescens* seed (Table 1). Poorest germination occurred when seed from green fruit was cleaned and presoaked in GA_3, although germination was somewhat better if this seed was presoaked in water. The best germination occurred when half-ripe or fully ripe seed was used, but cleaning and presoaking showed no systematic effects within the half-ripe and fully ripe seed treatments.

Seed maturity, presoaking, and cleaning, as well as the seed maturity and cleaning interaction, all had highly significant effects on time required for germination. The presoak interaction was less effective, although still significant. Fastest germination occurred with cleaned fully ripe or half-ripe seed. GA_3 presoaking slightly accelerated germination within these treatments. With the exception of the green, water-soaked, cleaned treatment, the slowest germination occurred when seed was not cleaned.

Fruit maturity had a significant effect on time required for germination of *S. romanzoffiana* seeds, but did not affect final germination percentage (Table 2). Cleaned green seeds germinated more rap-

Table 3. Effects of fruit maturity, seed cleaning, and presoaking on Phoenix roebelenii *seed germination.*

Fruit Maturity	Presoak	Cleaned	Germination Time (Days)	Final Germ. (%)
Green	None	no	79.5	9.0
Green	H_2O	no	79.5	6.0
Green	GA_3	no	101.5	4.0
Half-ripe	None	no	82.0	48.0
Half-ripe	None	yes	50.5	59.0
Half-ripe	H_2O	no	66.0	58.0
Half-ripe	H_2O	yes	37.0	49.0
Half-ripe	GA_3	no	72.5	40.0
Half-ripe	GA_3	yes	55.0	59.0
Ripe	None	no	63.0	51.3
Ripe	None	yes	54.0	74.0
Ripe	H_2O	no	74.0	65.0
Ripe	H_2O	yes	43.7	77.3
Ripe	GA_3	no	67.3	67.3
Ripe	GA_3	yes	56.0	87.0
Significant Effects				
Fruit Maturity			***z	***
Presoak			*	NS
Cleaning			***	**
Maturity × Presoak			NS	NS
Maturity × Cleaning			NS	NS
Presoak × Cleaning			NS	NS
Maturity × Presoak × Cleaning			NS	NS

z *, **, and ***, and NS indicate significance at 5%, 1%, and 0.1% levels, or not significant, respectively.

Table 4. Effects of fruit maturity, seed cleaning, and presoaking on Roystonea regia *seed germination.*

Fruit Maturity	Presoak	Cleaned	Germination Time (Days)	Final Germ. (%)
Green	None	no	287.7	24.0
Green	None	yes	259.3	24.0
Green	GA_3	no	262.3	24.0
Green	GA_3	yes	276.7	20.7
Half-ripe	None	no	281.3	18.7
Half-ripe	None	yes	261.7	37.5
Half-ripe	GA_3	no	281.0	18.7
Half-ripe	GA_3	yes	263.7	46.7
Ripe	None	no	301.7	18.0
Ripe	None	yes	331.0	52.0
Ripe	GA_3	no	306.3	11.3
Ripe	GA_3	yes	297.7	28.7
Significant Effects				
Fruit Maturity			**z	**
Presoak			NS	**
Cleaning			NS	***
Maturity × Presoak			NS	***
Maturity × Cleaning			NS	***
Presoak × Cleaning			NS	NS
Maturity × Presoak × Cleaning			NS	**

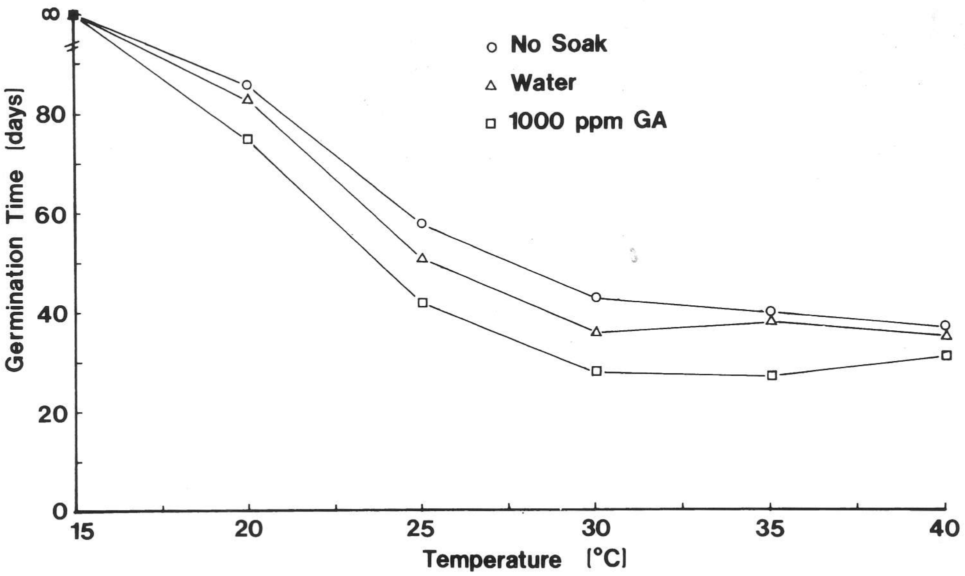
z NS, **, and *** indicate not significant or significance at 1%, and 0.1% levels, respectively.

idly than did half-ripe or ripe seeds, or uncleaned green seeds. Seed presoaking had no effect on germination or final germination percentage, but cleaning *S. romanzoffiana* seed greatly improved final germination percentage. The highest germination percentage was obtained when cleaned green or half-ripe seeds were used. The fact that ripe *S. romanzoffiana* seeds germinated more slowly and that very few uncleaned seeds germinated suggests the presence of a germination inhibitor in the pericarp of ripe fruit.

Fruit maturity, presoaking, and cleaning all had significant effects on time required for germination of *P. roebelenii* seeds, with cleaned ripe or half-ripe seed germinating the fastest (Table 3). Final germination percentage was also affected by fruit maturity, with cleaned ripe seeds germinating best.

Germination time was affected only by fruit maturity for *R. regia* (Table 4). Ripe seed generally germinated more slowly than half-ripe or green seed, but differences were slight. Final germination percentage was greatest for cleaned ripe seed which was not presoaked or cleaned half-ripe seed presoaked in GA_3. Poorest germination occurred in green or uncleaned ripe or half-ripe seed.

Acceleration of palm seed germination by GA_3 presoaks has been noted for *C. lutescens*, as well as *P. macarthurii* and *A. alexandrae* (Schmidt and Rauch 1982,

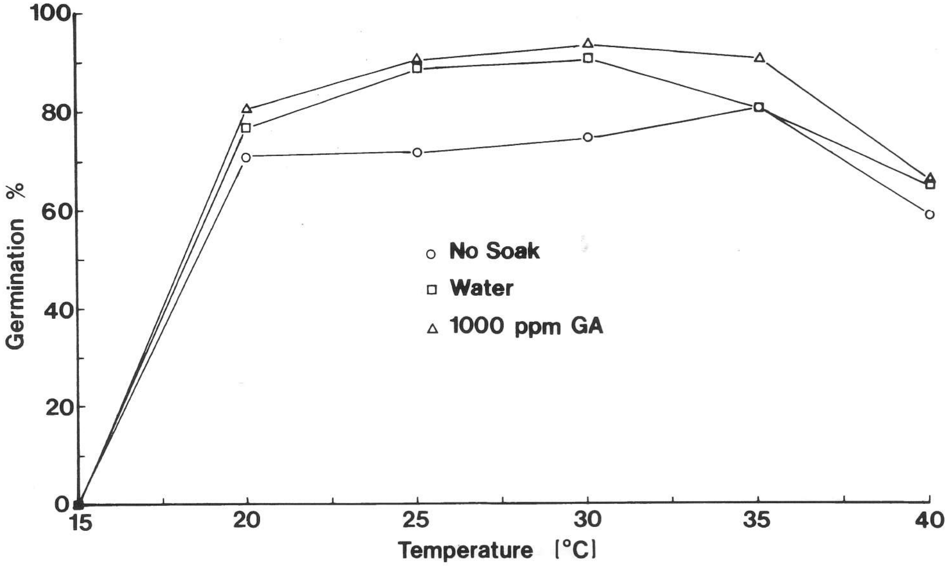


1. Effects of germination temperatures and presoaking seed on germination time for *Chrysalidocarpus lutescens* seed. Points represent means for treatments.

Nagao and Sakai 1979, Nagao et al. 1980) however, the effects GA_3 seed presoaking had on seedling morphology were not mentioned. Seedlings of *C. lutescens*, *S. romanzoffiana*, and *R. regia* from GA_3-soaked seeds elongated much more rapidly than palms from water-soaked seeds, resulting in tall, weak, unattractive plants. Seedlings of *P. roebelenii* from GA_3-soaked seeds were twisted and assumed a corkscrew-like appearance. Seedlings from these experiments were grown for a year after germination and GA_3 effects on shoot elongation were still apparent at that time. Since compact plants are usually stronger and more attractive, the slight decrease in germination time for GA_3 treated seeds would be more than offset by the inferior quality of the resulting plants.

Germination Temperature. Germination temperature had a highly significant effect on germination time of *C. lutescens*. Time decreased as temperature increased from 20 to 40° C, although differences between 30, 35, and 40° C were less than at lower temperatures (Fig. 1). Presoaking seeds in GA_3 decreased germination time significantly over water-presoaked and non-soaked seeds, and water-presoaked seed germinated faster than non-soaked seed. This is consistent with previous results obtained in other experiments (Schmidt and Rauch 1982, Nagao and Sakai 1979, Nagao et al. 1980). There was no significant interaction between germination temperature and presoak treatment.

Final germination percentage of *C. lutescens* increased significantly as germination temperature was increased from 20 to 25° C, but percent germination at 40° C was less than at slightly lower temperatures (Fig. 2). This could have been caused by desiccation of some of the barely covered seed at this higher temperature since those that did germinate did so more rapidly than at lower temperatures. No seed germinated at 15° C. Final germination percentage was not enhanced by GA_3 presoaking, but both water and GA_3 presoaking resulted in greater final germination percentages than non-presoaked seeds at temperatures of 25 to 30° C.

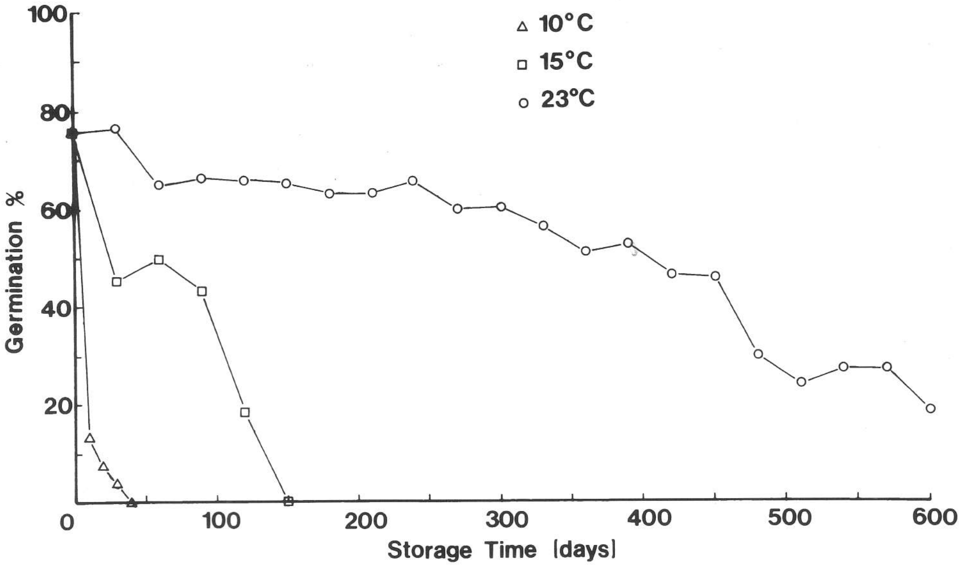


2. Effects of germination temperatures and presoaking seed on germination percentage of *Chrysalidocarpus lutescens* seed. Points represent means for treatments.

