ANTHURIUM CULTURE IN HAWAI'I
Edited by Tadashi Higaki, Joanne S. Lichty, and Darlene Moniz

THE EDITORS

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Cover Photo: Obake anthurium.

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INTRODUCTION

The anthurium, *Anthurium andraeanum* André, a native of Colombia, was first brought to Hawai'i from London in 1889 by S. M. Damon. Today, after over 100 years of cultivation and hybridization, the Hawaiian anthurium is one of the islands' principal ornamental exports to the U.S. Mainland, Canada, Japan, Italy, Germany, and other countries.

The anthurium belongs to the family Araceae which includes more than 100 genera and about 1500 species, chiefly from the tropics. Some of the better-known members of the family include the pig-tail anthurium, philodendron, monstera, taro vine, taro, calla lily, caladium, 'ape', aglaonema, and dieffenbachia, most of which usually grow under environmental conditions of low light and very high humidity.

The anthurium is a perennial herbaceous plant usually cultivated for its attractive, long-lasting flowers. What is commonly considered the flower is comprised of a colorful, modified leaf (spathe) and hundreds of small, botanical flowers on the pencil-like protrusion (spadix) rising from the base of the spathe (Figure 1).

The anthurium plant produces flowers throughout the year. One flower emerges from each leaf axil. The sequence of leaf, flower, and new leaf is maintained throughout the entire life of the plant. The interval between leaf emergences is shortened or lengthened with changes in environmental conditions. During the summer months when conditions are favorable for growth, more flowers can be expected per plant than during the winter when temperatures are lower and there is less light.

Growing anthuriums was, for many years, a "backyard" operation. It has rapidly become a commercial industry in which individual farms may be as large as 20 acres or more. In 1986, there were about 85 farms with total sales of $10,000 or more. There were a total of 440 acres in anthurium production, with over two million dozen cut anthuriums sold for a wholesale value of $9.9 million. Since then, anthurium production in Hawai'i has declined, due primarily to the bacterial blight disease. In 1992, there were 69 farms with over $10,000 in total sales cultivating about 240 acres under artificial shade structures, plastic-covered shade structures, and under natural shade. The wholesale value of Hawai'i's 828,000 dozen cut anthuriums sold in 1992 was $6.4 million.

Table 1. Standards for anthuriums in Hawai'i

Grade	Average of Length + Width of Spathe
Miniature	under 3"
Small	over 3" to 4"
Medium	over 4" to 5"
Large	over 5" to 6"
Extra Large	over 6"

COMMON CULTIVARS

Numerous anthurium cultivars have appeared over the years. Several cultivars that appeared during the 1930s and 1940s such as 'Ozaki', 'Nitta', 'Kozohara', 'Kaumana', and 'Madame Butterfly' have continued to enjoy popularity in commercial production. In the 1960s and 1970s, several new cultivars including 'Marian Seefurth', 'Mauna Kea', 'Anuenue', and 'Calypso' were introduced by the University of Hawai'i. More recently, cultivars with new colors such as purple, lavender, and green have appeared and expanded the diversity of anthuriums.

Anthurium cultivars can be broadly classified into standards, obakes, and tulip types. Standards usually have self-colored (single-colored) spathes. A desirable shape of the spathe is a broad, symmetrical, heart shape with slightly touching or overlapping basal lobes. The spadix should be shorter than the length of the spathe and gently reclined to facilitate packing for shipment. The five major anthocyanin spathe colors are red, orange, pink, coral, and white.

The obakes are extremely variable in size, shape, and color. They are usually bi-colors of green and the major spathe colors, e.g., red-green obake, orange-green obake,
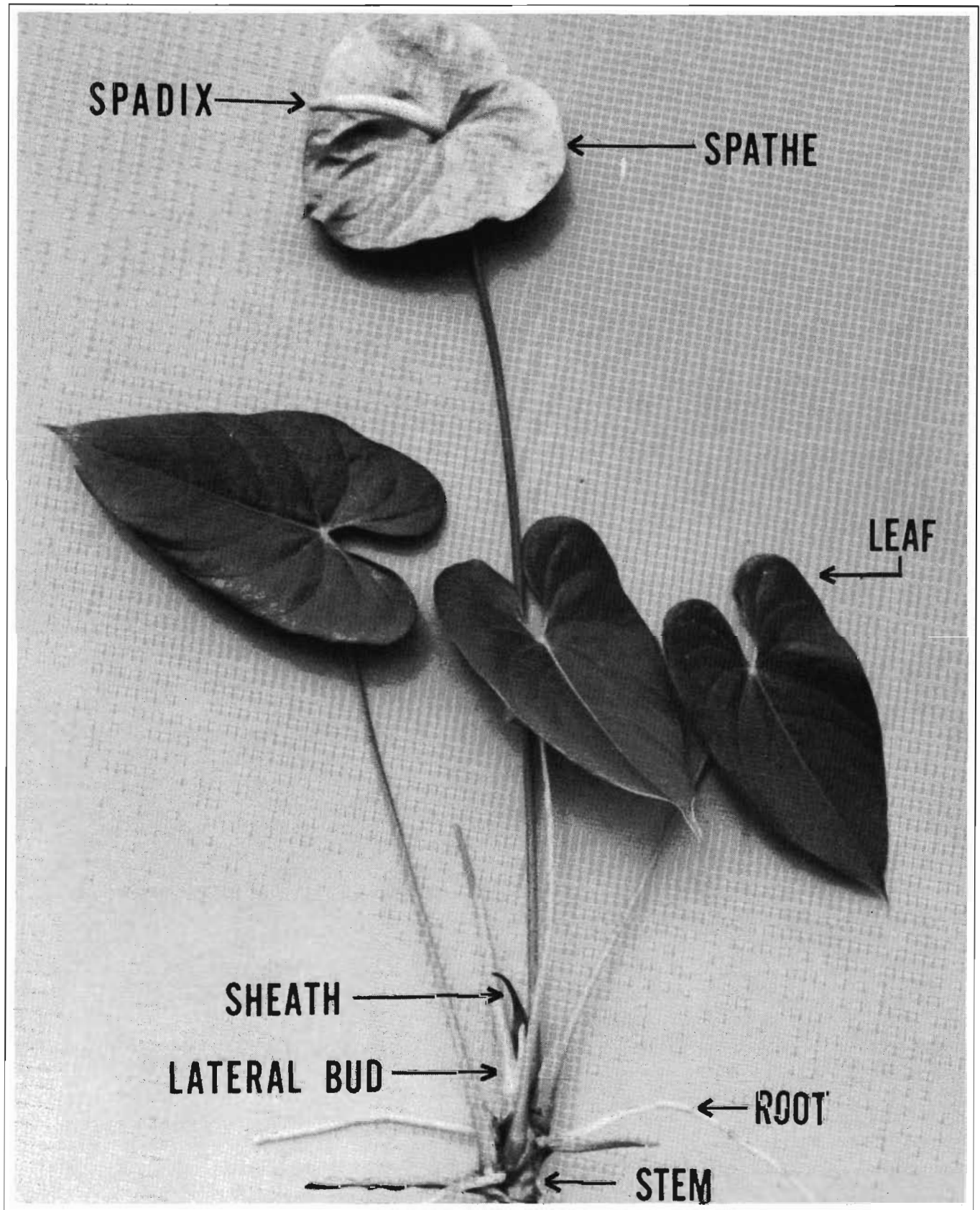
Figure 1. Typical *Anthurium andraeanum* plant.

etc. The amount of green shown by the spathes of obakes may change with the season (Figure 2).

The tulip-type anthuriums are relatively new to Hawai'i. They are generally characterized by an upright, cupped spathe similar to an individual tulip petal. The spadix is also straight and erect as contrasted to the reclining spadices of most standards.

Standards

'Ozaki'. The spathe is bright red, broad, heart-shaped with overlapping lobes, and usually large, measuring $6\frac{1}{2} \times 6\frac{1}{4}$ inches (length × width). The reddish purple spadix is usually $3\frac{1}{2}$ inches long and reclined but becomes more erect as the flower matures. It is a vigorous grower and produces suckers. To prevent sun burn, 'Ozaki' requires more shade than some other cultivars. 'Ozaki' is presently the most popular commercial cultivar. It is not as susceptible to anthracnose or spadix

rot as 'Kaumana' and 'Kozohara' but is susceptible to bacterial blight. This cultivar originated in the collection of O. Ozaki of Hilo about 1936 (Figure 3).

'Kaumana'. The dark red, heart-shaped spathe is usually $5\frac{1}{4} \times 4\frac{1}{4}$ inches (length × width). The spadix is usually $3\frac{1}{2}$ inches long and reclining. 'Kaumana' is one of the most reliable, high-yielding, small- to medium-sized anthuriums. It grows rapidly and produces many suckers. It is highly susceptible to anthracnose.

'Kozohara'. The spathe is dark red (similar to 'Kaumana'), heart-shaped with overlapping lobes, and usually large, measuring $6\frac{1}{2} \times 5$ inches (length × width). The reclining spadix is $3\frac{3}{4} \times 5$ inches long. It is highly susceptible to anthracnose.

