AN APPROACH TO THE CONTROL OF *PHYTOPHTHORA CINNAMOMI*

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Phytophthora cinnamomi has been isolated from plants found throughout the temperate and tropical regions of the world and has been described as "one of the world's major plant killers". It has severely restricted the numbers of *Castanea* species in large areas of the USA and is largely responsible for the recession of the eucalyptus forests in Australasia. It is surprising, therefore, to find that it was first described (on cinnamon) as late as 1922. The development of this disease in the hardy nursery stock industry of Western Europe escalated after the second World War until the disease was endemic in many areas. Throughout the United Kingdom, and especially certain areas of England, the incidence of the disease early in the 1970's reached alarming proportions.

In Northern Ireland in 1974 serious losses of hardy nursery stock caused concern and a survey was carried out on all nurseries where the disease had been isolated. The results were disconcerting, indicating that as much as 10% of all nursery stock was infected. Levels well above this were recorded on three of our largest nurseries, one of which had only been established late in the 1960's.

This serious situation was approached by pooling the resources of the advisory, experimental, and scientific departments of the Dept. of Agriculture (N. Ireland). The problem was identified as one of curtailing the spread of the disease, mainly in container-grown plants. Open ground production accounted for only 20% of total plant numbers and was in decline. A decision was taken to study the epidemiology of the disease with the object of identifying possible control measures.

Although many species of *Phytophthora* were identified in the survey, the most persistent and destructive was *Phytophthora cinnamomi*. *Phytophthora cinnamomi* has both the persistent spores (chlamydospores and occasionally oospores) as well as the minute motile zoospores. It thrives in moist soils, water being essential for the transportation of the motile zoospores and for their subsequent infection of roots. The life cycle can be seen in Figure 1.

Under suitable conditions of warmth and moisture sporangia are produced on slender stalks (sporangiophores) protruding from the surface of infected roots. The precise trigger for sporangia is