Seed Storage. Germination of cleaned *C. lutescens* seed stored in polyethylene bags was strongly influenced by storage temperature (Fig. 3). None of the seed stored at 0° C survived one hr, but seed stored at 5° C survived 8 hr with only a slight loss of germination (data not shown). No seed stored at 5° C germinated after 10 days and germination of seed stored at 10° C decreased from 13% at 10 days to 0% after 40 days (Fig. 3). Germination of seed stored at 15° C was reduced from nearly 50% at 30 days to 0% at 150 days, but germination percentage remained above 50% for 420 days with seed stored at 23° C. Nearly 20% of the seed germinated even after 600 days of storage at 23° C.

Germination percentage of *S. romanzoffiana* seed declined rapidly after 4 months of storage at 23° C (Fig. 4). *P. roebelenii* seed rapidly lost its viability after 8 months of storage, but germination of *R. regia* seeds stored for 9 months or less exceeded that of freshly planted seed. Approximately 1% of *R. regia* seeds planted immediately germinated within 6 weeks, but no additional germination occurred until about 8 months after harvest when most of the seeds germinated. Seed which had been stored in sealed polyethylene bags germinated 8 months after harvest, regardless of the storage time. This may be due to immature embryos in these seeds at harvest time (Hartmann and Kester 1983). Stored royal palm seed probably germinated better than seed planted immediately because seed stored in sealed polyethylene bags is less subject to desiccation than that which is planted and subjected to periodic partial drying.

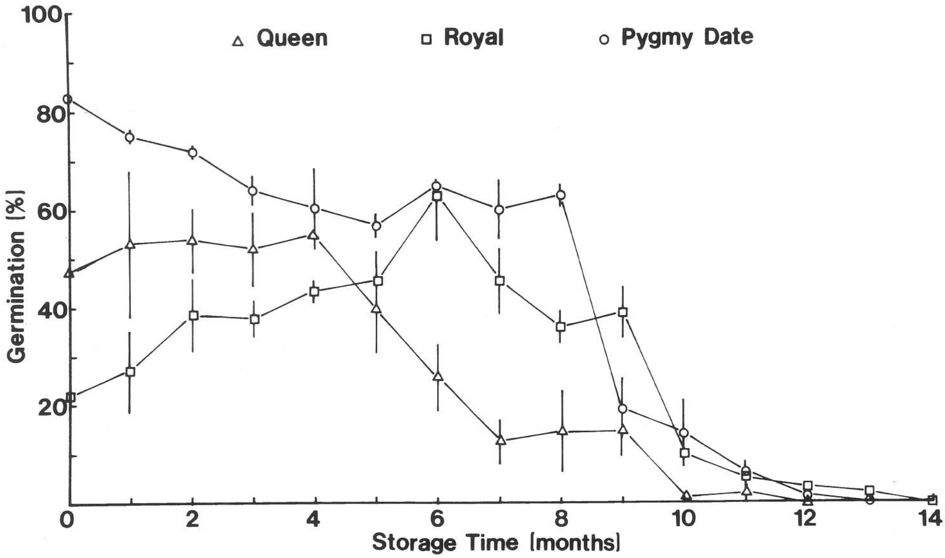
The method and preparation of seeds for storage also had a major influence on the germination of stored *C. lutescens* seed (Fig. 5). Less than 10% of the uncleaned ripe seed stored for 30 days at 23° C in polyethylene bags germinated, whereas nearly 80% of such seed germinated prior to storage. Germination of cleaned *C. lutescens* seed declined rapidly from 70% to almost 0% after 120 days when stored in porous paper bags, but cleaned seed



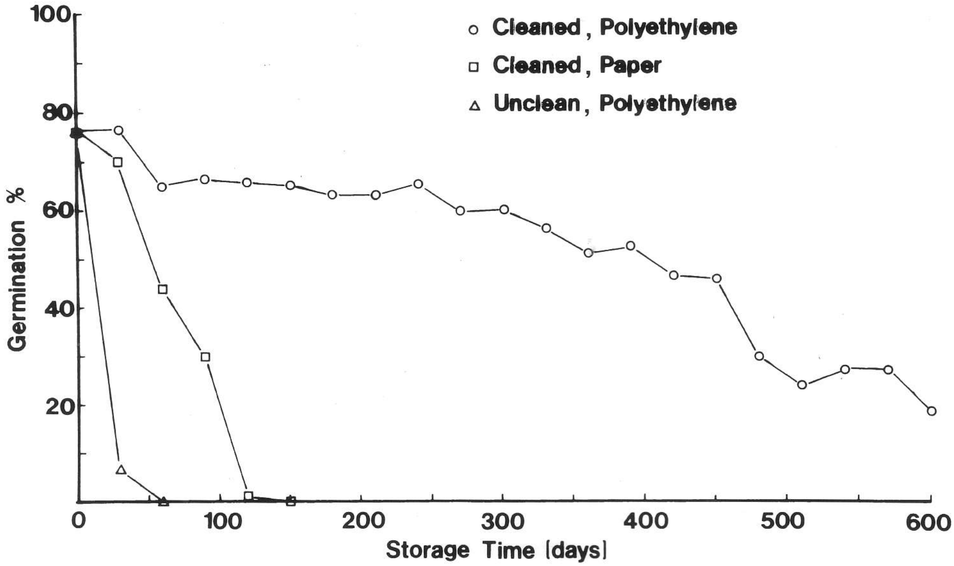
3. Effects of storage temperature and storage time on germination of stored cleaned *Chrysalidocarpus lutescens* seed. Points represent means for treatments.

stored in sealed polyethylene bags remained viable for over 600 days. Dissection of seed from known viable and non-viable lots showed that embryo desiccation was the main reason for non-viability among palm seeds.

The reason uncleaned seed stored so poorly is not known, but could involve the

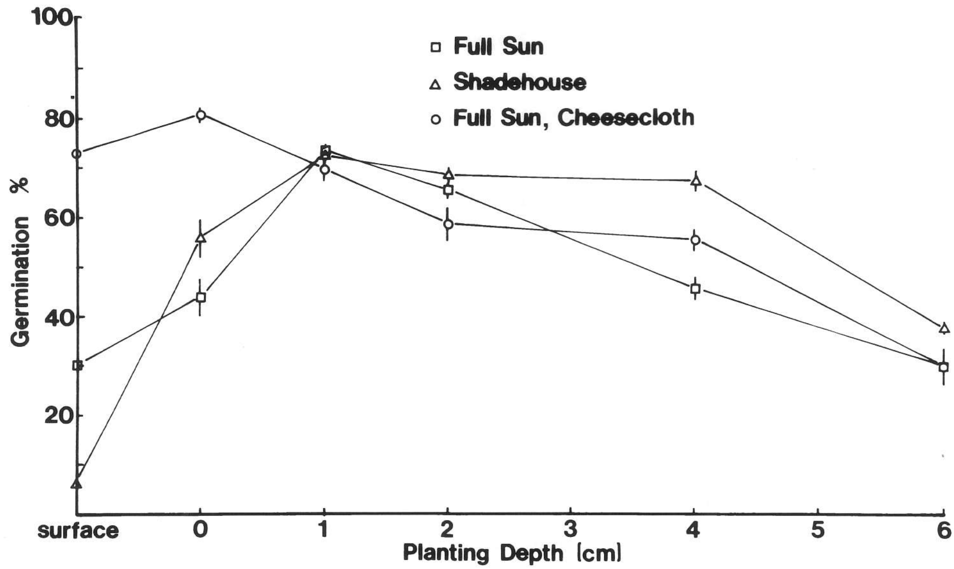


4. Effects of storage on germination of *Syagrus romanzoffiana*, *Phoenix roebelenii*, and *Roystonea regia*. Points represent means for treatments.

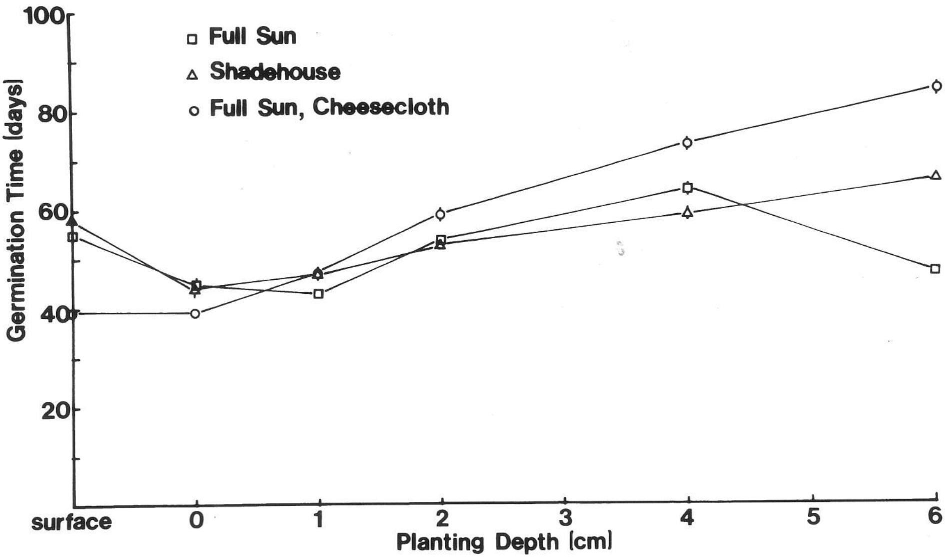


5. Effects of storage method and storage time on germination of stored *Chrysalidocarpus lutescens* seed stored at 23° C. Points represent means for treatments.

formation or release of germination inhibitors in overripe fruit. Anaerobic decomposition in sealed polyethylene bags is probably not involved since similar uncleaned seed stored in paper bags in preliminary studies (data not shown) germinated as poorly as those sealed in the polyethylene bags.



6. Effects of planting depth and germination environment on germination percentage of *Chrysalidocarpus lutescens* seed. Points represent means for treatments.



7. Effects of planting depth and germination environment on germination time of *Chrysalidocarpus lutescens* seed. Points represent means for treatments.

Planting Depth. Germination percentage and germination time were greatly affected by planting depth (Figs. 6,7). Final germination percentage for *C. lutescens* seeds germinated in full sun ranged from nearly 74% for seeds planted 1 cm deep to about 30% for seeds planted on the surface or 6 cm deep. Similarly, germination time in full sun decreased from 67 days for the seeds planted 6 cm deep to about 44 days for those covered with only 1 cm of medium. However, palms from seed covered with 2 cm or more of medium were more susceptible to iron deficiency symptoms when grown for 1 yr than those planted less than 2 cm deep (1). Optimum planting depth for *C. lutescens* seeds germinated in full sun but covered with cheesecloth occurred when seeds were barely covered, both with respect to final germination percentage and speed of germination. Excellent germination percentage under shadehouse conditions occurred with seeds covered with 1, 2, or 4 cm of medium, but seeds barely covered germinated faster. Thus, as the drying potential of the germination site increases from that of cheesecloth-covered containers through shadehouse conditions, and finally to exposed full sun conditions, optimum planting depth increases accordingly from barely covered for sites with low drying potential to 1 cm deep for sunny drier sites.