'Nitta'. The orange, broad, heart-shaped spathe with overlapping lobes is usually 6×5 inches (length × width). The white, reclining spadix is about $3\frac{3}{4}$ inches long. Among the orange cultivars, 'Nitta' is the
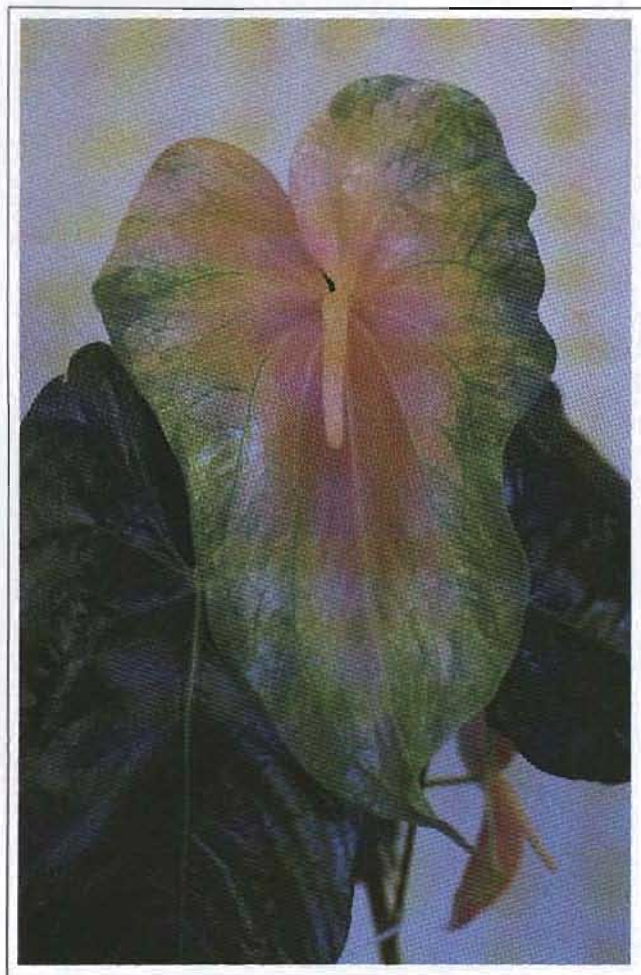
Figure 2. Obake-type anthurium.
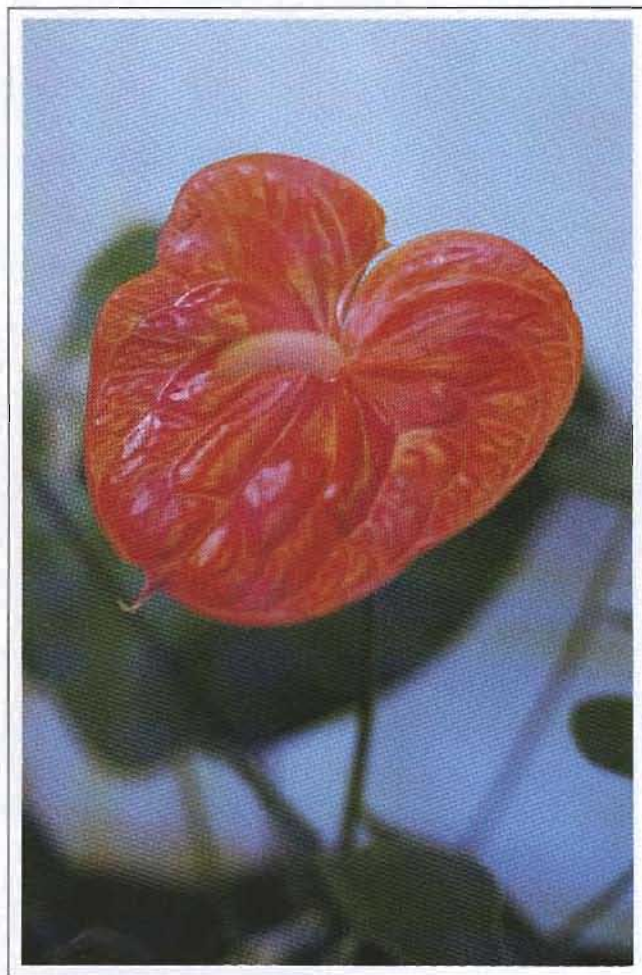
Figure 3. 'Ozaki'.

principal commercial variety. It is a vigorous grower, suckers freely, and produces high yields. Although the flowers are extremely strong and have a long shelf life, 'Nitta' has a tendency to produce flowers that are flat and more or less parallel in line with the stem. 'Nitta' originated in the collection of Asako Nitta in 1946.

'Marian Seefurth'. The broad, heart-shaped, rich rose pink spathe has unusually large ($6\frac{1}{2} \times 5\frac{1}{2}$ inches [length × width]), overlapping lobes. The spadix is $3\frac{1}{2}$ inches long, greenish yellow and reclining when young, and white and upright when mature. This University of Hawai'i introduction is an exceptionally high-yielding cultivar. It is highly resistant to anthracnose, but extremely susceptible to bacterial blight (Figure 4).

'Blushing Bride'. The medium-sized spathe, $6\frac{1}{2} \times 5$ inches (length × width) in size with overlapping lobes, is tinged with pink on a white base. The reclining $3\frac{3}{4}$-inch spadix is purplish pink. It is resistant to anthracnose and tolerant to bacterial blight. It suckers freely. This cultivar appears to be sensitive to diuron.

'De Weese'. The white, open, heart-shaped spathe is usually small, measuring $3\frac{3}{4} \times 3\frac{3}{4}$ inches (length × width). Its yellow, reclining spadix is $3\frac{3}{4}$ inches long. Flower yield is fairly high and it produces suckers freely.

'Ellison Onizuka'. The large, paper white spathe measures up to $8\frac{1}{2}$ inches long and $6\frac{1}{2}$ inches wide. It is heart-shaped with slightly fused basal lobes. The yellow, slightly reclining $3\frac{1}{2}$-inch spadix is resistant to anthracnose.

Obakes

'Madame Butterfly'. This miniature, dark red obake cultivar is also known as 'Mickey Mouse'. The spathe is about 3 inches long and $2\frac{1}{2}$ inches wide and triangular with open lobes. The spadix is red-orange. It can be grown for cut flowers or as a potted plant.

'Anuenue'. The green and coral pink spathe is 11×9 inches (length × width) and rounder than most other
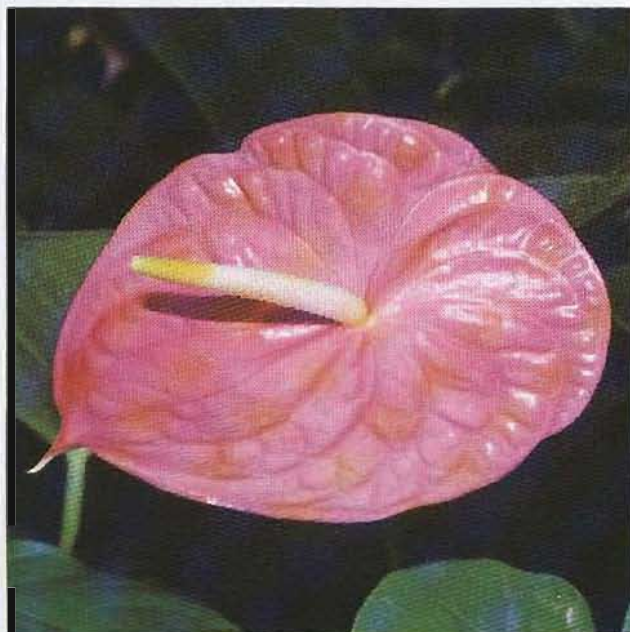
Figure 4. 'Marian Seefurth'.
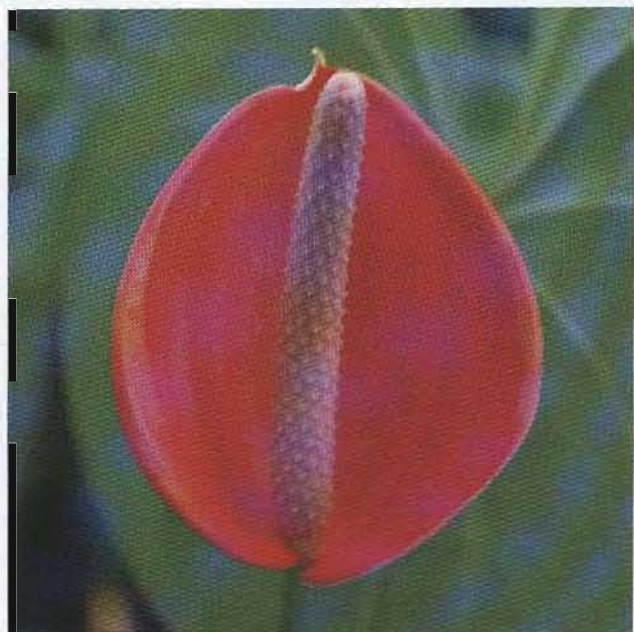
Figure 5. 'Calypso'.

obakes. Its yellow to white, 4-inch-long, reclining spadix is highly resistant to anthracnose. Flower stems are thick and about 25 inches long. Flower yield is good. However, internode length is long as is often the case with obakes.

'*Mauna Kea*'. The major attributes of this cultivar are its attractive white with green border, $8\frac{1}{2} \times 8$-inch (length × width), round spathe with overlapping lobes; its yellow to white, $3\frac{1}{2}$-inch, reclining, anthracnose-resistant spadix; and its relatively high yield. Flower stems are thick, erect, and about 24 inches long. Plant internode length is long and sucker production is poor.