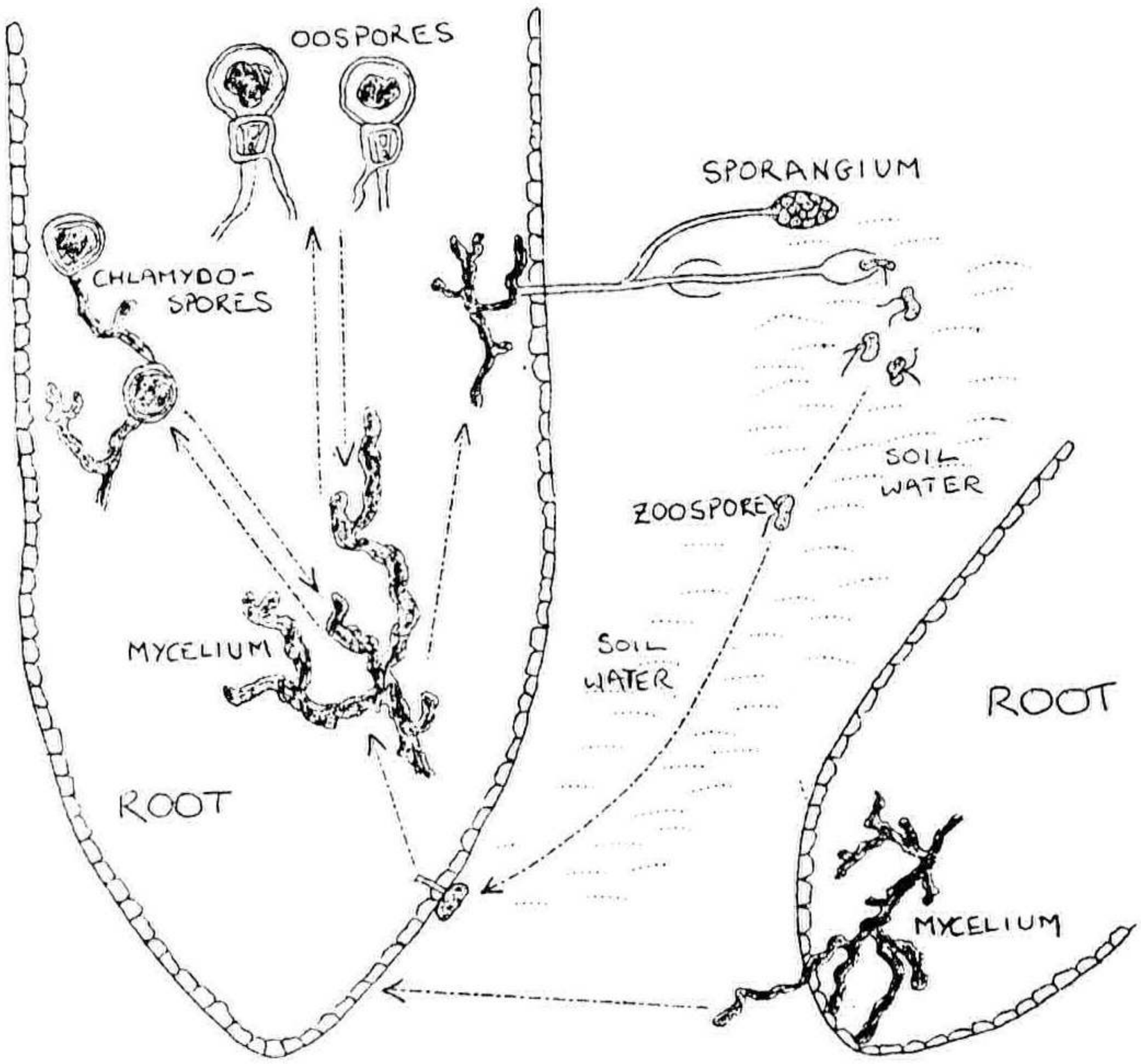

Figure 1. Life cycle of *Phytophthora cinnamomi.*

unknown but would appear to be related to biological activity (probably bacteria). These sporangia release small swimming spores (biflagellate zoospores). Although zoospores may be synchronistically released under laboratory conditions by means of a low temperature shock this probably does not occur in nature. Zoospores are chemically attracted towards plant roots but require a film of moisture as a vehicle of transport. Once formed they remain viable for only a short time and although motile are unable to travel against the flow of drainage water.

When infection has occurred the fungus spreads throughout the plant root system by means of feeding mycelium. The fungus eventually kills the plant by mechanically blocking the vessels which transport water and minerals from the roots to the above ground parts of the plant. As the pathogen is unable to live in dead tissue it must make preparation for either infection of another living plant or survival until another host plant becomes available.

In warm wet conditions infection may involve the direct penetration of adjacent roots by mycelium; or more usually by the production of further zoospores formed within a sporangium. Survival of the fungus is usually achieved in Northern Ireland by the production of chlamydospores, which can remain dormant in the dead roots for many years. Occasionally the sexual organs

(oospores) have been found. The opposite mating type of *Phytophthora cinnamomi* is not found in Northern Ireland and the oospores are produced following inducement by *Trichoderma viride* or contact with *Phytophthora cryptogea*. Both chlamydospores and oospores possess a thick protective coat which is remarkably resistant to attack by hostile agents, including toxic chemicals. These resistant resting bodies enable the fungus to survive for long periods. In the presence of suitable host roots both spore types can be stimulated to produce sporangia, mycelium, or both. Infection of roots then takes place as previously described and the life cycle continues.

Several factors are known to affect the development of *Phytophthora cinnamomi*, temperature and moisture having the greatest influence. The fungus is most active within the temperature range 20° to 30°C (68° to 86°F). In Northern Ireland these temperatures are achieved in containers from late May to the end of September. The optimum temperature for infection by motile zoospores is 25° to 26°C (77° to 79°F). It has been reported that *Phytophthora cinnamomi* makes little or no growth below 10°C (50°F) or above 34°C (93°F).

Water is an essential factor in the formation of sporangia, the transportation of zoospores, and the subsequent infection of roots. The severity of attack increases with soil moisture up to a maximum of field capacity. The optimum in soilless compost seems to be 60% to 95% moisture (the normal range for irrigated plants in containers).

All other factors (e.g. pH, conductivity), have little or no significant effect upon the development of *Phytophthora cinnamomi* but there is some evidence that high levels of surfactant added to peat as a wetting agent act as a stimulant in zoospore release.

As a result of the work carried out at the Plant Pathology Unit, methods of disease spread in order of priority were identified for Northern Ireland conditions. The most common method of spread is due to exchange of infected plant material among nurseries. Symptomless plants were found to be the largest danger. Methods of packing plants involving close contact in warm moist conditions enable rapid spread during transit. Contaminated containers and packing materials also assist in infection. All plants introduced onto a nursery must therefore be quarantined. This is best achieved by container growing on an isolated gravel bed for at least 12 months whilst their health is established by laboratory tests.

The unwitting spread of contaminated soil both on the nursery and between holdings is a major factor. If contaminated soil is left exposed then even a tractor wheel or hose pipe becomes a

vehicle of spread. Contaminated ground must be isolated with polythene or a grass sward established.