Conclusions. These studies show that many factors can affect palm seed storability and germination. Best *C. lutescens* seed germination occurs if half-ripe or fully ripe seed is used. Removal of the fleshy pericarp is not essential if the seed is planted immediately, but if it is to be stored for more than a few days, removal is essential. Optimum germination percentage for *S. romanzoffiana* seed occurred when cleaned green or half-ripe seed is used. Cleaned ripe or half-ripe seeds germinated best for *P. roebelenii* and *R. regia*.

Presoaking seed in 1,000 ppm GA_3 for 48 hr slightly decreased germination time, but also caused excessive and undesirable elongation of the seedlings. Presoaking for 48 hr in water alone had a lesser but still significant effect on germination time and is preferable to GA_3 presoaking, since water did not cause distortion in the growth of the seedlings. If green *C. lutescens* seed must be used, it should not be cleaned.

Optimum germination temperatures for *C. lutescens* seed were determined to be between 30 and 35° C. Since embryo desiccation is a major cause for seeds not germinating, seed germinated in full sun or similar drying conditions should be covered with 1 cm of medium, while optimum planting depth for *C. lutescens* seed under less drying conditions is barely covered.

C. lutescens seed can be successfully stored for a year or more if fresh ripe seed is cleaned, air-dried at 80–90% relative humidity, treated with a seed protectant fungicide, sealed in polyethylene bags, and stored at temperatures of approximately 23° C. Using similar procedures, seed of *S. romanzoffiana* can be stored for up to 4 months, *P. roebelenii* for up to 8 months, and *R. regia* for about 9 months.

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Conservation-Conscious Collecting: Concerns and Guidelines

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For those of us from the temperate zone, our knowledge and appreciation of palms is based almost exclusively on cultivated species. Perhaps we grew up with potted plants in our homes, or admired potted palms in bank lobbies and airport terminals, or visited botanical gardens. Whatever the circumstance, most of us would have no firsthand knowledge of the splendor and unique habits of palms were it not for their cultivation. The number of palm species in cultivation is continually increasing, as both professional and amateur palm growers collect and distribute seed worldwide, and as new species are discovered in the wild. Yet, as we admire the precious seedlings and mature plants that we so fortunately obtain, how often do we consider these questions: Where did the plant or seed stock come from? Is this species threatened? What impact has seed collection had on natural populations? What other plant and animal species may be affected by seed collection? Did the country of origin benefit from permitting export of seed?

I raise these questions because I am deeply concerned about the future of all palm species, particularly those species that populate the world's vanishing tropical rain forests (Myers 1979, Caufield 1984). An estimated 75% of the world's palms live in tropical rain forests. Many of these species occur in low densities in rain forests that are shrinking in area every day. In Latin America and the Caribbean alone, a total of 53 palm species are now officially classified as endangered (Johnson 1987). Only those species whose ranges are within actively protected forest reserves, wildlife refuges, or parks are granted some degree of protection from the threat of extinction. Even these exceptions provide little consolation; in South America and Southeast Asia combined, less than 2% of the tropical rain forests are in reserves or parks (Myers 1979).

In response to the global crisis of tropical deforestation, the World Conservation Strategy adopted by the International Union for the Conservation of Nature and Natural Resources (IUCN) maintains that captive or cultivated populations and their propagation can and must be integral parts of the global strategies to preserve the biotic and genetic diversity of the planet. During the past decade there has been much emphasis on establishing germ-plasm banks and seed-storage facilities as a means of safeguarding against species extinction. These strategies have not proven very effective for palms, because most palm seeds are incapable of dormancy or long-term storage. For these reasons, botanical gardens, live germ-plasm banks (such as those for *Bactris gasipaes* in Costa Rica and Brazil), and horticultural nurseries are the only means of maintaining populations of palm species outside of their native habitats.

However, preservation of species is best accomplished by protection of their natural habitats. Botanical gardens can, at best, be living museums that maintain live individual plants rather than entire populations (Whitmore 1980). Protection of natural habitats contributes not only to the survival of thousands of otherwise threatened plant and animal species, but also preserves the important ecological interactions among these species (Futuyma 1983). Furthermore, the genetic diversity of species that is maintained in wild populations is often greatly diminished through cultivation (Frankel 1974). This genetic diversity is the basis for the evolutionary and adaptive potential within all species.

Recently, I have become concerned about the collection of rainforest palms, seeds, and foliage for horticultural purposes. I have received many requests to collect seed of palms that, although they would be highly desirable in cultivation, have not yet been grown outside of their natural forest habitat. I am a professional botanist, and have been studying the ecology and morphology of rainforest palms for the last six years (Chazdon 1986). All of my research has been conducted in either privately-held, nonprofit nature reserves or in national parks. In both cases, plant and seed collection is limited to research needs and requires administrative approval for each species and each project. For several years, I have been struggling with the apparent conflict between seed collection for private and commercial propagation and species protection in natural habitats. If few or no populations of these palms exist outside of nature reserves, how can seed be collected without negative consequences for the natural populations under protection? Unfortunately, we do not know enough about the population biology of most rainforest palm species to understand what the long-term impact of varying degrees of plant or seed collection would be. Furthermore, the conservation status of a large number of palm species is insufficiently known (Henrik Balslev and Rodrigo Bernal, personal communication).

One issue of particular concern is the widely held belief that seed collection (and the *ex situ* propagation of plants) ensures the protection of threatened palm species. Although there are cases where horticulture has played a significant role in the preservation of rare and endangered plant species (such as the maidenhair tree *Ginkgo biloba* L. and the cycad *Encephalartos woodii* Sander), *ex situ* propagation of species is appropriate only as a last-ditch strategy for conservation, when a species can no longer survive in the wild (Thompson 1975; Raven 1976; Myers 1979; Frankel and Soule 1981; Foose 1983). A high proportion of collections in botanical gardens are lost within a few years due to difficulties in propagation (Thompson 1975). The genetic diversity of cultivated species will deteriorate over time, especially if the original collections were restricted to a small number of individuals, and natural pollinators are absent. Yet, continuing evolution and adaptation to natural conditions, which provide the only real hope for long-term survival of species, are possible only within natural habitats (Frankel 1974). Without long-term ecological studies of natural populations of these species, successful reintroductions or reinforcements of naturally-occurring populations will be extremely unlikely (Frankel and Soule 1981). In the words of former IUCN Director General Gerardo Budowski (1976):

> Let us not forget, that conservation through living collections is *no* substitute to conservation in the wild. It is useful as a complement, in some cases a fire brigade approach, but under no circumstances should a breeding programme convey a soothing feeling of reassurance, for instance, 'now that we have good living collections, we need not worry anymore.'

Ideally, parallel efforts of both *in situ* and *ex situ* conservation should be advocated, because both approaches will contribute to species protection.

Collection and cultivation of palms can further the cause of species protection in many ways. Cultivated plants can be extremely valuable for educational, scientific, touristic, and esthetic purposes (not to mention commercial horticulture). Once in cultivation, one of the simplest, more effective, and least costly ways in which botanical gardens can ensure the perpetuation of particular species is to introduce them successfully into the horticultural trade, or to encourage their wide use in horticulture (Raven 1976). Making such plants widely available helps to relieve collecting pressure on wild populations and ensures the distribution of large numbers of plants without endangering wild populations. The knowledge and expertise of growers and private collectors in understanding the cultivation requirements of palms will further aid efforts to establish stock plants for seed sources of horticulturally desirable species.

It is clear, however, that to make lasting contributions to species preservation efforts, we need to develop mechanisms to channel at least a percentage of these benefits to the countries that so generously permit (knowingly or not) the collection and exportation of palms, palm seed, and palm foliage from their forests. We need to create links between species appreciation in developed countries and rain forest conservation efforts in the developing countries that contain wild plants and seed sources. Of the world's 458 botanical gardens, only 82 are located in developing countries (Myers 1979). And where does close to 100% of wild palm seed come from, but developing countries!

Whatever your interests in palm cultivation may be, the survival of many palm species in the wild may very well be affected by your activities. I encourage collectors, horticulturists, palm owners, and all those who appreciate palms for their many fine qualities to think seriously about the global crisis of tropical deforestation and species extinction. Without major conservation efforts, "palm hunting" as we know it will become a thing of the past. There *are* ways in which collecting, growing, selling, and buying palms can be conservation-oriented activities. One important way is to contribute (financially as well as other ways) to conservation of natural palm habitats through local and international conservation organizations, such as the Nature Conservancy International Program or the World Wildlife Fund. The World Wildlife Fund is currently involved in conservation projects specifically concerned with threatened palm species in Latin America (Sweeney 1985, Johnson 1987). Consider "adopting" a favorite palm species: find out where it grows naturally, and whether natural populations are protected in at least part of its range (this might be an appropriate project for a local Palm Society Chapter). Become an outspoken advocate for protection of this species and its habitat. Even better, plan a palm safari and visit the species in the wild. Take photographs, talk to local conservation groups, and show your pictures to other palm enthusiasts to rally support. Conservation efforts in developing countries are seriously hindered by lack of funds. Have a palm auction to raise money for protecting rainforest areas rich in palms (such as Braulio Carrillo National Park in Costa Rica). Through these actions, we can help to ensure a promising future for new generations of palms and palm-admirers.

Recently, a group of plant conservation advocates developed guidelines for conservation-conscious scientific plant collecting (Schaller 1987). Collecting guidelines for nursery people are also being developed (Dennis Johnson, personal communication). Below, I propose guidelines for professional and amateur palm collectors and growers that attempt to transform collecting into a conservation-conscious act of lasting benefit to both developed and developing countries. It is my sincere hope that through conservation-conscious collecting and other conservation-oriented activities, Palm Society members can help to reverse or at least slow down the overwhelming tide of extinction that threatens not only palms, but all the inhabitants of the earth.

Conservation Guidelines for Palm Collectors and Growers

1) Use of horticulturally-produced seed is greatly preferable to collection of wild seed.
2) Seed collection is greatly preferable to collection of established plants. Do not kill a plant for the sole purpose of collecting seed. When entire plants are needed, collect no more than one plant out of 20 per site per year.
3) Obtain proper collecting and exporting permits from the appropriate agency of the host country and importation permits from the home country. Obtain permission from the landowner before collecting and exporting seed or live plants.
4) Do not collect wild plants or seeds if you know or suspect that the species is locally or globally threatened. Become informed about the conservation status of palms in collecting sites.
5) Treat seeds with care and respect. Use all of the seed you collect, and distribute seed only to those who have appropriate germination and growing facilities. Do not collect immature seed.
6) Collecting a few seeds from several individuals is preferable to collecting many seeds from a single plant. Keep collections from each plant separate to avoid problems of mis-identification and/or hybridization.
7) Harvest seeds prudently. Collect only what you need, even if seeds are abundant. Avoid collecting repeatedly in the same sites.
8) If a species is rare, collect only a small portion of the fruit crop of any individual (no more than 10%). If only one fruiting individual is found in the vicinity, do not collect any seed.
9) Document your collections so that the location (including elevation), plant, and species identity can be known. If at all possible, make proper herbarium specimens for identification.
10) Whenever possible, use seed from wild palms to establish stock plants for seed distribution and breeding purposes, rather than for direct sale. Distribute seed to botanical gardens and educational institutions with adequate propagation facilities.
11) Use your knowledge of threatened palms or habitat destruction to publicize the urgency of protecting rain forest areas. Communicate with palm specialists, ecologists, and conservationists, and work together with them to identify threatened species and habitats.
12) Seeds are natural resources just like petroleum (a plant product) and precious metals. In exchange for the privilege of collecting and exporting seed, collectors should offer financial or other assistance to the host country. One appropriate action is to donate a percentage of all profits made from the sale of seed or plants to local or international conservation organizations.
13) Become involved with conservation organizations. These international organizations are directly involved with rain forest conservation efforts all over the world:
(1) The Nature Conservancy International, 1785 Massachusetts Avenue, N.W., Washington, D.C. 20036;
(2) World Wildlife Fund-U.S., 1601 Connecticut Avenue, N.W., Washington, D.C. 2009;
(3) Rainforest Action Network, 300 Broadway, Suite 28, San Francisco, CA 94133.