Tulip Types

'*Calypso*'. The spathe size of this cultivar is $4\frac{1}{4} \times 3$ inches (length × width). The spathe is dark pink to light red on the inner surface, while the outer surface is light pink. The dark pink spadix is about $4\frac{1}{8}$ inches long. Sucker production is poor (Figure 5).
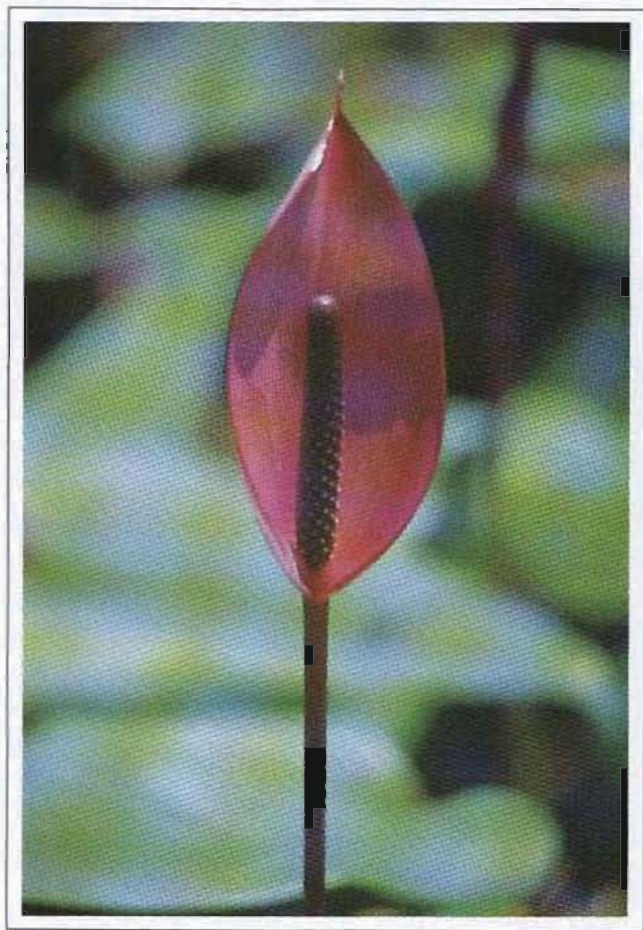
Figure 6. 'ARCS'.

'*Trinidad*'. The spathe size of this cultivar is about $4 \times 2\frac{1}{4}$ inches (length × width). The color is off-white with a light flush of maroon. 'Trinidad' has a somewhat less cupped spathe than 'Calypso'. The spadix is nearly 4 inches long, slightly darker in color than the spathe, and erect. Sucker production is fair.

'*ARCS*'. The purple color of the spathe of this cultivar is novel. The upright, slightly cupped spathe is 5 inches long and $2\frac{3}{4}$ inches wide. The purple spadix is upright and about $3\frac{1}{4}$ inches long. It is resistant to anthracnose. 'ARCS' has four *Anthurium* species in its background: *A. andraeanum*, *A. formosum*, *A. kamemotoanum*, and *A. amnicola* (Figure 6).

'*Lavender Lady*'. Like 'ARCS', 'Lavender Lady' has a color new among anthurium cultivars. It is a sibling of 'ARCS'. The lavender spathe is 6 inches long and $2\frac{1}{2}$ inches wide, narrow, pointed at the tip, and recurved. The purple spadix is about $2\frac{1}{2}$ inches long, erect, and resistant to anthracnose.

Other Cultivars

'*Midori*'. The medium green, broad, heart-shaped, glossy spathe is usually $8\frac{1}{2} \times 7\frac{1}{4}$ inches (length × width) with overlapping lobes. The reclining spadix is medium green and approximately $4\frac{3}{4}$ inches long. The plant is a vigorous grower with short internodes. Its cut flower has an outstanding vase life. It appears tolerant to anthracnose.

'*Asahi*'. The spathe is white with red veins and a distinct red border. The heart-shaped spathe is usually $6\frac{1}{2} \times 5\frac{1}{2}$ inches (length × width) with overlapping lobes. It is tolerant to anthracnose but is highly susceptible to bacterial blight. This cultivar was originated by Raymond Hayashi and released in the early 1980s (Figure 7).

BIOTECHNOLOGY-ASSISTED ANTHURIUM ENGINEERING

To help meet the demands of the global market for new anthurium cultivars and for high-quality flowers and pot plants, the University of Hawai'i supports an active anthurium breeding program. This research has resulted in several UH cultivars grown successfully in Hawai'i (see **COMMON CULTIVARS**).

Due to the slow growth rate of anthurium, progress in breeding can be difficult. Bacterial blight, a serious disease of anthurium, has also presented a new challenge to breeders. As a result of these two

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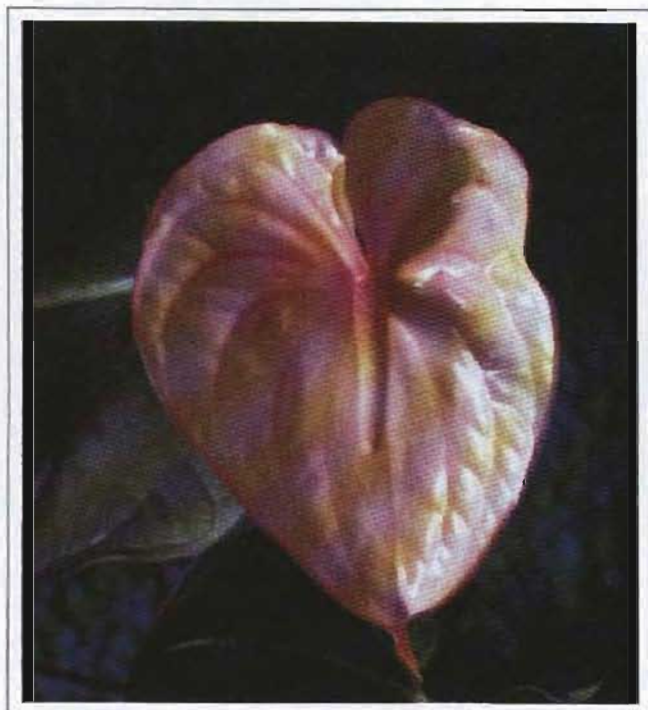
Figure 7. 'Asahi'.

bottlenecks, research using new biotechnology is now part of the anthurium breeding program. Biotechnology has allowed the development of gene transfer methods for anthurium which bypass the conventional cross-fertilization and seedling selection required in traditional crop improvement. These new methods may provide anthuriums with some resistance to bacterial blight and may also facilitate breeding of other desirable traits in anthurium, such as novel flower color.

There are several steps involved in genetic engineering of anthurium. First, genes coding for traits of interest, such as bacterial disease resistance, must be identified and isolated. This is carried out via collaboration between the breeder and molecular biologists. Next, a gene transfer system is used to introduce the new genes into the anthurium tissue. To do this, new tissue culture techniques were developed. One technique uses a "natural genetic engineer," the soil-bacterium *Agrobacterium tumefaciens*, to move the new genes into cuttings of tissue-cultured plantlets. Cuttings are then induced to produce callus and/or shoots from which new, genetically altered plantlets may be obtained.

Somatic embryos can also be used in treatments. These structures are equivalent to embryos found in a seed but are produced using leaves cultured in vitro. Af-

ter treatment with *A. tumefaciens*, somatic embryos can germinate into plantlets which can then be evaluated for the introduced genes.

A second genetic engineering technique uses microparticle bombardment to treat various tissue-cultured materials (leaves, callus, somatic embryos) with the foreign genes. Small tungsten beads coated with the DNA of the new genes are propelled into the tissues using high speeds generated by a device similar to a gun. The tissues can then be induced to produce plantlets, some of which may carry the new genes in their chromosomes.

The entire process for both techniques, from treatment to plantlet production, can take up to 2 years. These plantlets must then be integrated into the normal evaluation procedures of the anthurium breeding program to assure utility and quality before a new cultivar is released.

PROPAGATION

Cuttings

Vegetative propagation, the asexual method of propagation, ensures that the offspring will be identical with the parent. One common vegetative propagation method of increasing a particular cultivar is topping. The plant is grown until some roots have developed near the stem top. The top with these roots is then removed (topped) to produce a new plant. The remaining base of the stem with its roots will then develop two or more side shoots (suckers). By repeating this procedure, large numbers of plants may be propagated. The tendency of a plant to produce suckers is not only inherent in the cultivar but is also influenced by the plant's environment. Another method of vegetative propagation is placing mature or large stems on their side in damp propagating media to encourage the production of new shoots.

When anthurium plants are propagated by seeds and/or by tissue culture, the plantlets go through a juvenile phase followed by a generative or flower production stage. During the juvenile phase, stem internodes are extremely short, resulting in many nodes per unit length of the stem as compared to the generative stage. The topping method discussed earlier, when used on juvenile plants, results in more sucker development in most cultivars than when flowering plants are topped (Figures 8a and 8b).
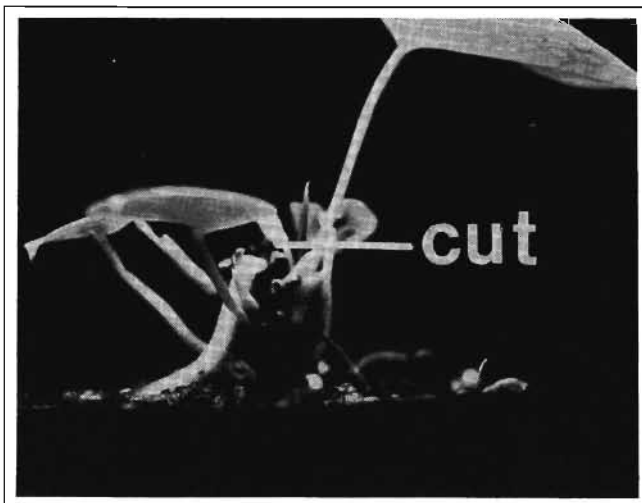
Figure 8a. Juvenile anthurium plant after topping.

Figure 8b. Topped juvenile anthurium plant with suckers.

Only disease-free plants should be used for propagation. To prevent spread of disease in propagative material, stock plants should not be harvested for flowers and strict sanitation procedures (disinfesting all tools, etc.) should be followed. Propagation areas should be isolated from production fields. The use of plastic cover and surface irrigation to keep foliage dry will help keep propagation plants disease-free.

Seeds

To grow anthurium plants from seed is a lengthy process. It may take 3 years from seed to bloom. Each new plant grown from seed will be different in some way from each parent.

An individual anthurium flower is, in reality, a small segment of the spadix. The individual flowers are hermaphroditic, with a two-carpelled ovary and four anthers. The rudimentary perianth consists of four fleshy tepals. When mature, the stigma appears as a rounded protuberance on the spadix. When these are ready to be pollinated, they are damp and shiny (Figure 9).