The microclimate produced under a system of intermittent mist is ideal for the development of *Phytophthora cinnamomi*. It is essential therefore that propagating materials should be free from contamination. All stock plants should be grown in isolation and checked for freedom from the disease. When in doubt the prospective material should be treated with a fungicide 14 days before cutting collection. All material is best taken from the higher parts of the stock plant. It is vitally important both during propagation and subsequent potting to maintain hygiene at a very high level to avoid contamination of the young plant material.

Disease spread within the holding is almost always from infected to healthy plants during the production cycle. This usually occurs when zoospores are released and are able to move from one container to the next via a "water bridge" (a continuous film of water). (N.B. A "water bridge" exists between adjacent plants on a sand bed.) This can happen at any stage in the production of hardy nursery stock — viz, mist unit to standing ground.

It is essential to break this "water bridge" by allowing free drainage from the base of the containers at all times. Standing areas consisting of free draining gravel are recommended as they also prevent reinfection of subsequent crops due to the fact that *Phytophthora cinnamomi* cannot persist in this medium.

Direct infection by mycelium following root contact is possible only when plants are allowed to root through into the standing ground. Plunging containers into peat thus provides not only the vital "water bridge" but also the opportunity for root contact.

Irrigation water is an ideal vehicle for infection as water is the natural environment of the zoospore. Infection can be rapid and complete (up to 90% of susceptible plants after only one application of heavily infected irrigation water). Two factors must therefore be strictly controlled. Drainage water from production areas must always be allowed uninterrupted passage from the holding to avoid contamination of the source of irrigation water. All water used for irrigation purposes should be from a clean source (mains or deep bore water is preferred).

Water splash on the surface of soil or compost during heavy rain or irrigation has been suggested as a major cause of spread. In Northern Ireland very few zoospores were found within the top 10 mm of soil. The only exception seemed to be with capillary watering, when it is feasible, that spores are carried upwards by capillary action.

The present chemical agents at the disposal of nurserymen

are limited to those capable of inhibiting mycelial growth (fungistatic) — viz, etridiazole and aluminium tris (ethyl phosphonate). There are as yet no chemicals capable of destroying the resistant resting bodies. Consequently when chemical treatment begins to become less effective or where the fungicide has failed to reach the mycelium, zoospores are released and can infect other plants and maintain spread without symptoms being seen Once treatment has been discontinued the fungus will resume attack and symptoms will soon become apparent.

When all the available information is considered it is apparent that prevention is the only practical approach. If the disease is to be controlled then it is necessary to destroy all known sources — total eradication. This in turn depends upon quick accurate diagnosis of the disease in the absence of symptoms. A method adopted by the Plant Pathology Unit for this purpose was to "bait" for the zoospore from a soil/root leachate by the use of pine needles (*Pinus radiata*) and plate this out on selected media. *Phytophthora cinnamomi* produces an easily recognizable botryose mycelium. From early May until late September results can be available in 5 days with 95% success of detection

In 1976 the Department of Agriculture for Northern Ireland offered a service to all hardy nursery stock producers for the eradication of *Phytophthora cinnamomi.* The specialist advisor when visiting a growers holding, either by direct request or during normal advisory visits, would collect samples of suspect plants. These were isolated in a sealed polythene bag for diagnosis by the Plant Pathology Unit. When *Phytophthora cinnamomi* was confirmed a representative number of soil/root samples were taken from all stock on that particular holding. Each batch of plants found to be infected was destroyed.

Subsequent advice was formulated for the particular holding and was based upon four main factors:

1. *Phytophthora cinnamomi* is seldom found more than 300 mm above soil level Cuttings taken above that height are seldom infected. No plant known to be infected should be used for propagation purposes.
2. Hygiene is of the utmost importance. Soil, sand, and containers that come into contact with the organism almost always cause a fresh outbreak.
3. Drainage water from an infected plant is the main cause of spread within the nursery. Drainage water should therefore be allowed an uninterrupted passage from the base of the pot through a gravel bed into a drainage system which discharges away from the nursery.
4 Most spread between holdings is by infected plants. All

plants should therefore be quarantined in a separate area until their health has been established. This is particularly important where plants have been treated with fungistatic chemicals. Liners bought in should be grown on a separate bed and not between batches produced on the nursery.

By the end of 1977 most of the major nurseries had been surveyed and had carried out our recommendations even when *Phytophthora cinnamomi* had not been isolated. No outbreak of the disease was recorded in the second half of 1977.

Five cases were recorded in 1978 where the infection occurred in Northern Ireland. All of these could be attributed to a disregard of our recommendations. Three outbreaks occurred on sandbeds and two as a result of a breach of quarantine precautions.

The health of our nursery stock has become well known and nurseries are increasing sales because of the very low incidence of this disease. Several nurseries requested regular surveys even though there was no suspicion of any infection as they have realized the potential value of clean stock.