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Some Observations on Seed Germination, the Seedling, and Polyembryony in the Needle Palm Rhapidophyllum hystrix

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The needle palm, *Rhapidophyllum hystrix* (Pursh) H. A. Wendl. & Drude, is a rare shrubby, cespitose (or single-stemmed), mainly acaulescent, dioecious species belonging in the subfamily Coryphoideae (Dransfield and Uhl 1986). It is restricted to the coastal plain of the southeastern United States with a scattered distribution from central Florida north to Georgia and west to Mississippi (Shuey and Wunderlin 1977). This paper discusses aspects of seed germination, seedling morphology and development, and the occurrence of polyembryony in *R. hystrix*.

Several gross morphological features readily distinguish the needle palm from other sympatric palms such as *Sabal palmetto* Lodd. ex Schultes and *S. minor* (Jacq.) Persoon; namely the multi-ribbed leaf segments, the extremely reduced inflorescence (especially the female), and the spiny leaf bases (the spines may reach a length of 40 cm). Because of the short female inflorescence axis, mature fruits are consistently trapped among the spines and leaf bases and frequently these seeds germinate upon the parent plant with no possibility of surviving (Fig. 1). These seedlings eventually perish after exhausting the food stored in the endosperm of the seed. In addition, many seedlings can be found growing at the base of the parent plant (Fig. 2); these have little chance of surviving (pers. obs.), being outcompeted for light, nutrients, and moisture by the parent plant.

Methods

A large population of *Rhapidophyllum* in central Mississippi has been studied for several years. From this population seeds were collected in December 1984 and December 1985 and several germination experiments undertaken. Seedlings from the wild were transplanted into the greenhouse and their development monitored. Seedling morphology and development was described based on these seedlings.

Two separate treatments were employed, prior to the planting of *Rhapidophyllum* seeds, in the germination experiments. In the first treatment, 44 seeds, with pericarps removed, were placed in moistened peat moss in polyethylene bags and stored in a refrigerator at 3–5° C for 12 days (=stratification). After removal from cold storage the seeds were scarified (filing through the seed coat) until the endosperm was visible and then planted. The second treatment involved removal of the pericarp from 77 fruits followed immediately by planting. These seeds were neither stratified nor scarified. Seeds in which the pericarp only was removed (second treatment) were collected on 16 December 1984 and planted on 17 December 1984. They were monitored until the conclusion of the

1. *Rhapidophyllum hystrix* seedlings growing among the spines and leaf bases of parent plant. ×½.

2. *Rhapidophyllum hystrix* seedlings growing in the soil directly beneath the parent. ×½.

Table 1. Summary of germination experiments for Rhapidophyllum hystrix. *Treatments: pr, pericarp removed; sc, scarified; st, stratified. Both experiments were concluded on 1 June 1986.*

Date Planted	Treatment	Sample Size	Days to First Germination	#/% Germinated
1/01/86	(1) pr, sc, st	44	42	8/18.2
12/17/84	(2) pr	77	435	11/14.3

experiment on 1 June 1986 (530 days). Seeds collected in December 1985 were stratified from 20 December 1985 until 1 January 1986, at which time they were scarified and planted; this experiment (first treatment) was concluded 150 days later on 1 June 1986.

All seeds were placed, ca. 8–10 per 15 cm pot, in a 1:1:1 mixture of sterilized soil : perlite : shredded (sieved) pine bark and placed in a glass greenhouse (21° C during the day and 13–18° C at night). No bottom heat was provided.

Results and Discussion

Germination Experiments. In this study two germination treatments were tested in an attempt to determine conditions necessary for seed germination in *Rhapidophyllum.* Table 1 summarizes the results. The stratification-scarification treatment resulted in the earliest germination, in which the first seed germinated 42 days after planting, and by the conclusion of the experiment (150 days after sowing) 18% (8 of 44 seeds) had germinated. The seeds that were planted directly into the potting media after pericarp removal, but without the stratification and scarification pretreatments, required 435 days for the first seed to germinate. At the conclusion of the experiment (530 days from sowing) 14.3% (11 of 77 seeds) had germinated. Seeds of *Rhapidophyllum* have been reported to take from 6 months to 2 years to germinate (Popenoe 1973, Shuey and Wunderlin 1977, Wagner 1982). One treatment employed here resulted in a substantially faster germination time of only 6 weeks. Wagner (1982) planted 15 *Rhapidophyllum* seeds and only one (6.7%) of these had germinated after 195 days. The extended period usually required for *Rhapidophyllum* seeds to germinate may contribute to its reduced fecundity.

Seed Germination. As with most palms, seed germination in *Rhapidophyllum* is hypogeal. There are three basic variations in germination in palms: remote-ligular, remote-tubular, and adjacent-ligular (Tomlinson 1961, Moore and Uhl 1973). *Rhapidophyllum* illustrates the remote-ligular type. In this type of germination, a ligule extends from the distal end of the cotyledonary sheath. All subsequent leaves in *Rhapidophyllum* are also ligulate. In contrast, most other palms with ligulate cotyledons do not possess ligules on subsequent leaves (Tomlinson 1960*b*).

Figure 3 shows the morphology of a recently germinated *Rhapidophyllum* seedling. The elongated cotyledonary petiole and the ligule are quite distinctive. Figure 4 shows various stages of seedling development from emergence of the radicle at 2 days (a) to development of the eophyll at 8 weeks (d). In the remote-ligular type of germination the radicle breaks through the seed coat and, along with the cotyledonary sheath (which contains the hypocotyl) and the cotyledonary petiole, grows away from the seed. The cotyledonary petiole in *Rhapidophyllum* reaches a length of ca. 10 cm. Its growth is positively gravitropic which serves to thrust the young seedling into the soil. We would like to emphasize, however, that many of the seeds remain on the parent plant where the seedlings are thrust into the leaf bases and establishment is not possible (Fig. 1). After the cotyledonary petiole grows away from the seed the cotyledonary sheath splits allowing the plumule and successive leaves to grow to the soil surface. The plumule (which is white) and first eophylls (which are green) are all narrow linear leaves which facilitates their movement up through the soil (Tomlinson 1960*a*).

According to Tomlinson (1960*b*, 1961) remote-tubular and remote-ligular germination appear to be ecological adaptations of palms to xeric habitats. However, a substantial number of palms in mesic habitats exhibit remote-ligular or remote-tubular germination. Nevertheless, these sites are still susceptible to arid conditions, especially if the seeds are exposed to the air for extended periods of time as they are in *Rhapidophyllum*. Consequently, a type of germination which serves to physically bury the young seedling would be an ecological advantage.

Prior to germination (at least a month) the seed forms a haustorium which develops from the distal end of the cotyledon. The haustorium is characteristic of palm seeds in general. This structure has also been called a suctorial or suctional organ (Thiselton-Dyer 1910, Tomlinson 1960*a*) since it digests the endosperm and transfers this food reserve via vascular connections to the developing seedling. The importance of this source of nutrition for the seedling is indicated by the observation that *Rhapidophyllum* seedlings quickly died when severed from the seed and its endosperm. The haustorium is somewhat convoluted which increases its surface area and maximizes the hydrolytic and enzymatic breakdown of the endosperm.

The Seedling. The seedling stage in *Rhapidophyllum* is quite long. There is a gradual, prolonged transition from the seedling through the juvenile to the adult. We have arbitrarily defined the seedling stage as that period of time when the plant has simple, undivided leaves. The juvenile stage begins when a plant starts to develop palmate leaves with few to several segments, while the adult stage is defined by the onset of reproduction. According to Shuey and Wunderlin (1977), *Rhapido-*

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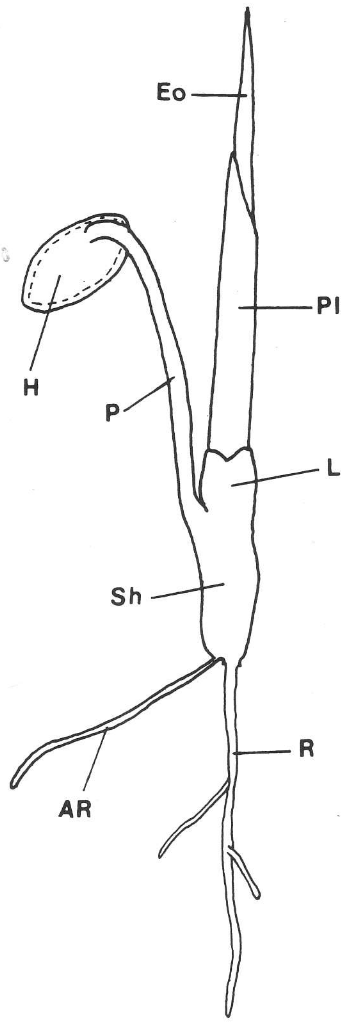
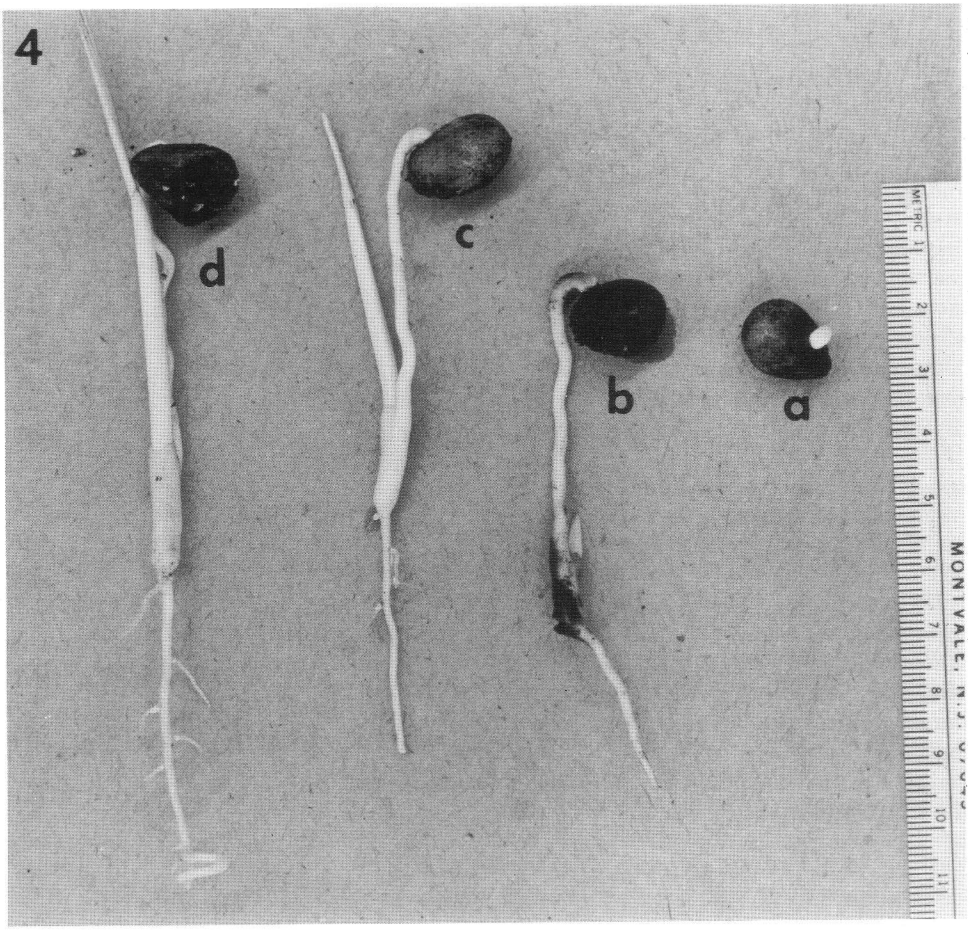