Controlled pollination is accomplished by grasping the pollen-laden spadix with the fingers and/or a soft brush, then transferring the pollen to the receptive stigmas of another flower by rubbing the spadix with the same fingers or brush. Once pollination and fertilization are accomplished, the spadix gradually takes on a warty appearance. After 6 to 7 months, many mature, two-carpelled, one- to two-seeded, yellow berries are formed. The yellow berries are collected and pressed lightly to squeeze one or two green seeds out of the pulp.

In planting, these seeds are scattered on finely shredded hāpu'u (Cibotium chamissoi or tree fern fibers) or other appropriate media and stored under 75 to 80 percent shade. The seeds germinate immediately and can be transplanted within 4 to 6 months. The seedlings can be expected to flower no sooner than 1½ years after seeding, and 2½ to 3 years are often required for the majority of the seedlings to come into flowering.

Tissue Culture

Bud Culture

A procedure involving the use of tissue culture technique has been developed for rapid clonal propagation of anthuriums. Aseptic cultures are initiated by (1) excising vegetative shoots of approximately $\frac{1}{13}$ inch (2 mm) at their base; (2) disinfecting the excised shoots in 0.26 percent sodium hypochlorite solution for 20 to 40 minutes; (3) rinsing the shoots in sterile water; and (4) placing the shoots in nutrient medium of Murashige and Skoog's (MS) inorganic nutrients, 15 percent (by volume) coconut water, and 2.7 oz/gal (20 g/l) (2 percent) sucrose.

When a shoot has grown into a plantlet of 0.8 to 1.5 inches (2 to 4 cm) in length, miniature cuttings of two nodes each are prepared. The basal cuttings are placed on medium of MS inorganic nutrients, 2.7×10^{-5} oz/gal (0.2 mg/l) (0.2 ppm) benzyladenine, and 2.7 oz/gal (20 g/l) (2 percent) sucrose and solidified with

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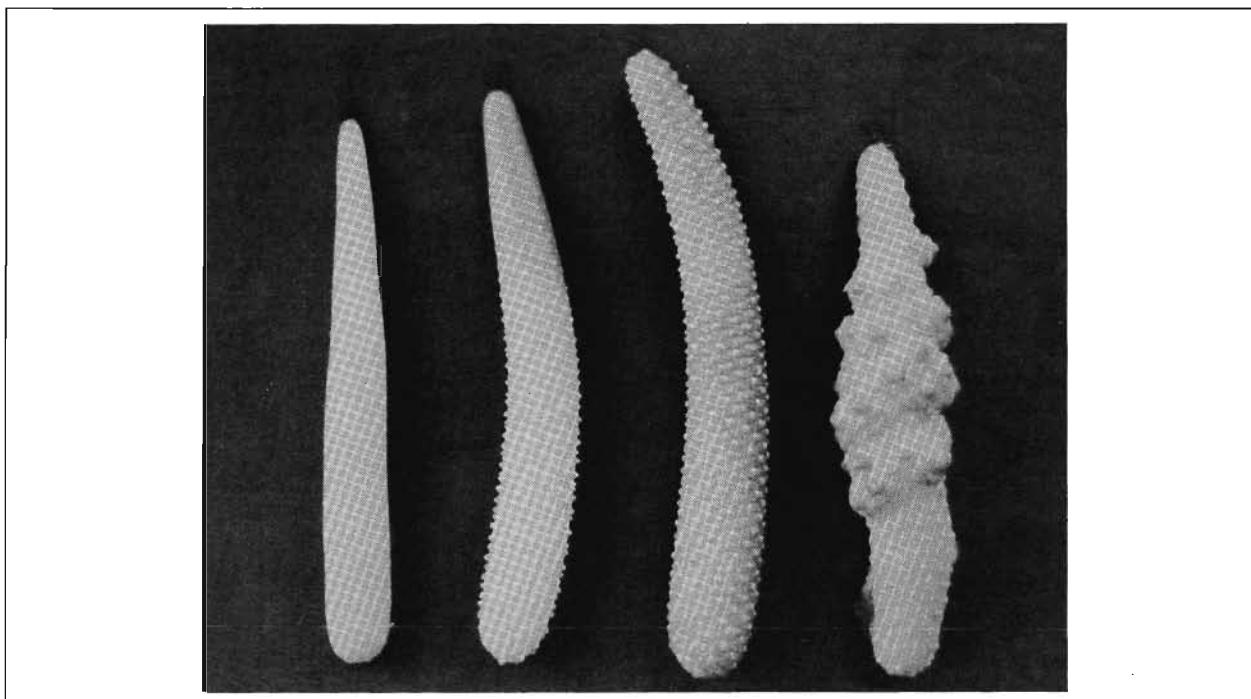
Figure 9. Left to right: Young spadix, spadix with female flowers visible, spadix with pollen visible, and spadix with seeds developing.

1.1 oz/gal (8 g/l) (0.8 percent) agar. On this medium, multiple shoots will be induced on the basal cuttings. The apical cuttings are maintained as a stock from which more basal cuttings can be obtained later.

When multiple shoots have grown into plantlets of sufficient size, they are also used as a source of basal cuttings to hasten the propagation process. Plantlets are rooted on agar medium of the same nutrient constituents as the initial culture. Rooted plantlets are then ready to be deflasked (Figure 10).

Response to nutrient salt concentration may differ by cultivar. For example, 'Marian Seefurth' grows well in 1 MS, 'Anuenue' grows poorly in 1 MS but very well in ½ MS, and 'Ozaki' grows poorly in ½ MS but well in ⅓ MS. When a cultivar grows poorly in each stage of micropropagation, check for salt toxicity.

Callus Culture and Somatic Embryogenesis

Anthuriums can also be cultured in vitro through callus culture. Callus culture involves induction of callus tissue (an unorganized mass of cells) from anthurium explants (usually petiole or leaf explant with one major vein). The explants are disinfested then placed on a modified MS medium for callus induction.

Once callus is formed, callus tissue is transferred to a modified MS shoot and root induction medium. Propagation of anthuriums through use of callus culture is more rapid than use of bud culture. However, the mutation rate may be greater for some cultivars regenerated from callus. This may make use of callus culture economically unfeasible for propagation of some cultivars. Limiting the number of in vitro cycles has been reported to minimize plant variation.

Propagation of anthuriums by use of somatic embryogenesis is being tested. Somatic embryos are thought to originate from one cell rather than from multiple cells (i.e., callus) and tend to have less genetic variation. Explants are placed on a modified MS medium for somatic embryo production. Somatic embryos are then moved onto regeneration medium to form plantlets. This method, like callus culture, is a much more rapid means of micropropagation than bud culture.

Deflasking

The ideal growing conditions (high humidity, nutrient availability, and freedom from disease) within the culture vessel produces "soft" plantlets which must be hardened or acclimatized before they can be put in

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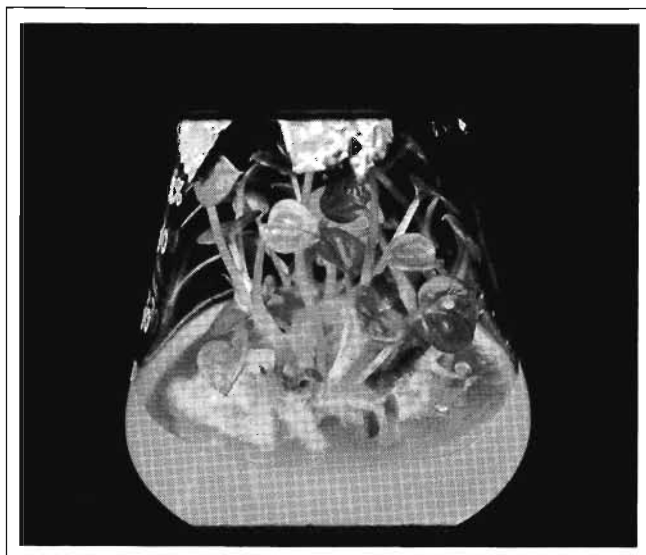
Figure 10. Tissue-cultured anthurium plantlets.

the field. Plantlets can be taken out of culture successfully by a number of methods, provided that the needs of adequate water, shade, media for root anchorage, and freedom from pests and diseases are met.

If a fogging or misting bench is available, plants may be transplanted into community pots and left on the bench for several weeks until roots become established. Community pots can also be placed in a home-made humidity box (any design to provide high humidity and adequate light) for several weeks. Commercially available plastic trays with fitted clear plastic domes are another alternative. A sheet of polyfenol foam root cubes (1 inch × 1 inch) may be used in the trays. Rockwool, peat/perlite, wood bark, or other sterile media with good water-holding capacity and drainage may be used in the trays or community pots.

The plantlets are removed from the flask by shaking the flask gently to loosen the medium. Forceps are then used to transfer plantlets from the flask to a container of clean water. If the medium is slightly soft, water can be added to the flask, the flask shaken gently, and the contents poured into a container of clean water. The medium is removed from the roots by shaking plantlets in the water. The media in tray or pots should be watered before transplanting. Plants may be placed directly into the holes of root cubes. If other media are used, a pencil or chopstick can be used to make holes (approximately 20 per 6-inch pot) and also to place plant and roots gently into the hole. Pots are

placed under mist or in a humidity box; trays are covered with a clear plastic dome. The plants may be kept in the mist, humidity box, or domed trays under 80 to 85 percent shade for several weeks or until roots are established. Once roots are established, the plants are moved out of the mist or humidity box, or the plastic dome is loosened. Plants can then be fertilized lightly with a complete fertilizer. Anthurium plantlets are easily damaged if over-fertilized. Plants at this stage are very susceptible to bacterial blight, thrips, rain damage, and slugs. Keeping plants under hard cover for additional hardening is helpful.

CULTURE AND ENVIRONMENT

Temperature
Anthuriums thrive when the night temperature is never lower than 65°F and the day temperature is about 80°F.