It is now felt that our recommendations are sufficient to prevent any major spread within a holding and that most nurseries in Northern Ireland are reasonably free from this organism. If growers remain vigilant when importing new plant material onto their holdings the good health status of the nurseries should be maintained.

I would like to acknowledge the help given by Mr John Flack (formerly of the Department of Agriculture, Plant Pathology Division, Newforge Lane, Belfast), in the preparation of this paper

J. WARD: In reply to a question by A. Carter, we tested a 5% representative sample from the whole nursery.

A. CARTER: Five percent, and that was enough to free the nursery. This is what puzzles me. How do you manage to get rid of it?

J. WARD: When we found *Phytophthora cinnamomi* in any batch of plants we recommended that that batch of plants should be destroyed.

A. CARTER: The whole batch?

J. WARD: The whole batch. This accounted for something like 20% of the production on some nurseries.

P. WATSON: Doesn't it nevertheless mean that although you

have got the gravel bed laid down that there is no reason why the zoospores in their resting stage shouldn't still be in the sand or soil beneath the gravel?

J. WARD: We hose down the gravel bed and, provided the contaminated material is buried below the surface, the zoospores cannot come back up against the drainage water. If there is too much material we would have to renew the bed, but that has not happened yet. We recommend that where there has been contamination the most resistant species are placed on that bed the following year.

B. MORGAN. Would you say how you managed to adjust the compost that you used for this gravel base system, also the quantities of irrigation water that are required for container plants grown this way.

J WARD: Two very important points: If you are growing on gravel then you cannot use the same compost as you would use if you were on sand beds. It has to be a more open free-draining compost because during the winter months, in particular, in the lower levels of these pots the water just doesn't get away, and you get build-up. We use a 3 to 1 peat-sand mix but we use a gravel, not sand, and the gravel goes up to ¼". The easy test is that you should have more volume after you mix the sand into the peat than with peat alone. If you put sand into it you will find the volume actually decreases because it takes up the pore space and you get less volume. The granules of sand that we are putting in must be larger than pore space as we cannot block that pore space with the sand; if we do we are in trouble, so we use a sand that is a grit rather than a sand. We have a fair amount of water in Northern Ireland, not quite as much as coming down from the heavens as you may think, but we do have a fair amount of irrigation water. We haven't calculated too closely the exact requirements but the system does demand a very heavy amount of water.

D. HUSBAND: I'm interested in this swimming of spores against drainage water. To my mind if there is no precipitation then there is more danger in storage water.

J. WARD: If we do not have the precipitation then the water film will be broken as it drains away and there will be small dry areas that make it very difficult for the zoospores to actually swim up through the gravel. They don't get on very well in gravel because they have a long way to swim round each particle. It is even more difficult for it to get through broken stone and this is really why we recommend broken stone. We would like experiments to confirm this but we haven't managed to get the zoospores to come back up through the gravel.

K. LAWRENCE: How did you recommend these particular growers get rid of their infected stock?

J. WARD: The plants had to be destroyed, best by fire, destroying the container and composts as well. We covered them with a polythene tent open at the end. It takes about three weeks to dry the material to a degree where we can then burn it. We put it into very large polythene bags that are to be taken away and burnt.

DISCUSSION GROUP REPORTS
GROUP A
SUNFRAMES AND
LOW POLYTHENE TUNNEL PROPAGATION

CHAIRMAN — S.J. HAINES

The numbers attending this session showed the great interest there is in low cost propagation techniques.

On our pre-conference visit to Boulton Brothers nursery we had seen a full frame of conifers rooted under double glass, cuttings inserted in August and now ready for moving on.

Several members were using combinations of polythene and lights on their frames. Lila Dick favoured the polythene over the frame lights, thus sealing the lights and preventing pools of water accumulating on the polythene laid over the cuttings.

It was agreed that cuttings were often inserted at too great a density, and that better results were achieved by giving more space, this being particularly true of larger-leaved species such as hydrangeas. Roger Platts favoured potting these on in later summer, but care must be taken with many subjects due to overwintering problems.

John Ward and others spoke of their experience in using large tunnels as cover protection over low "inner" tunnels in which were rooted a wide range of summer cuttings.

David Whalley of the Glasshouse Crops Research Institute quoted the work of Keith Loach on light intensity and relative humidity under polythene. In lay terms their work boils down to finding the balance between the light required for photosynthesis and the need to conserve water in the cutting during the period prior to root formation. Light levels are measured in the currently favoured international units known as megajoules per square metre. In July radiation can measure up to 20 megajoules per square metre (20 MJ/m^2), but the desirable light level for rooting is between 1.5 MJ/m^2 and 3.0 MJ/m^2, so 78% shading would be