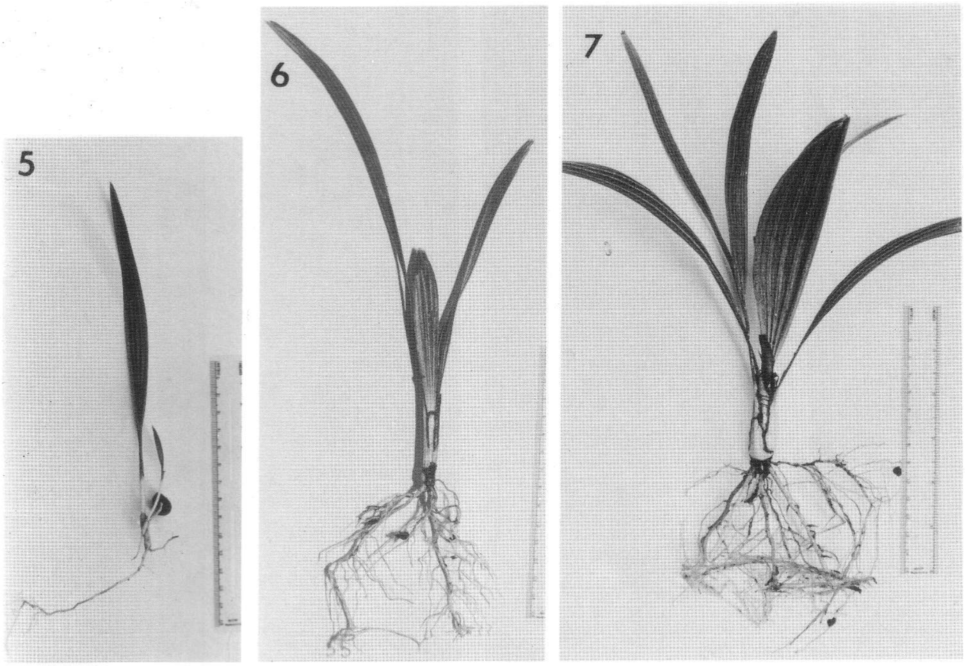
3. Drawing illustrating the morphology of a recently germinated seedling of *Rhapidophyllum hystrix*. ×1.8. Details: Eo, eophyll; Pl, plumule; L, ligule; P, cotyledonary petiole; H, haustorium within seed; Sh, cotyledonary sheath; R, primary root; AR, adventitious root.



4. Four stages of seedling development in *Rhapidophyllum hystrix.* (a) radicle just emerging at ca. 2 days, (b) seedling 2 weeks old, (c) seedling 4 weeks old, and (d) seedling 8 weeks old. Note the elongation of the cotyledonary petiole (arrow) which serves to thrust the growing seedling into the soil.

phyllum seedlings first begin to produce divided leaves about three years after germination. This time frame corresponds closely to our greenhouse-grown seedlings, which at 3½ years had begun to develop slightly palmate blades. On the average, seedlings produced 9 simple, linear leaves before the formation of the first divided leaf blade. The first palmate blades were found to have either two or three segments. The seedling in Figure 7 is estimated to be 3 years old and still has simple, undivided leaves. For purposes of comparison, Figure 5 is of a seedling 5 months old while Figure 6 is of a seedling 2 years old. Table 2 lists morphological characteristics for leaves from 2 year old seedlings. Although there was a wide range in leaf dimensions, the number of major veins per leaf was fairly constant, ranging from 3 to 5.

In the wild, no seedlings over 3 years in age (based on size comparisons with greenhouse seedlings) have been found growing beneath the parent, and only a very few have been seen growing in the



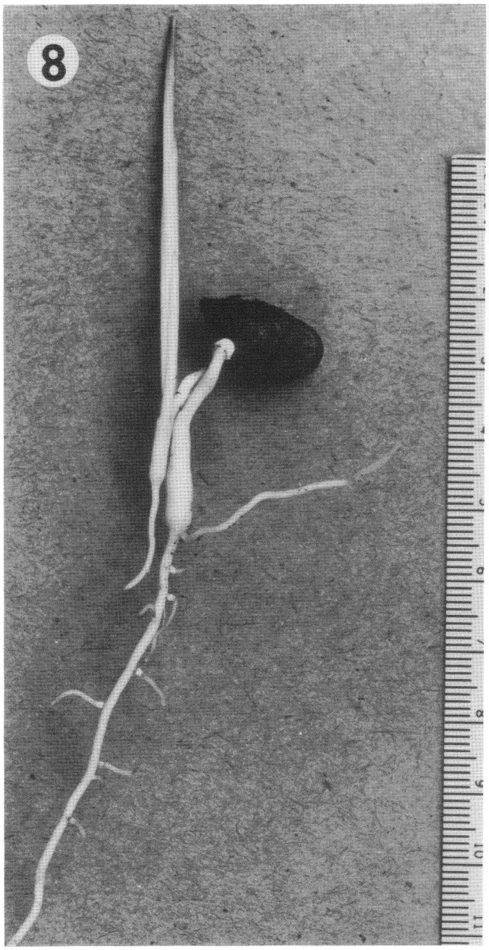
5–7. Later stages of seedling development in *Rhapidophyllum hystrix*. 5) Seedling at ca. 5 months (note the 2 seedlings developing from one seed. 6) Seedling at 2 years. 7) Seedling at 3 years. Note that all leaves are still undivided.

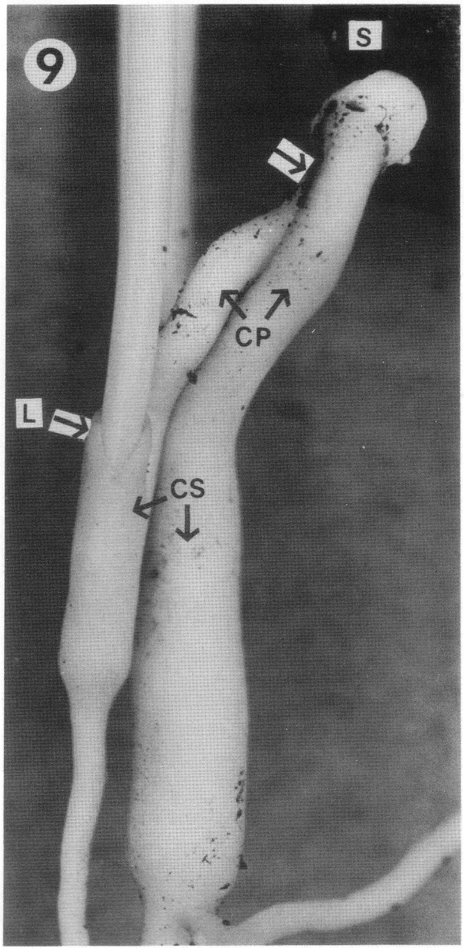
open. There is no known effective dispersal agent for *Rhapidophyllum*, although Manley (1967) thought mice chewed on the ripe fruits.

Polyembryony. During the germination experiments, one seed produced two seedlings. This resulted from the embryo splitting in two (cleavage polyembryony). However, one of the "twins" was definitely more vigorous than the other (Figs. 5, 8–10). The two seedlings were joined to a common petiole just beyond its emergence from the seed; below this point the seedlings were independent having separate cotyledonary sheaths, radicles and plumules (Fig. 9). However, they were both dependent on a common food reserve from the same endosperm. After 8 months both seedlings were still growing (Fig. 10). A comparison of Figure 5 (at 5 months) with Figure 10 shows that the smaller seedling had grown very little, if any, during these 3 months, while the larger seedling had grown substantially.

Table 2. Morphological characteristics of 2 year old seedlings of Rhapidophyllum hystrix. *All leaves are still entire and all measurements are for fully developed leaves.*

Characteristic	Mean ± s.d.	Range	Sample Size
Leaves per plant	3.6 ± 0.8	2–5	16
Leaf length (cm)	17.5 ± 3.6	10.0–24.4	45
Leaf width (cm)	1.9 ± 0.6	0.8–3.4	53
Major veins per leaf	4.0 ± 0.4	3–5	53

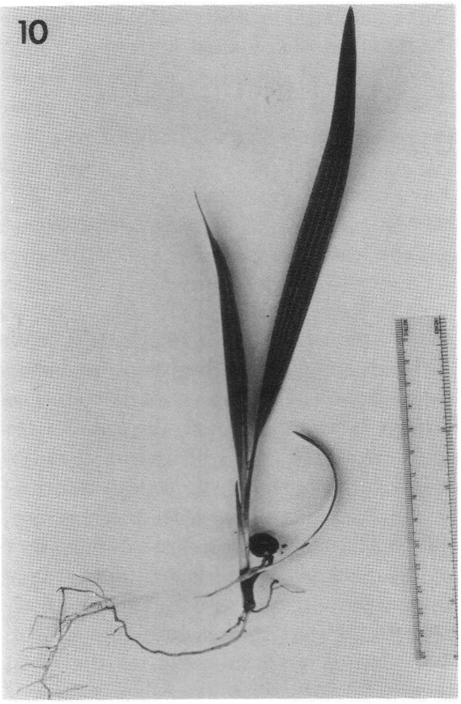




8–10. Polyembryony in *Rhapidophyllum hystrix*. 8) The double seedling resulted from the embryo splitting in two (cleavage polyembryony). Ruler is metric. 9) Close-up of part of Figure 8. Note the point where the 2 seedlings had split (arrow). Details: CP, cotyledonary petiole; CS, cotyledonary sheath; L, ligule; and S, seed. 10) The double seedling at 8 months. Note how much more one seedling has developed and compare with 5 month stage in Figure 5.

In palms, polyembryony is rare. The occurrence of multiple seedlings derived from one seed has been reported by Davis (1979) for the coconut *Cocos nucifera* L. and by Fisher and Tsai (1979) for the date *Phoenix dactylifera* L. Fisher and Tsai also reported on polyembryony in the palm *Syagrus* (*Rhyticocos*) *amara* (Jacq.) Glassman, which had three seedlings that had emerged from the same "eye", each with a separate cotyledonary stalk. These developed either from a single carpel which had three separate embryos or a single embryo which had divided to produce identical triplets.

Rhapidophyllum is apocarpic. Its gynoecium consists of three separate carpels, only one of which normally develops (occasionally 2 or 3 carpels from a flower reach maturity, pers. obs.). The double seedling shown in Figures 5, and 8–10 was derived from a single uniovulate car-



pel; the two seedlings are joined by a common petiole and consequently are genetically identical, a result of cleavage polyembryony. At the moment the occurrence of polyembryony for the needle palm is only a curiosity.

Acknowledgments

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Notes on Pollinating the Eleutheropetalum Group of Chamaedorea

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Don't be intimated by the long word, "eleutheropetalum!" It is easy to pronounce if broken into little pieces, and pronounced thus: E-loo-ther-o-pet'-a-lum. Say it aloud a few times and it will just roll off your tongue!

There are four members of the genus *Chamaedorea* that were once placed in a separate genus, *Eleutheropetalum* because their flowers are unique. They are described as having "bright flowers and pistillate flowers with valvate, hooded petals." The four species are *C. ernesti-augusti, C. metallica, C. stolonifera,* and *C. sartori.* The pollen from this group of plants is dry and, to some degree, is transported to the female inflorescences by air currents, but because the flowers are so brightly colored and fragrant, a good portion of pollination probably is achieved by insects that are found where the plants grow. The pollinating techniques that I will describe ensure that a maximum number of seed will be set on the female plants.