Shading and Protective Structures
Shading from sunlight is necessary for normal growth of anthuriums. The degree of shading varies with the cultivar, the age of the plant, and the climate under which it is grown. The shade requirements usually range from 50 to 90 percent of full sunlight. Insufficient shading often results in damage to leaves and flowers, and may cause plant death. Damage to flowers includes fading of spathes (especially pastels) and burning of spadices.

Growers use various methods to provide shade for anthuriums. Many producers use saran cloth houses supported with lumber or galvanized pipes and wire (Figure 11). Saran cloth houses have several advantages over natural shade. They provide uniform shade; permit plants to be planted immediately after construction; reduce bird, insect, and wind damage; provide maximum freedom of work space; and provide more flowers per unit area. Large houses (greater than 7 to 10 acres) can have reduced ventilation. Elevated temperatures within such houses can increase disease conditions.

Lath houses or plantings of tree fern, 8 × 8 feet apart, are also used to provide shade. Approximately 600 tree ferns are required per acre and they take approximately 1½ years to provide sufficient shade for the anthuriums. Some growers use citrus or lychee trees or a forest area cleared of underbrush for shading.

Figure 11. Anthurium saran house.

To minimize infection of the bacterial blight disease and mechanical damage due to rain, some growers are now growing anthuriums under polyethylene plastic in combination with saran shade cloth to keep rain water off the anthurium foliage.

Media

Anthuriums grow best in a well-aerated medium with good water retention capability and with good drainage. A good medium needs to be able to anchor the roots and stem so that the plant will not topple over as it grows larger, yet provide sufficient moisture, nutrients, and aeration to the plant. Organic matter (i.e., wood shavings, sugar cane bagasse, tree fern chips, taro peel, macadamia nut shells, or coffee parchment), volcanic cinder, or an artificial medium (i.e., rockwool, polyfenol foam) can serve as a good medium to anchor roots for anthurium plant growth and flower production.

Volcanic cinder is presently the most utilized medium among commercial growers because of its availability and relatively low cost. Many growers also utilize a cinder/organic media mix for moisture and nutrient retention. Growers remulch as plant height increases to provide anchorage for adventitious roots.

Spacing

The most common bed planting distance is 1 × 1-foot plantings providing approximately 25,000 plants per acre. Dense plantings decrease air circulation and hinder chemical spray penetration; leaf pruning and spraying schedules must be rigidly followed to keep disease and insect damage at a minimum. An anthurium plant may be pruned to a minimum of four leaves without any adverse effect on flower production and quality. Anthuriums can also be grown in containers such as plastic pots or bags. For 8-inch plastic pots, approximately 40,000 to 45,000 plants per acre can be cultivated.

Planting distance varies with the vigor of the cultivar, the type of shade, and the planting rotation plan of the grower. Generally, 'Kaumana' may be planted closer than 'Kozohara', which produces larger flowers. Saran cloth houses can accommodate denser planting than that of plants grown under tree ferns. Hard-covered houses can accommodate even higher densities because of improved disease control.

Replanting

The replanting schedule of mature anthurium beds depends on plant size and disease susceptibility of culti-

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vars. Beds of smaller cultivars with greater tolerance to bacterial blight may not require replanting for over 5 years. Larger cultivars, cultivars with long internodes, or those highly susceptible to bacterial blight may require a rotation schedule of 4 years or less to keep beds manageable and productive. Commercial growers generally schedule replanting for flower production to resume in time for the peak market demand in winter.

Fertilization

Recommended fertilization rates for optimum field production of anthuriums are 278-400-335 lb N-P-K/A/yr (278-917-404 lb $N-P_2O_5-K_2O$/A/yr). Granular fertilizers such as 5-10-10, 10-20-20, or 16-16-16 may be applied at the rate of approximately 300 lb N/A/yr. However, care must be taken to water the plants after application of fertilizer because the fertilizer has a tendency to burn foliage and roots. The pelletized and slow-release forms are less damaging, less labor intensive, and more often used. Many growers use organic fertilizers such as chicken manure or tankage. Annually, approximately 2000 to 3000 pounds of organic fertilizers are applied per acre with applications made every other month or quarterly. Special anthurium fertilizers are usually combinations of organic and inorganic chemicals. They should be applied as recommended on the container.

Liquid fertilizers may be applied as foliar applications or incorporated at lower concentrations into surface irrigation water, especially when plants are grown under plastic cover.

To prevent a color breakdown in spathes caused by Ca deficiency, 400 lb/A/yr of Ca is recommended. Ca can be applied as calcium carbonate (1000 lb/A/yr) or dolomite (1840 lb/A/yr), divided into at least two equal applications. Dolomite has the advantage of also supplying Mg to plants. Liquid fertilizers such as calcium nitrate can also be used to apply Ca.

Crop logging or sampling of elemental levels in foliar tissue is recommended to monitor effectiveness of fertilization programs. Leaves subtending a three-fourths matured flower are used as the index tissue. Leaf tissue levels of major elements associated with maximum flower production are 1.87 percent N, 0.15 to 0.19 percent P, 2.07 percent K, and 1.50 percent Ca.

Irrigation

Irrigation is needed for optimum production, especially during dry periods, if anthuriums are grown in a porous or well-drained medium. To minimize spread of disease, surface irrigation is best.

PESTS AND DISEASES

Insects and Mites

False Spider Mites

The red and black flat mite, commonly known as the false spider mite, *Brevipalpus phoenicis* (Geijskes) is the major mite pest on anthuriums. Plant injury is characterized by bronzing of the petiole-spathe junction and lower surface of flowers and leaves (See HITAHR Brief No. 073, Figure 8a). Life stages include egg, larva, nymph, and adult. The eggs are oval, bright red, and are usually found on both leaf surfaces. The larvae are about $\frac{1}{200}$ inch long, bright red, and have six legs. Nymphs have eight legs and are larger than the larvae. Adult flat mites are red and about $\frac{1}{100}$ inch long. The developmental time from egg to adult is about 29 days. False spider mites are widely distributed, occurring on all major continents. Other hosts include allamanda, azalea, canna, chrysanthemum, coffee, citrus, daisy, guava, hibiscus, mango, orchid, papaya, and passion fruit. See HITAHR Brief No. 082 and Hara et al. (1990) for additional information.

Pest Management. Apply two to three applications of an effective, registered miticide at 2-week intervals. Sprays should be directed to the underside of leaves and flowers.

Anthurium Thrips

The most serious pest of anthuriums is the anthurium thrips, *Chaetanaphothrips orchidii* (Moulton). In severe infestations, anthurium thrips can damage every flower produced in infested shade houses. Anthurium thrips invade the unopened bud soon after the bud emerges from the stipules. The mature flower is deformed with white streaks and/or scarring on upper and/or lower surfaces of the flower and is unmarketable (See HITAHR Brief No. 073, Figure 5a). Life stages include egg, two larval instars, prepupa, pupa, and adult. Eggs are oviposited into the surface of the spathe and hatch in about 8 days. The emerging nymphs are yellow and feed inside the developing bud. Second instar nymphs migrate off the plant and pupate in the media and plant debris below. Adults are

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about $\frac{1}{25}$ inch long and are yellow with banded wings. The developmental time from egg to adult is about 27 days. Anthurium thrips are also found in South America, Europe, Japan, Puerto Rico, and Florida. Alternate hosts include alternanthera, asystasia, bougainvillea, chrysanthemum, corn, night-blooming cereus, parsley, orchids, wandering jew, and several weed and grass species. See HITAHR Brief No. 086, Hara et al. (1987, 1988, 1990), and Hata and Hara (1992b) for additional information.

Pest Management. Anthurium thrips can be effectively controlled with contact and contact-systemic insecticide sprays. An effective insecticide requires a minimum four to five insecticide spray applications at 2-week intervals or 7 to 8 weeks after initial spray application to significantly reduce thrips injury. This 7- to 8-week period is needed because flower buds already injured at the time of initial spray application are harvested 6 to 8 weeks later as injured flowers. Effective insecticides protect newly developing anthurium buds from thrips injury during the 7- to 8-week period of spray applications.

Anthurium Whitefly

The anthurium whitefly, *Aleurotulus anthuricola* (Nakahara), is endemic to Colombia and several islands in the lesser Antilles and appears host specific to anthuriums. Although the anthurium whitefly causes no obvious physical injury to anthuriums, its presence is a quarantine problem for flowers that are exported from Hawai'i to the continental United States and other uninfested areas of the world. The whitefly lives its entire cycle from egg to adult within the anthurium sheath. Plants infested with whiteflies are easily recognized by copious amounts of white, waxy secretions on the inner surface of anthurium sheaths. (See HITAHR Brief No. 073, Figure 6a). Life cycle stages include egg, several nymphal stages, pupa, and adult. Whitefly eggs are dull white to cream and are deposited in an upright position. Nymphs are nearly transparent and secrete a white, waxy material. Pupae, which are black and oval, also secrete a white, waxy material. Adult whiteflies have white wings and a yellow body and are less than $\frac{1}{25}$ inch long. The developmental time from egg to adult is about 35 days. See HITAHR Brief No. 087, Hara et al. (1988), and Hata and Hara (1992a) for additional information.

Pest Management. The anthurium whitefly is a difficult pest to control because it occurs within the leaf and flower sheaths, protected from insecticide contact. In addition, the wax secreted by the whitefly prevents contact insecticide penetration. Apply an effective, registered systemic insecticide to the developing floral area. A minimum of four insecticide applications must be applied at 2-week intervals to bring the whitefly under control.

Black Twig Borer

The black twig borer, *Xylosandrus compactus* (Eichoff), bores into leaf and flower petioles. The affected leaf is chlorotic and has a necrotic segment where the borer entered (See HITAHR Brief No. 089, Figures 1 and 2). Black twig borers may also bore into the stem, resulting in death of the plant. The black twig borer completes its life cycle from egg to adult within the petiole. Life cycle stages include egg, larva, pupa, and adult. Eggs are oval and white and are laid on the ambrosia fungus cultivated by the female beetle. Larvae are white, legless grubs with distinct head capsules. The pupae are white, changing to brown as they approach maturity. Adults are shiny black and are about $\frac{1}{16}$ inch long. The black twig borer is widely distributed and occurs in Japan, Indonesia, Vietnam, Malasia, Sri Lanka, India, Madagascar, Mauritius, Seychelles, tropical Africa, Fiji, Florida, Georgia, Alabama, Louisiana, and Hawai'i. Black twig borers have been reported to attack over 100 species of plants in 44 families, including avocado, citrus, cacao, coffee, hibiscus, lychee, macadamia, orchids, pikake, and red ginger. See HITAHR Brief No. 089, Hara and Beardsley (1979), and Hata and Hara (1989) for additional information.

Pest Management. An effective method of controlling the black twig borer is field sanitation along with an effective insecticide program. The infected petioles should be removed from the field and destroyed by burning or burying. Infected petioles remaining in the field may remain active resulting in reestablishment of this pest.

Phytotoxicity

A limiting factor in using certain insecticides and acaricides on anthurium is phytotoxicity, or plant injuries resulting from pesticide applications. Phytotoxic responses differ among anthurium cultivars. An insecticide that is safe on one cultivar may not be safe on

another. Injury symptoms from contact insecticides appear more quickly than those from systemic insecticides. Phytotoxic contact insecticides directly damage buds and leaves, whereas most phytotoxic systemic insecticides cause injury to the primordia which produce observable damage only to flower and leaves that emerge several weeks after the application. In addition, each grower has unique growing conditions. It is highly recommended that the grower first test the pesticide on a small group of plants before treating the entire crop. Two to three applications should be made at 5- to 7-day intervals, allowing 2 to 3 weeks after the last application to observe for phytotoxicity. When testing systemic insecticides, the waiting period should be extended 4 to 6 weeks. See HITAHR Brief No. 097 for additional information.

Consult your local cooperative extension service or the Hawai'i Department of Agriculture for authorized special local need registrations or additional information.

Diseases

Anthurium Blight

Anthurium blight is caused by the bacterium *Xanthomonas campestris* pv. dieffenbachiae. It was first reported on anthuriums in Brazil in 1960. It was reported for the first time in Hawai'i on Kaua'i in 1971, then on Hawai'i and Maui in 1980. There is evidence that it was present on O'ahu for about a year be-

fore its discovery on Hawai'i. It has also been reported in Florida and California in the continental United States and most anthurium-producing countries throughout the world. It is uncertain whether it is present in the Netherlands.

Symptoms. Non-systemic foliar symptoms usually begin along the margins of leaves where hydathodes are located. The first symptoms are small, scattered, irregularly shaped, water-soaked spots that are more pronounced on the underside of leaves (Figure 12). The tissue surrounding the spots eventually turns yellow and dies. Usually, a bright yellow band separates the dead from the live tissue (Figure 13).

Once systemic, the bacterium spreads quickly throughout the entire plant. The bacterium causes leaves closest to the point of entry into the stem to turn a dull yellow as the bacterium clogs the vascular system and prevents the translocation of water and nutrients. Cultivars vary in susceptibility to the systemic phase. Therefore, symptoms and the rate of symptom progression may differ. A characteristic symptom of advanced systemic infection is the discoloration of the vascular system. The petioles of plants with advanced infection are often easily removed as a result of the formation of the abscission layer and the discolored vascular bundles are readily seen as scattered brown spots (Figure 14). Longitudinally cut stems or petioles may have brown streaks formed by the discolored vascular system.

Biology. The origin of the bacterium is unknown.

Figure 12. Early foliar symptoms of anthurium bacterial blight.

Figure 13. Advanced foliar symptoms of anthurium bacterial blight.

The bacterium has been found on all plants belonging to the family Araceae that were tested. Virulence of the bacterium isolates obtained from these plant species vary considerably from the strain that causes anthurium blight. The bacterium is suspected of being able to infect plants without exhibiting visible symptoms.

The bacterium cannot survive outside the plant for long periods. In soil, a maximum of about 5 to 6 weeks has been demonstrated.

The bacterium is spread in several ways. The most important means of plant to plant spread are: splashing rain or irrigation water; contaminated cutting tools used for harvesting, leaf pruning, and propagation; planting infected materials; people walking through and brushing against infected plants with wet clothing and rain wear; and movement of infested soil on footwear, vehicles, tools, and other equipment. The spread of the bacterium in small water droplets (aerosols) probably occurs but is not considered a major means of spread.

Management. Anthurium blight is a very difficult disease to control because of the systemic and highly contagious nature of the disease. If a grower is fortunate enough not to have the disease, preventing its introduction onto the farm through propagation material, personnel, tools, and equipment is most important.

If the disease is already present on a farm, every effort must be made to eradicate the disease or to contain the disease and prevent its spread to other fields. This involves removing early foliar infection and roguing systemically infected plants. Because of the difficulty in assessing whether an infection is systemic or not, it is best to rogue all infected plants. Rogued plants should be bagged, burned, or buried to prevent them from being a source of inoculum for further spread.

Replanting is ideally done by sections of a large house or entire small houses. For smaller growers, the minimum area for replanting is a bed. Replanting by sections is more manageable in implementing control measures and will further isolate the new plantings from active infection foci. Since the bacterium does not survive outside the host for long periods, a 2-month fallow after removing all plant parts from beds is effective in eliminating the bacterium. Instead of fallowing, beds may be fumigated with metam-sodium and tarped. In either method, increasing the activity of beneficial microorganisms in the beds during fallow or after fumigation is desired.

Planting disease-free material is essential. Planting material must be produced under strict disease-free conditions on benches. They should also be under cover, protected from rain, drip or surface irrigated, and always isolated from production fields. Bud- or tissue-cultured plants are currently the most reliable source of clean planting material. However, they may
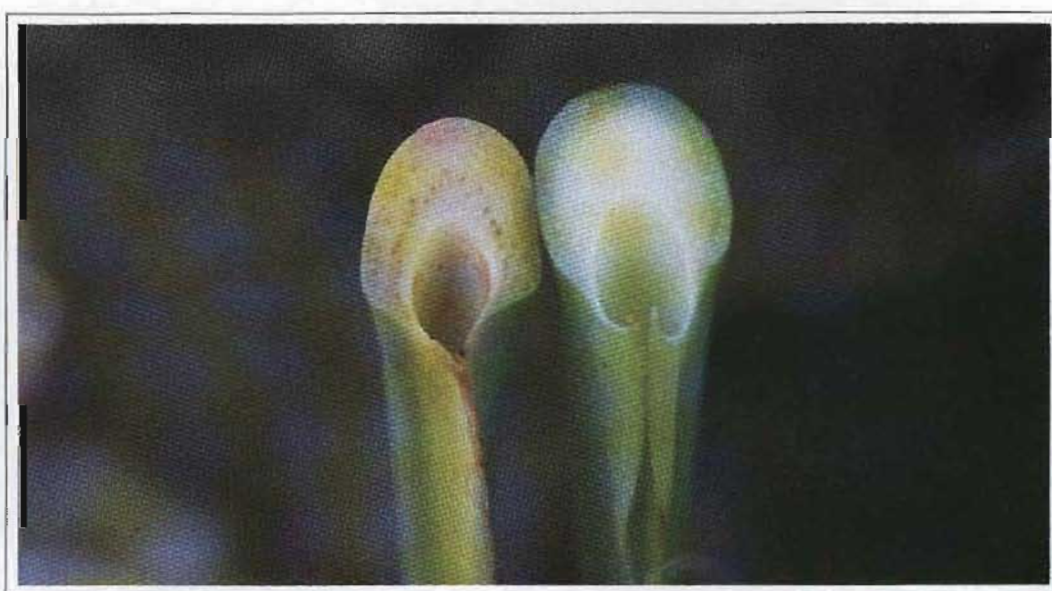

Figure 14. Discolored vascular bundles caused by anthurium bacterial blight.

also become infected if they are not grown in disease-free locations under strict quarantine after removal from flasks.

Any measure that minimizes the wetting of foliage of production fields should reduce the spread of the disease. Overhead cover with surface irrigation has been shown to reduce disease incidence.

Antibiotics are not recommended as a routine part of a control program. This is because streptomycin and oxytetracycline are incapable of killing the bacterium under normal use rates and resistance to streptomycin is a serious problem. The label directions should be closely followed and the field "sanitized" prior to each application. The bacterial population should be closely monitored for antibiotic resistance.

Since the bacterium can be readily spread from plant to plant on contaminated harvesting shears, shears should be disinfested as frequently as possible. To obtain quick (5 seconds) kill, disinfestants must be used at a high concentration. Currently, 50 percent strength household bleach (2.6 percent NaOCl), 10 percent active ingredient solution of quaternary ammonium compounds (e.g., Physan™), and LD™ (a chlorine-dioxide-based material) are recommended. Working with and alternating a minimum of two tools

lengthens the exposure to the disinfestant and increases the probability of killing the bacterium on the blade surface.

Anthracnose

Anthracnose (black-nose, spadix rot) is caused by the fungus *Colletotrichum gloeosporioides* and is a common cause for flower rejection in Hawai'i. The fungus is common on numerous ornamental, fruit, and vegetable crops grown in tropical and subtropical areas. Many specialized strains that differ in host range are suspected to be included in this large species group.

Symptoms. The disease primarily affects the individual flowers on the spadix. Infection starts as a tiny, dark spot that expands to a triangular or other angular shape depending on the number and pattern of tepals infected (Figure 15). Each infection site usually remains isolated, is surrounded by adjacent healthy tissue, and may be scattered individually or in narrow or broad zones. Under wet and warm conditions with a high level of inoculum, a general rot of the entire spadix may occur.