Of the four, *C. stolonifera* is the only one with multiple stems. *C. sartori* is the only one which commonly has divided fronds, and is not widely grown by palm collectors. It tends to grow rapidly into a rather spindly plant and there are other chamaedoreas with a similar appearance, such as *C. oblongata* which has more attractive glossy foliage.

Although there are exceptions to the rule, the pistillate or female inflorescences of *C. ernesti-augusti* and *C. metallica* are single, while those of *C. stolonifera* and *C. sartori* are branched. The pistillate flower spike of *C. ernesti-augusti* is erect and perpendicular to the trunk and may grow some distance above the foliage. The female inflorescences of the others are interfoliar. The male or staminate inflorescences on all four species are interfoliar and flowers are borne on numerous rachilla. The flowers on the respective sexes of the species are similar in appearance though they vary in size, the pistillate flowers of *C. ernesti-augusti* being the largest.

I learned to hand pollinate the eleutheropetalums years ago when I moved from Miami to San Francisco. I had a small collection of palms and other plants that I moved by suitcase and crates from Florida to California. In this hodgepodge of plants was a female *C. ernesti-augusti*; in California I had obtained some plants of *C. metallica* of both sexes. When the *C. ernesti-augusti* started to flower, I tried to pollinate it with *C. metallica* pollen since they looked similar, and that was all I had. I soon learned this would not work, so I brought back fresh *C. ernesti-augusti* pollen from Florida.

My first attempts were to stick the female inflorescence into the bag of pollen and shake vigorously. The old "shake the bag" method worked but not very well. Only a very small percentage of fruit set. During this period *C. ernesti-augusti* was a rather rare palm in the United States, certainly on the west coast, and I wanted to produce as many seeds as possible, so I sought a better method of setting seeds. I finally reached the conclusion that a majority of the flowers were not being pollinated because the pollen was not touching the stigma within the female flowers. I solved the problem by breaking away the three petals of the flower to expose the stigma and then brushed the pollen on each one individually.

This method worked very well, but over the years I have improved on the technique by using a little more sophisticated equipment and learning more about the process. To begin with, the most important aspect of this operation is to know when the female flowers have reached anthesis (when they are ready for pollination). The female (and male) flowers go through several subtle changes as they near maturity. As the flowers begin to expand over a period of days they go through several color changes. At first they are dull purplish brown, then deep magenta or wine, then a mustard color and finally a bright orange. At about the magenta or mustard color the petals begin to open, but the color phases and the speed at which the flowers open is affected by temperatures. The warmer it is, the more rapid the process.

Unlike many of the other chamaedoreas, the female flowers of the eleutheropetalums are receptive to pollination for a brief period, usually about 24 hours. Again cooler temperatures may extend this period, but the ideal temperature range is in the mid 70's to 80's. In addition to the opening of the flowers and the bright orange color, the arrival of anthesis is announced by a pleasing sweet fragrance. In a room or greenhouse, the sweet odor may be detected from several feet away. The flowers should be pollinated during this period of fragrance. The flowers may be pollinated a day or two ahead of expected anthesis so the pollen will be in place on the stigmas when anthesis arrives, but never after anthesis (or after the fragrance has gone away).

It is best to water the plant that is to be pollinated a day or two ahead of the expected time of pollination. This will allow the plant to absorb moisture and make the flower petals turgid and crisp so they can easily be broken away. If the plant is on the dry side, the petals may be flaccid and very difficult to remove. I once used a sharpened toothpick to remove the petals, but I've found a 90 degree angle dental tool is more handy.

Removing the petals can be a little tedious and time consuming, so I try to make the operation as simple and comfortable as possible. Since my plants are growing in containers, I place them at a comfortable height on a table or stool. I use an illuminated magnifying glass with a flexible arm so as to see better what I'm doing. I start at the top of the inflorescence so that I may steady it by lightly holding it with one hand. A light pressure of the finger tips on the lower part of the inflorescence will not crush the turgid flowers.

The tip of the dental tool is inserted just inside the tip of each petal and an outward and down pressure will cause the petal to snap away at its base. I'm careful not to touch the stigma with the dental tool or my fingers. This is the reason I work from the top to the bottom of the flower spike. Usually I will remove the petals about ⅓ down the spike, and then make a mark with a felt tip pen below the last flower depetaled. Then I brush the pollen on the exposed stigmas. I do this in ⅓ increments along the flower spike as soon as the petals are removed. The stigmas are rather delicate so the pollen should reach them while they are moist. Also with fewer flowers to be brushed at a time, I'm sure each one will be touched.

I use a soft sable hair watercolor brush. Camel hair is too coarse to pick up the fine dusty pollen. It is best to use fresh pollen, but pollen stored at room temperature up to two or three weeks is satisfactory. It may be stored even longer if placed in a refrigerator and kept dry. The pollen should be collected when the male flowers are bright orange and the inflorescence is fragrant. When I think the pollen is ready to be collected, I gently tap the inflorescence to see if pollen is beginning to fall from the flowers. Often the male flowers may be fragrant for one or two days before they begin to release pollen. If they are collected too soon the flowers may not readily release the pollen, and if too late, most of it may already have fallen from the flowers.

I collect the pollen by cutting off the entire male inflorescence into a mailing envelope in which the bottom corners have been folded up and taped. (The fine pollen can sift out of the corners if they haven't been folded up.) The opened envelope is placed under the inflorescence before it is cut as a slight jarring might cause much of the pollen to fall from the flowers. After the inflorescence is enclosed in the envelope the bottom should be rapped several times on a table to dislodge the remaining pollen from the flowers. The pollen will collect in the crease on the bottom of the envelope and can easily be picked up with the brush.

The pollen should be labeled and dated, since it may be needed later. I also label and date the flower spike, especially when hybridizing. Remember, it is important to collect pollen at the right time and save it, because when the female is ready there may not be any fresh pollen available. Fortunately, the male plants usually start blooming two or three weeks before the females. It is very frustrating to have the females ready and no pollen on hand.

If done properly and the timing is right, fruit can be set on every single flower on a spike. As the seeds begin to mature, the flower spikes will enlarge and turn bright orange. The black mature fruits on the bright orange spikes set against the beautiful green foliage is a striking sight. It takes about six to nine months for the fruits and the seeds within to mature. Maturity has been reached when the fruits turn black. The mature fruit may remain on the flower spike for several months. Care should be taken if the mother plant is left outside, as birds and two-legged-palm-seed-predators have an affinity for the seeds.

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The Use of Palms by the Cayapas and Coaiqueres on the Coastal Plain of Ecuador

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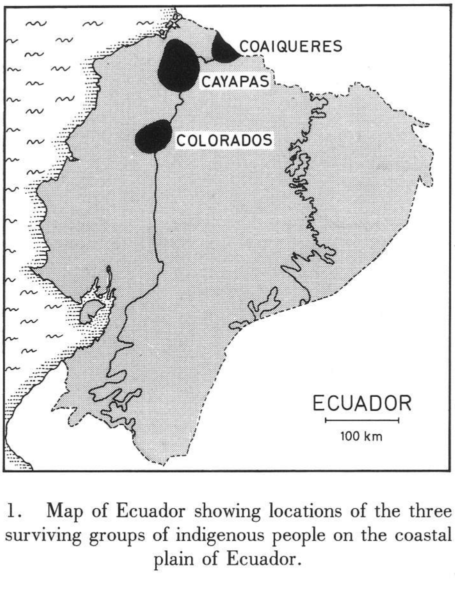
The Cayapas and Coaiqueres are two of the three remaining indigenous groups of western lowland Ecuador (Fig. 1). The third group, the Colorados (Tsatchela), will not be discussed although they are well known for their conspicuous appearance and folk medicine near Santo Domingo.

The Coaiqueres (Awas) include between 500 and 1,000 individuals living in the forest of Carchi province in northern Ecuador, near the border with Colombia. They have had extended contact with outsiders and their daily life includes elements foreign to their culture, such as western clothing, knives and shotguns. Most of them speak Spanish as well as their native language. Although outside influence appears to have been limited, it seems that the Coaquier culture is vanishing rapidly. At this time they still demonstrate an extensive knowledge of the natural resources surrounding them. In the special case of medicinal plants it appears, however, that they know less than other indigenous groups. This may be explained by their recent migration into the areas they now occupy, and that they may not have had enough time to explore these regions for plants with medicinal properties. In March of 1985 we visited the Coaiqueres and collected information on their uses and local names of palms. As in our previous ethnobotanical work with the Coaiqueres, we found that they were reluctant to provide Coaiquer names for the plants they knew, thus we can cite only Spanish names. The Coaiqueres have been described by Ehrenreich & Kempf (1978), Holm-Nielsen & Barfod (1984), Kvist & Holm-Nielsen (1987), and Villareal (1985).

The Cayapas (Chachi) live in the drainage basins of the Cayapas and Santiago rivers east and south of Borbón in the province of Esmeraldas. They comprise some 7,000 individuals who have lived for many years under the influence of western culture through contacts along the rivers and the presence of missionaries in their territory. Like the Coaiqueres they have assimilated some aspects of western culture, but appear to have maintained much of their traditional ways. Their knowledge of plants is extensive, although medicinal properties of plants are known mostly by



1. Map of Ecuador showing locations of the three surviving groups of indigenous people on the coastal plain of Ecuador.

Table 1. Alphabetical listing of the palms encountered in the territories of the Cayapas and Coaiqueres on the coastal plain of Ecuador, with collecting numbers of voucher specimens at AAU and OCA in brackets, their native names and uses.

Aiphanes sp. (Barfod & Skov 60.003).
Coiaqueres: Chontilla. Palm heart edible.

Astrocaryum standleyanum L. H. Bailey (Barfod & Skov 60.078).
Cayapas: Poca-chi. Raw fruit edible, fibers for hammocks etc. extracted from leaves.

Bactris gasipaes Kunth (Barfod & Skov 60.010, 60.113).
Cayapas: Cano-chi. Palm heart and cooked fruit edible. Wood used for house construction, fishtraps, blowguns, spears, and marimba keys. Edible larvae collected from decomposing stems.
Coaiqueres: Chonta duro. Palm hearts edible. Wood used for blowguns and marimba keys. Edible larvae collected from decomposing stems.

Bactris sp. (Barfod & Skov 60.110).
Cayapas: Pi-cano-chi. Palm heart and cooked fruits edible.

Catoblastus aequalis (O. F. Cook & Doyle) Burret (Barfod & Skov 60.002).
Coaiqueres: Gualte de parar. Trunks used for house construction.

Desmoncus sp. (Barfod, Kvist & Nissen 41.455).
Coaiqueres. Bora negra. Stems used for weaving baskets. Raw fruits edible.

Euterpe chaunostachys Burret (Barfod & Skov 60.001, 60.103).
Cayapas: Mamba-san-chi. Palm heart and raw fruits edible.
Coaiqueres: Palmito. Palm heart edible.

Geonoma gracilis H. A. Wendl. ex Spruce (Barfod & Skov 60.099, 60.104, 60.118).
Cayapas: Ya-ha-chi. Raw fruits edible.

Geonoma linearis Burret (Barfod & Skov 60.115).
Cayapas: Yullpo-pi-chui-tape. Ritual plant.

Geonoma sp. (Barfod & Skov 60.004).
Coaiqueres: Cola-pato. Leaves used to wrap food.

Iriartea deltoidea Ruiz & Pavon (Barfod & Skov 60.097).
Cayapas: Boun-chi. Wood used for blowguns, house construction, fishtraps, spears, marimba keys etc. Palm heart edible. Edible larvae collected from decomposing stems.
Coaiqueres: No name. Wood used for marimba keys. Edible larvae collected from decomposing stems.

Jessenia bataua (Mart.) Burret (Barfod & Skov 60.006, 60.079).
Cayapas: Cola-pa-chi. Leaf base fibers for blowgun darts.
Coaiqueres: Chapil. Leaf base fibers for blowgun darts. Palm heart edible.

Oenocarpus mapora Karst. (Barfod & Skov 60.102).
Cayapas: Uin-ga-chi. Leaf rachis fibers for weaving baskets.

Palandra aequatorialis (Spruce) O. F. Cook (Barfod & Skov 60.111).
Cayapas: Din-chi. Leaves used for thatch and fibers. Endosperm and mesocarp edible.

Pholidostachys dactyloides H. E. Moore (Barfod & Skov 60.109).
Cayapas: Ah-casta-ya-ha-chi. No uses.

Prestoea sejuncta L. H. Bailey (Barfod & Skov 60.107).
Cayapas: Chapin-sa-chi. Palm heart edible. Leaves used for thatch.

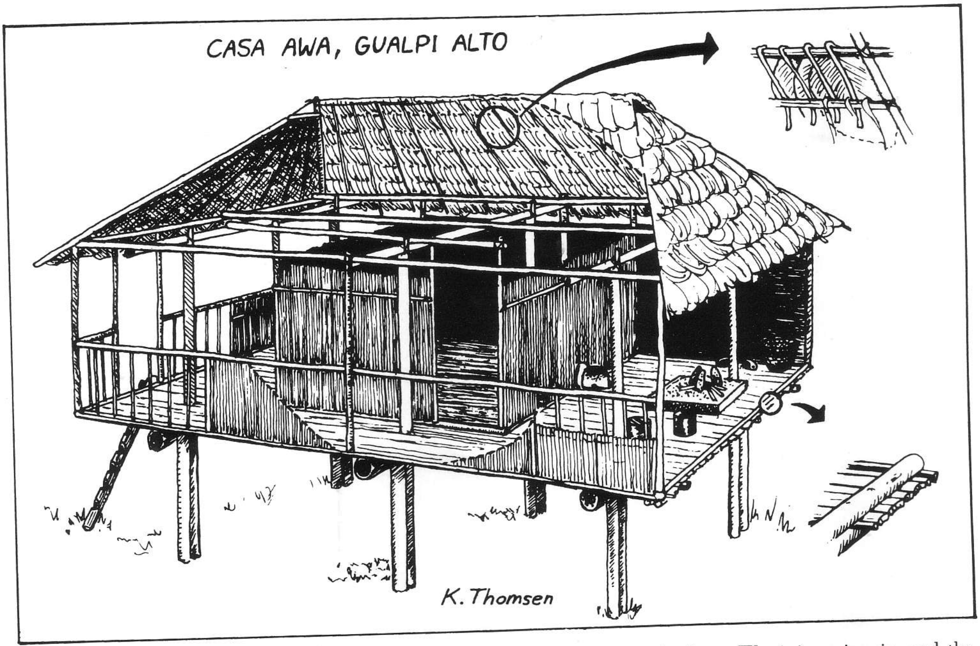
Socratea exorrhiza (Mart.) H. A. Wendl. (Barfod & Skov 60.007).
Cayapas: Pin-ua-chi. Trunks used for house construction. Palm heart edible. Edible larvae collected from decomposing stems.
Coaiqueres: Gualte crespo. Trunks used for house construction. Cooked fruits edible. Edible larvae collected from decomposing stems.

Synecanthus warscewiczianus H. A. Wendl. (Barfod & Skov 60.117).
Cayapas: Bo-chui-cano-chi. Inhabited by evil spirits.

Wettinia quinaria (O. F. Cook & Doyle) Burret (Barfod & Skov 60.005, 60.112).
Cayapas: Yan-chi. Leaves used for thatch. Trunks used for house construction. Fruits edible. Edible larvae collected from decomposing stems.
Coaiqueres: Gualte, Gualte bola. Trunks used for house construction. Edible larvae collected from decomposing stems.

older people. The younger generation is familiar with the many structural properties of plants, and how to use them for wood, fibers etc. In April 1985 we visited the Cayapas and collected information on the uses of and local names for palms. They were much more willing to give us indigenous palm names than were the Coaiqueres. The Cayapas were described by

2. In the Coaiqueres territory *Wettinia quinaria*, one of the most common trees, is used for house construction. This palm is also extensively used by the Cayapas.



3. A typical Coaiquer house. Note the raised floor which is made of trunks from *Wettinia quinaria*, and the corner posts which are of the same species.

4. *Palandra aequatorialis* is commonly used for thatch by the Cayapas. The leaves are split in half, and fastened to the roof with the fibers from the leaf rachis of the same species.

5. Before the leaves of *Palandra aequatorialis* are placed on the roof, they are soaked in water for several weeks to prevent them from curling when dry.

Barfod et al. (in press), Holm-Nielsen & Barfod (1984), Holm-Nielsen et al. (1983), and Kvist & Holm-Nielsen (1987).

In the following we discuss our findings which are summarized in Table 1. The information is documented with voucher specimens in the herbaria of the Botanical Institute, University of Aarhus (AAU) and of the Pontificia Universidad Católica in Quito (QCA).

Construction

Iriartea deltoidea, *Socratea exorrhiza*, *Wettinia quinaria*, *Catoblastus aequalis* and *Bactris gasipaes* are all used for house construction by both tribes. The species used depends upon availability and energy necessary for extracting it from the forest. The Coaiqueres prefer *Wettinia quinaria* (Fig. 2) for house posts. It is a very common species in their territory, and they leave it when the land is cleared for cultivation or pasture because its stem is so hard to cut, and because it takes up little space in the fields. The floors of the houses are raised 1.5–2 meters above the ground. The same species of palm is used for making the floors (Fig. 3). For that purpose the stem is split open, the soft central part is removed, and the outer fibrous part of the stem then forms long, but very strong boards. These boards are convenient for floors because they are flexible and therefore comfortable to walk and sit on. The longitudinal spaces between boards permits dirt to fall through to the ground. When *Wettinia quinaria* is hard to find, *Socratea exorrhiza* and *Catoblastus aequalis* replace it. These species are used in the same way.

The Cayapas may use the same species as the Coaiqueres for house construction, but more often they use *Bactris gasipaes*. Its wood is even harder than that of other

6. Cayapa blowgun made of wood from *Iriartea deltoidea* and bound with pieces of natural rubber.

species, and forms very durable boards, but is more difficult to cut. In addition, *Bactris gasipaes* is cultivated for its fruits, and therefore it may be undesirable to fell it for wood. *Iriartea deltoidea* is also used by the Cayapas for posts and boards for their houses.

The Cayapas use *Wettinia quinaria* and

7. Coaiqueres blowgun made of wood from *Bactris gasipaes*. The blowgun pipe is bound with natural fibers and sealed with beeswax.

Prestoea sejuncta for thatch, but *Palandra aequatorialis* (Fig. 4) is most commonly employed for that purpose. In the Cayapa territory this palm is common along the rivers, and it is left standing when the forest is cleared for cultivation. The leaves are 5–6 meters long with 80–90 cm long pinnae inserted irregularly along the central rachis. When the leaves are harvested for thatch, only the outer ones are taken. The juvenile leaves are left so that the plant may survive. Harvested leaves are split into two halves along the rachis and then soaked in a pond for several weeks (Fig. 5). This prevents the pinnae from curling up when the thatch is later exposed to the sun. The leaves are then placed on the roof, and the rachides are fastened to the roof construction. The pinnae are not woven into each other.

The Coaiqueres do not use palm leaves for thatch, but instead an undescribed species of *Calathea*, Marantaceae (H. Kennedy, pers. com.).

Fibers

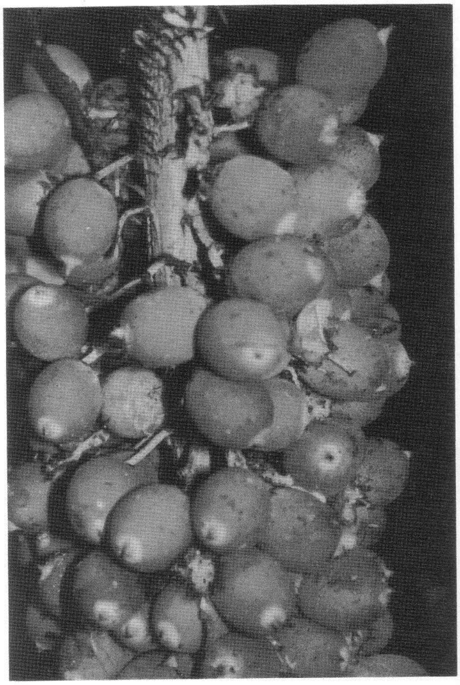
The Cayapas use fiber extracted from the leaf pinnae of *Astrocaryum standleyanum* for hammocks, fishing nets and baskets. This palm is not common in the part of the Cayapa territory we visited, and we did not observe this use, but were told of it. The leaf rachis of *Palandra aequatroialis* produces fibers which are used to fasten the leaves of the same palm on the roof as thatch. The leaf rachis of *Oenocarpus mapora* produces fine fibers which are used for weaving baskets. In addition to these palm fibers, the Cayapas extract fibers from *Carludovica palmata*, Cyclanthaceae (the Panama Hat Plant).

The Coaiqueres use the stems of the climbing palm *Desmoncus* to make baskets

8. Coaiqueres fishtraps made of wood from *Bactris gasipaes*. When the water level is high, fish enter the trap through the basal opening. A shutter falls down and blocks the exit when the fish nibbles at the bait.

9. Coaiquer marimba with keys made of wood of *Iriartea deltoidea.*

and ropes. Its stem is only a few centimeters thick, but may reach 30 meters in length, and the entire plant climbs through the vegetation by means of the apical leaf segments which are modified into strong hooks. The Coaiqueres do not have a palm for finer fibers such as those of *Astrocaryum*, but instead use "Pite," a species of



10. *Astrocaryum standleyanum* fruits are eaten raw by the Cayapas. They are of an inferior quality compared with those of *Bactris gasipaes* because the mesocarp is fibrous.

Fourcroya, Agavaceae, which may be a recent introduction to their territory.

Weapons, Traps and Instruments

Both the Coaiqueres and the Cayapas make blowguns (Figs. 6, 7) out of the wood of *Bactris gasipaes*. The Cayapas may also use the wood of *Iriartea deltoidea* for this purpose. To make a blowgun two pieces of wood are cut out of the outer strongly sclerified layer of the trunk. Each piece is about 5 × 2 cm in cross section and 3–5 meters long. The pieces are cut so that they are semicircular in cross section, and a straight furrow is carved in the center of the flat side of each piece. The two pieces are then joined to form a long tube with a fine central bore. The Cayapas join the two halves with long pieces of rubber from a locally common tree (*Castilla elastica*, Moraceae). The Coaiqueres bind a plant fiber around the two halves to keep them together. They then rub it with beeswax, and heat it over a fire until the beeswax is black. This process assures an airtight pipe. For blowgun arrows both groups use the strong fibers produced at the leaf base of *Jessenia bataua*. These are collected from the palm, sharpened on one end with a knife, and used without further treatment. To assure an airtight position of the arrow in the blowgun bore, one end is wound with a small pellet of the fibers from the Kapok tree (*Ceiba pentandra*, a Bombacaceae).