The fungus can infect leaves following an injury. Petioles and pedicels are also susceptible and develop elongated, diamond-shaped lesions.
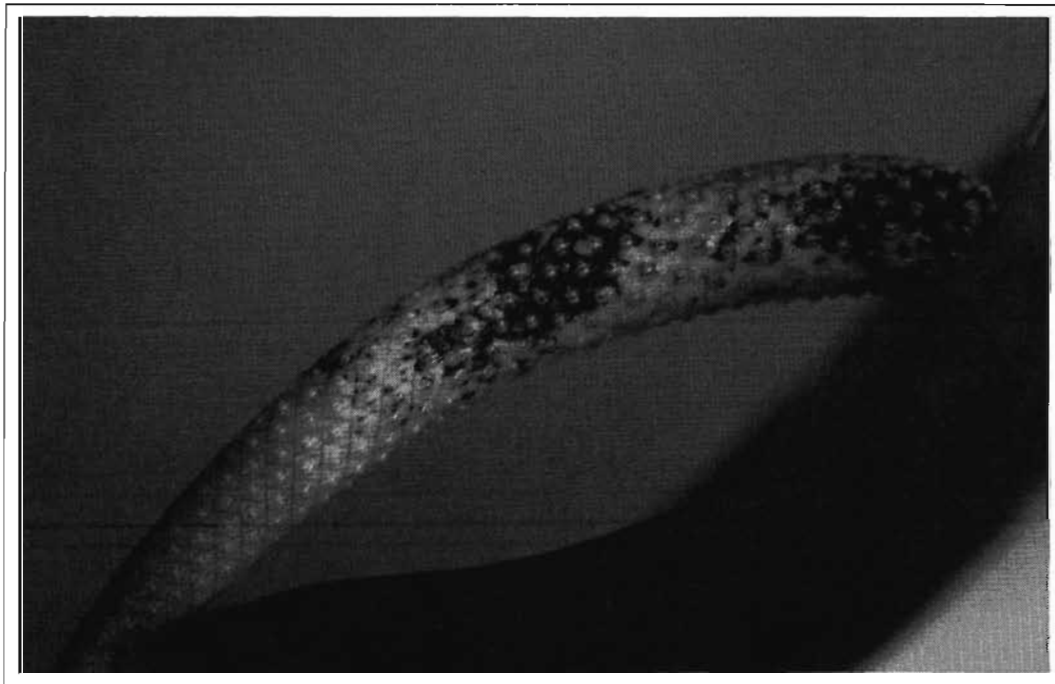
Figure 15. Anthurium anthracnose or black-nose.

Biology. Although cross-inoculation between anthurium and papaya isolates of the pathogen has been demonstrated, the major source of spores is infected anthurium flowers from within the field. Moisture and temperature have a major influence on disease incidence. Disease incidence is lowest between November and May. It begins to increase during the summer months and peaks in September and October, traditionally the warmest months of the year. Anthuriums grown at lower elevations have higher anthracnose incidence than those grown at higher elevations because of higher temperatures.

Spores are produced in large numbers and are spread primarily by splashing rain and irrigation water. Insects, mites, and other organisms are potential carriers of the spores but are not a major factor in transmission.

Management. Many of the older commercial cultivars are susceptible to anthracnose and require periodic use of a fungicide. Most cultivars released by the University of Hawai'i are selected for resistance to anthracnose and, therefore, should require little effort to control this disease. Since anthracnose incidence is influenced by temperature, it should be monitored from about May through October. Recommended fungicides should be sprayed when the disease level reaches the economic threshold level.

Anthurium Decline

Anthurium decline is caused by the burrowing nematode, *Radopholus similis*, a microscopic round worm. The burrowing nematode is about $3/100$ inch long and is known to feed on the roots of more than 300 plant species. The burrowing nematode is found on all the islands of Hawai'i and in many tropical and subtropical areas.

Symptoms. Plants affected by burrowing nematodes show signs of nutrient deficiency, stunting, fewer and smaller leaves and flowers, premature leaf yellowing, and overall poor plant vigor. Roots of infected plants have few to many dark necrotic lesions. In the advanced stages of the disease, the entire root system may be deteriorated as a result of secondary invading microorganisms. Although it is often difficult to differentiate between burrowing nematode damage and root rot, the key symptom of burrowing nematode infection is the dark lesions on the stem below and especially above ground, sometimes extending up to the apical part of the plant. Examination of these lesions under a dissecting or compound microscope should reveal numerous burrowing nematodes.

Biology. The nematode enters the root by puncturing and breaking the wall of root cells. As they feed on the root cells, the lesion enlarges. Eggs are laid, hatched, and large colonies containing nematodes of all growth stages develop. The cortices of infected roots are often severed causing large sections of the roots to die. Infected areas are then invaded by opportunistic fungi and bacteria. The nematode requires living host tissue as a food source but can survive outside of roots for about 4 to 6 months. The life cycle from egg to egg is about 21 days depending on environmental conditions.

A population of burrowing nematodes from anthurium was recently characterized as having five chromosomes and capable of infecting sour orange rootstock, thus possibly identifying it as belonging to the citrus race. The citrus race is most noted as the cause of the serious "slow decline" disease of citrus in Florida. The strict quarantine restrictions on rooted plants from Hawai'i and Florida to California is due to the burrowing nematode.

The burrowing nematode is most commonly spread through infected planting materials. Infested media carried on footwear, tools, and equipment are also important means of transmission. Although nematodes are capable of moving only a few inches a year on their own power, they can be spread long distances through surface or subsurface movement of irrigation or rain water.

Management. The most important control measure is to start with nematode-free planting material and media. Every effort should then be directed toward preventing the introduction of nematodes into the planted area, with special emphasis on vehicle tires, equipment and tools, footwear, and flow of runoff water into the planted area.

Planting material can be treated with hot water at 121.5° F (50°C) for 10 minutes to control nematodes. Although effective, precise temperature control is required and treatment of large quantities is costly. Plant tissue damage is likely to occur and differences in heat sensitivity among cultivars may be a problem.

Post-plant nematicides, such as fenamiphos, are effective in reducing nematode populations but should not be used indiscriminately. Testing of media and plants for burrowing nematodes should be done periodically. Existing populations can be used as an indicator

for timing of pesticide applications.

Root Rot

Anthurium root rot is common with anthuriums planted in beds as well as in containers. Numerous fungi are recovered from dead and dying roots including: *Pythium splendens*, *Calonectria crotalariae*, *Rhizoctonia* sp., *Phytophthora* sp., *Pythium* spp., and *Fusarium* sp. None of these fungi, in the absence of one or more external factors, has been shown to be a primary cause of root rots.

Symptoms. Symptoms of root rot include reduced plant height, smaller leaves and flowers, lack of leaf and flower sheen, and a general lack of vigor. Plants have varying degrees of root mortality. In severe cases, all the roots, except for a few aerial roots that have not yet begun growth into the medium, may be rotted. Roots often have a strong odor of decay as a result of secondary invasion by bacteria.

Biology. Roots are prone to rot if drainage is poor. Common causes of poor drainage are: old decomposed media, media that retain excessive moisture, shallow depth of media over a solid substrate such as pāhoehoe lava, and failure to shake off old medium from roots of containerized plants when transplanting into a different medium. Tissue-cultured plants are usually grown in a soil-less medium consisting of peatmoss, vermiculite, and perlite. If these plants are transplanted into 100 percent volcanic cinders without first shaking off the old medium, the difference in texture between the two media can result in excess moisture retention by the old medium and can eventually lead to root rot. Roots are also predisposed to decay as a result of fertilizer damage, interactions with excessive amounts of pesticides, and burrowing nematode damage.

Management. Make sure that nematodes are not the cause of root rot. A thorough evaluation of all cultural practices needs to be made. Ensure that drainage is good and that the medium does not retain excessive moisture. Avoid using only resin-coated, slow-release fertilizers because of differential release rates and the tendency of nutrients to accumulate in the medium during hot, dry periods with reduced rainfall. Monitor pH levels of the medium and apply Ca as necessary. A number of fungicides are registered for root rot control on anthuriums and can be used judiciously. However, the most effective control of root rots is usually carried out through changes in cultural practices.

Anthurium Bleach

The anthurium bleach problem was first observed during the last week of September 1981. The symptoms, however, were identical to an earlier incidence in September 1980 involving an undetermined cause, which was limited to a single grower. Within a month, 30 percent of the anthurium acreage under shade cloth became affected. Affected growers experienced a 20 to 60 percent reduction in marketable flower production.

Symptoms. The first visible symptom is usually small, purple spots on the spathe with a small, necrotic center on the back side. Subsequent acute symptoms, in general order of appearance, include browning of leaf veins, scorching of immature leaves, and necrotic lesions on leaves, petioles, and pedicels. Plants then go into a chronic or long-term symptom stage that is characterized by severe chlorosis and stunting of new leaves (Figure 16). These leaves have a leathery, stiff texture. Spathes and spadixes do not develop their normal color and appear faded, hence, the name "bleaching" (Figure 17). Other symptoms are root tip necrosis and general browning of the root system, browning of the internal parts of the main stem, suppressed lateral root development, suppressed rooting of stem cuttings, and, with severely affected plants, necrosis of the apical meristem. Chronic symptoms generally last for 3 to 6 months or sometimes longer.

Suspected cause. The cause of the bleaching problem has not been positively determined because of in-

Figure 16. Chlorosis and stunting of new leaves due to anthurium bleach.

Figure 17. "Bleaching" of anthurium flowers.

ability to reproduce symptoms under controlled conditions. However, it appears to be a physiological disorder brought on by the high and continuous fertilization with a predominantly ammonium form of N during unusually hot and dry periods. Bleaching is thought to be intensified by factors such as the use of high rates of resin-coated, slow-release fertilizers that release at faster rates as temperature increases. For example, a 5- to 6-month formulation of a popular brand released 45 percent of its N after 3 months at 70°F but released 90 percent of its N at 90°F. Other factors suspected of intensifying bleaching are a low cation exchange capacity of the planting medium and the use of overhead sprinklers during hot, dry weather to wet only the root zone. This is suspected to cause the accumulation of fertilizer in the root zone without periodic leaching. The initial acute symptoms were usually precipitated by the return of normal, cool, wet weather following a prolonged dry period.

Under normal weather conditions, excess fertilizer is leached out of the root zone by the abundant rainfall. However, the rainfall in the anthurium-producing areas was considerably below normal during the period preceding the outbreak of anthurium bleach in 1981. The volcanic cinder medium does not sustain a good population of nitrifying bacteria, which are important for the conversion of ammonium N to nitrate.

Soil and tissue analyses have not shown consistent, significant differences that might help determine a cause. Although total leaf N from bleached and nor-

mal plants are not significantly different, the levels of nitrate N have usually been significantly lower and Mn higher in bleached leaves than healthy leaves.

Management. Select a fertilizer program where nitrate N makes up a minimum of 50 percent of the total N requirement. Do not rely on a single type of fertilizer for all of the anthurium nutrient requirements. The fertilizer requirement should be met with a combination of resin-coated, slow-release fertilizer (for the rainy season), and a low-analysis, organic fertilizer, such as processed manures, and supplemented with liquid fertilizers when insecticides or fungicides are applied.

Although weather and rainfall cannot be controlled, shade houses should be designed to minimize internal heat buildup. Shade house size, orientation, and ventilation should be considered.

Other Pests

Snails and Slugs

Snails and slugs feed on leaves of deflasked, tissue-cultured anthurium plantlets. Commercially available bait formulations for snails and slugs can be spread on the ground around plants or benches.

Birds

The mejiro bird and other birds nip the unopened upper tip of the spathe during some seasons of the year. Bird nets to cover open areas of saran can help control damage. Bright or colorful objects such as tin pie plates or strips of colored plastic hung in the vicinity of the planting will help scare these birds away.

INTEGRATED PEST MANAGEMENT (IPM)

Integrated Pest Management, or IPM is a holistic approach to pest control that can help reduce pest damage to economically acceptable levels by maximizing the use of nonchemical control tactics and minimizing the use of toxic pesticides. Nonchemical control tactics include cultural control (resistant varieties, sanitation, pruning, use of protective structures), mechanical control (exclusion by barriers, trapping), environmental control, biological control, and regulatory control.

IPM requires regular observations in the field, checks on plant growth and health, and monitoring of pest damage. Farm workers should be trained to detect early signs and symptoms of the most common prob-

lems. Early detection of problems and keeping records of problems enable the grower to take necessary precautions before the problem becomes serious. This is the key to a successful IPM program. By knowing the patterns of damage over a number of years, growers can determine trends that will help them prepare for similar problems in the future. For example, thrips injury fluctuates depending on rainfall and temperature, but generally increases during the summer and decreases during the winter. Knowing the danger periods can help the grower to determine the times when spraying may be necessary.

Use of sprays may be warranted when no other available controls are feasible or when the pest population passes the predetermined economic injury level for the crop (the level of pest damage at which pesticide application is economically justified). The economic injury level for anthuriums can be determined by comparing pesticide application costs and value of pest injury using the worksheet in Table 2.

Weeds

Rout (EPA REG. No. 58185-27), is a 3 percent granular formulation of two preemergence herbicides, oxyfluorfen (2 percent) and oryzalin (1 percent). This combination provides for a very wide spectrum of preemergence weed control including common moss species (see the product label for a complete list of weeds controlled). This product will not, however, kill established weeds. An ingredient in Rout can cause vapor injury to succulent tissues. Rout should not be used near plant materials newly removed from tissue culture containers. Avoid contact with leaf petiole attachments and flowering parts. Rout should be applied with granular spreaders that allow for placement at the base of plants. In enclosed structures, Rout should never be applied to more than one-fourth of the production area. Consideration should be given to providing optimum ventilation to areas treated with Rout to avoid injury due to vapors.

Apply the recommended rates of Rout into weed-free beds to prevent weed establishment. In new beds, planting materials should consist only of vegetative stem pieces with a minimum amount of soft, succulent tissue. At recommended rates, Rout will provide up to 3 months of weed control.

The herbicide rate listed in Table 3 was developed on the following anthurium cultivars: 'Ozaki', 'Nitta',

'Kozohara', and 'Marian Seefurth'. Research results indicated that no yield reductions were recorded for any cultivar. However, slight reductions in flower size and stem length were recorded where Rout applications were made. Rout should be applied with a minimum of 3 months between applications.

Water anthuriums immediately after herbicide application to remove chemicals from leaf surfaces.

Woven polypropylene (black) ground cover provides excellent broad-spectrum weed control with no effect on yield. Care should be taken to distribute granular or slow-release fertilizers evenly under the mulch to prevent burning of roots.

POSTHARVEST

Vase life of anthurium varies widely. For example, 'Ozaki' vase life can vary from 8 to 68 days. The average of most cultivars is 15 to 20 days. Mean maximum temperature during the 2 months before harvest and vase life is positively related. Higher growing temperature yields flowers with longer vase life. High-N fertilizer rates reduces flower vase life. This reduction is alleviated by K fertilizer. P fertilization did not influence cut flower vase life. Optimum fertilizer rate for maximum vase life was 0 lb/A/yr N; 200 and 400 lb/A/yr K are equally effective. Preharvest factors influence 63 to 71 percent of the variation in postharvest vase life of anthuriums.

Harvesting

Anthuriums are usually harvested once per week. After harvest, the flower stem should be placed into a clean bucket filled with 4 to 6 inches of clean tap water. The water in the buckets should be changed regularly, at least every day after 2 days of use. Buckets should be cleaned with a disinfectant at least once a week to remove any microbial growth on the inside. The cheapest disinfectant is bleach. However, it must be freshly prepared as it is unstable. Commercial disinfectants are more stable and more effective. Rinse buckets with clean water before use.

The maturity of the flowers for harvesting is determined by firmness of the peduncle and the degree of color change of the spadix. The botanical flowers mature on the spadix from the base towards the apex. As they mature, a change in color can be discerned that moves from the base to the tip of the spadix within a period of 3 to 4 weeks. After the lower half of the spa-

Table 2 . Worksheet for determining economic injury levels (EIL) for pests of anthurium. The example given is for thrips at the economic injury level of 5.28 percent (modified from Stuckey et al. 1984).

1.	Anticipated returns without thrips injury	
	a. Estimated yield per acre per mo (doz)	694.00
	b. Expected market price per doz	$6.00
	c. Gross returns per acre (1a × 1b)	$4,164.00
2.	Anticipated loss damages without treatment	
	a. Estimated percent yield loss	5.28%
	b. Dozens loss per acre per mo (1a × 2a)	36.64
	c. Dollar loss per acre (1b × 2b)	$219.84
3.	Cost of control treatment	
	a. Total cost of pesticide per application at 200 gal/A (Malathion 5 EC, 2.5 pts/100 gal)	$14.38
	b. Cost of pesticide application per acre (labor, fringe benefits, equipment maintenance)	$30.17
	c. Cost of treatment per acre (3a + 3b)	$44.55
	d. Total number of applications required to control pest	4
	e. Total cost of treatment per acre (3c × 3d)	$178.20
4.	Anticipated loss even with treatment	
	a. Estimated percent yield loss with treatment	1%
	b. Dozen yield loss per acre treated (yield/A [694] × 4a)	6.94
	c. Dollar loss per acre ($ per doz × 4b)	$41.64
5.	Determining economic justification for pesticide application	
	a. Per acre value of increased yield from pesticide application [anticipated loss damages without treatment (2c) minus anticipated loss even with treatment (4c)]	$178.20
	b. Net returns [increased yield (5a) minus cost of control treatment (3e)]	0.00

Table 3. Recommended herbicide rate for anthurium cinder beds

Growth Medium	Herbicide Formulation	Amount of Formulation per 1000 ft^2 (lb)	Amount of Formulation per Acre (lb)	Active Ingredient per Acre (lb)
volcanic cinders	Rout	2.3	100	3.0

dix has changed color, the anthurium is referred to as one-half mature. Flower maturity affects postharvest longevity. Fully mature flowers last 10 percent longer than half-open flowers. Maximum vase life occurs when anthuriums are harvested with $\frac{3}{4}$ or more of the flowers on the spadix open. Anthurium flowers should be harvested at this stage.

Handling

Anthurium vase life is probably limited by plugging of the stem water-conducting tissue. There is conflicting evidence as to whether it is bacterial or physiological blockage of the water conducting tissue that reduces anthurium vase life. Both causes can be reduced by proper postharvest handling. The use of clean buckets and clean water to hold cut flowers is an essential first step.

Two postharvest treatments have been found to increase postharvest life: (1) treating the recently cut stem with 1000 ppm silver nitrate for 10 to 20 minutes, or (2) waxing the flower with wax formulations. However, only a few wax formulations are effective.

Ethylene is a low molecular weight gas that is notorious for reducing the quality of floral crops. In anthuriums, it causes spathe blueing and shedding. The vase life of anthuriums can be cut almost in half by a short exposure to ethylene. Ethylene sources include ripening fruit, diseased and injured plants, and internal combustion engines. Exposure to ethylene should be avoided.

Anthuriums should be packed carefully to reduce the risk of injury during shipping. Mechanical injury due to creasing of the spathe or bruising from the spadix touching the spathe during shipping are major problems. These injuries lead to unsightly black marks and downgrading of flowers.

Anthurium flowers should not be held in the packed condition during shipping for more than 4 days. During shipping, temperature extremes can cause loss of flowers. Flowers should not be exposed to temperatures less than 50°F for more than 1 day. Chilling injury occurs below these temperatures. In anthuriums, it is expressed as darkening of the flower spathe and spadix.

Commercial preservative should be recommended for the retailer and consumer. These solutions assist in reducing microbial growth and therefore contribute to the maintenance of water uptake. Submerging the flowers in water or misting regularly to revive wilted flowers are also useful recommendations.

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