The Cayapas make fishtraps (Fig. 8) with the wood of *Bactris gasipaes* and *Iriartea deltoidea*. The fibrous wood of these two species is water resistant and fishtraps produced from it last long, even if left permanently in the river. The wood of the same two species is also used for spears.

The Marimba (musical instrument) of the Cayapas and Coaiqueres (Fig. 9) has keys made of wood from *Bactris gasipaes* or *Iriartea deltoidea*. This instrument is not indigenous, but was introduced, probably from the Caribbean region.

Food Products

The Cayapas cultivate *Bactris gasipaes* for its edible fruits. This species is planted throughout tropical America, and its origin as a domesticated plant is uncertain. It is a tall, spiny palm with many stems. Its fruits are the size of a hen's egg and bright orange when ripe. The fruits are boiled before they are eaten, and are a local small-scale commercial product. The Cayapas also eat the fruits of *Astrocaryum standleyanum* (Fig. 10), but it is not cultivated. Its fruits are about the same size as those of *Bactris gasipaes* and are eaten raw, but they are of an inferior quality because the edible mesocarp is quite fibrous. The fruits of *Euterpe chaunostachys* and

11. The palm heart of *Prestoea sejuncta* is extracted by a Coaiquer Indian. The sheathing leafbases form a crownshaft which is soft and easy to cut. The palm heart is about 50 cm long and 3–4 cm thick.

Geonoma gracilis are also eaten raw. The Cayapas drink the watery endosperm of *Palandra aequatorialis* as a refreshment when travelling or working in the forest. When the endosperm starts solidifying and is gelatinous it is still considered a delicacy. The mesocarp of the same species is edible when the fruits are young; it is yellow and has a bland taste. The Cayapas eat the palm hearts of a number of species. They prefer *Euterpe chaunostachys* (Fig. 11) and *Prestoea sejuncta*, but may use others such as *Bactris gasipaes*, *Bactris* sp., *Iriartea deltoidea* and *Socratea exorrhiza*. The palm heart consists of the unopened new leaves which are protected in the crownshafts by many layers of old leaf sheaths. The individual palm from which a heart is extracted is killed by the extraction, so it is a very destructive use of the forest. Usually however, palm hearts are harvested from trees felled for construction or other purposes.

The Coaiqueres cultivate *Bactris gasipaes* less frequently than the Cayapas do, but use it for the same purposes. They also eat the raw fruits of *Desmoncus* sp., which are quite small but claimed to be delicious. The palm heart of *Euterpe chaunostachys* is the most popular among the Coaiqueres, but they, like the Cayapas, eat the hearts of a variety of palms including *Aiphanes* sp., *Bactris gasipaes* and *Jessenia bataua*. When travelling in the forest the Coaiqueres sometimes subsist only on the palm hearts they are able to collect.

Beetle Larvae

The Cayapas and the Coaiqueres collect larvae from decaying stems of *Bactris gassipaes*, *Iriartea deltoidea*, *Wettinia quinaria* and *Socratea exorrhiza*. The larvae are up to 7 cm long and come from a weevil, *Rhynchoporus palmarum* (Latv.) (Fig. 12). When the wood decomposes, the

12. Larvae found on rotting palm stems are considered a great delicacy by the Coaiqueres and Cayapas.

13. *Synecanthus warscewiczianus* is one of the understory palms which the Cayapas believe is inhabited by evil spirits. The shaman often uses these palms to drive away other evil spirits that may cause illness.

larvae produce an intense odor that can be detected from some distance. The larvae are considered a delicacy by most indigenous peoples and are eaten either raw on the spot or brought home for frying. They have a soap-like aftertaste when raw, but otherwise taste good and are probably very nutritious.

Ritual Uses of Palms

Palms are used in many rituals without obvious physiological effect. These rituals often have religious significance and it is often difficult to obtain reliable information. The shaman's curing rituals are especially secret because members of the tribe are afraid of the close contact between the shaman and the spiritual world. Many of the understory palms such as *Synecanthus warscewiczianus* (Fig. 13) and *Geonoma* spp. were said to be inhabited by evil spirits. All organic and inorganic matter is inhabited by spirits according to the animistic belief of the Cayapas. Some are believed to be more evil than others and these are often mentioned. Passing close to a plant or an object that houses a spirit does not enhance the risk of being attacked. However, the Shaman is able to drive these spirits out from where they live and use them in curing (Barfod et al., in press).

Conclusions

We have recorded 19 species of palms known and used by the Cayapa and Coaiquer tribes of the coastal plain of Ecuador.

This corresponds with 45 percent of the total number of palms recorded for the coastal plain of Ecuador by Balslev & Barfod (1987). The Cayapas know 15 of them, and the Coaiqueres know 10. Most uses of the palms are based upon their structural properties. This includes construction of houses, thatch, musical instruments, weapons, etc. This category of uses is explained by the high content of fiber and sclerenchyma in the stems and leaves, which make palms particularly well suited for such purposes. The other important category is food plants. Fruits especially are edible and nutritious but the palm heart is very commonly eaten by both groups. Other uses are less important, and it is noteworthy that no palms are used medicinally by the Cayapas and Coaiqueres, which may be explained by the absence of physiologically active secondary metabolic compounds.

Acknowledgments

The Ethnobotanical studies among the indigenous groups of coastal Ecuador was started by L. B. Holm-Nielsen with support from the Museo Antropológico del Banco Central, Guayaquil. It was during these studies that Anders Barfod became acquainted with these people. The Danish Natural Science Research Council has supported Henrik Balslev's work on Ecuadorean palms. Flemming Skov participated in the fieldwork. We are most thankful for the help and support given to us in these projects.

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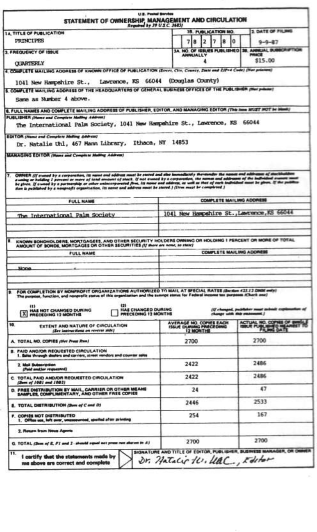
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NEWS OF THE SOCIETY

The I.P.S. Downunder September 1988

The 1988 Biennial Meeting is going to be unique! Centered in the heart of palm country in tropical North Queensland, Australia, with the opportunity to see up to 40 species of palms in the wild, "Palms in Habitat" is the natural theme of the meeting. In addition, Australia celebrates its 200th birthday this year and Brisbane hosts Expo '88, so what better time to visit downunder?

The meeting and post meeting activities are split into a number of segments, with options, each with a character and theme of its own.

The meeting starts with nine days in the Cairns district hosted by the Palm and Cycad Societies of Australia. Conference activities include addresses, slide shows, dinners, an Australian banquet with a difference, and a host of guest speakers as well as a number of tours to:

1) The lowland tropical rain forests of the Raintree and Cape Tribulation areas, soon to be considered for World Heritage listing.
2) One of the largest private collections of palm species in the world, with over 500 species under cultivation, in large grounds in a rural setting. This is guaranteed to turn those from cooler climates green with envy.
3) Mt. Lewis in the rugged partially unexplored ranges inland from Mossman and Raintree, sometimes referred to as a natural botanic garden. This area is often cloaked in cloud.
4) Flecker Botanic Gardens with its variety of natural and man-made habitats from lakes and *Licuala* swamps to a rain-forested hillside.
5) Coastal areas south of Cairns including lowland swamp forests and *Nypa* in habitat.
6) The historic mining town of Ravenswood and a beautiful unnamed glaucous *Livistona* sp., destined to be submerged by the rising waters of a new dam.

Palms seen in habitat will include: *Archontophoenix alexandrae, A.* sp. (Mt. Lewis), *Arenga australasica, Calamus australis, C. caryotoides, C. hollrungii, C. moti, C. radicalis, Hydriastele wendlandiana, Laccospadix australasica* and the unusual single stemmed variety, *Licuala ramsayi, Linospadix aequisegmentosa, L. microcarya, L. minor, L.* sp. (Mt. Lewis), *Livistona decipiens, L. drudei, L. muelleri, L.* sp. (Palunrea Range), *L.* sp. (Ravenswood), *Normanbya normanbyi, Nypa fruticans, Ptychosperma elegans, Oraniopsis appendiculata,* and for the cycad buffs *Cycas media, Bowenia spectabilis* and variants, *Lepidozamia hopei.*

The meeting culminates by moving to Townsville for five days for a major event to all palm enthusiasts and researchers, the official opening of the Townsville Bicentennial Palmetum, a new botanic garden set in 25 hectares. The perhaps unique concept of "Environmental Preference Planning" takes full advantage of the distinct naturally occurring range of habitats. This segment is hosted by the North Queensland Palm Society and the Townsville City Council. It includes a visit to an amazing aquarium, the "Barrier Reef Wonderland," and visits to displays illustrating palm products, palm exploitation, and palms in horticulture, presented in conjunction with the Palmetum opening.

A post meeting segment offers optional visits to Brisbane and Sydney.

Spend three days in Brisbane hosted by the Southern Queensland Group of the P.A.C.S.O.A. Tour the city's two famous botanic gardens, visit Expo '88 and perhaps travel farther afield to our rain forests or the Gold Coast beach resort.

Palms to be seen in habitat in the Brisbane area include *Archontophoenix cunninghamiana, Calamus muelleri, Linospadix monostachya, Livistona australis* and for the cycad people *Lepidozamia peroffskyana, Macrozamia lucida, M. miquelli.*

The venue for the final segment of the tour is Sydney, Australia's largest city and center for the Bicentennial celebrations. The New South Wales Chapter of the I.P.S. hosts this segment and offers an active three day program, including visits to private gardens, sightseeing on Sydney's famous harbor, and a visit to the Royal Botanic Gardens and herbarium. The Garden has an extensive collection of mature palms amd this humid temperate collection will be a contrast to the palms of tropical North Queensland.

After 20 days it's farewells and head for home. A full itinerary, package costing, and registration form will be mailed to you soon. Please return the registration form early to assist the organizers and feel free to write for further information.

DAVID TANSWELL
Director I.P.S. 1986–1990
President, Palm and Cycad
Societies of Australia

The Biennial 1988

Seldom do members have an opportunity to attend an IPS biennial conference which offers the wealth of activities anticipated for the 1988 Biennial in Australia (see above).

Initially headquartered in the Queensland city of Cairns, the conference offers one-day access to the palm-rich areas of Daintree, Cape Tribulation and Mt. Lewis. Moving south to Townsville, conferees will participate in the dedication of an ambitious palmetum. This experience will illustrate the extraordinary potential of cooperation by the public and private sectors to enhance knowledge and appreciation of palm species, both native and exotic.

Well established botanic gardens with mature palm specimens await conferees when they arrive in Brisbane and Sydney. Also waiting will be the activities associated with the 1988 World Exposition in Brisbane and Sydney. And always there is the lure of beaches, boating, and the barrier reef.

My wife and I had the good fortune to visit Australia on the post-conference tour after our 1984 Biennial in San Francisco. The richness of travel which Australia offers—and which we barely tapped—assures our return. I hope that you will join us and share our excitement.

IPS members and chapters in Australia are working diligently to make our 1988 Biennial a series of enticing and informative experiences. Let's show our appreciation and support by responding enthusiastically. We can start by responding promptly to requests for information. The first such request will reach you soon. If you have even tentative plans to attend, please complete the forms and return them as soon as possible. I feel sure that you will be glad that you did, whether your "bag" is seed collecting, bird watching or scenic adventure.

EDWARD M. MCGEHEE
President, International Palm Society