Ultrastructure of Calcareous Dinophytes (Thoracosphaeraceae, Peridiniales) with a Focus on Vacular Crystal-Like Particles

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Abstract

Biomineralization in calcareous dinophytes (Thoracosphaeraceae, Peridiniales) takes place in coccoid cells and is presently poorly understood. Vacular crystal-like particles as well as collection sites within the prospective calcareous shell may play a crucial role during this process at the ultrastructural level. Using transmission electron microscopy, we investigated the ultrastructure of coccoid cells at an early developmental stage in fourteen calcareous dinophyte strains (corresponding to at least ten species of Calciodinium, Calcigenellum, Leonella, Pernambugia, Scrippsiella, and Thoracosphaera). The shell of the coccoid cells consisted either of one (Leonella, Thoracosphaera) or two matrices (Scrippsiella and its relatives) of unknown mineral composition, whereas calcite is deposited in the only or the outer layer, respectively. We observed crystal-like particles in cytoplasmic vacuoles in cells of nine of the strains investigated and assume that they are widespread among calcarious dinophytes. However, similar structures are also found outside the Thoracosphaeraceae, and we postulate an evolutionarily old physiological pathway (possibly involved in detoxification) that later was specialized for calcification. We aim to contribute to a deeper knowledge of the biomineralization process in calcarious dinophytes.

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Introduction

Biomineralization is defined as the fundamental biological process by which living organisms produce minerals, often to harden or stiffen existing tissues or subcellular organic matrices. Mineralized structures have evolved multiple times independently and are taxonomically widely distributed over the tree of life. Subsequently, many similar cellular steps take place in distantly related lineages [1–4], and the resulting, occasionally complex crystal architectures may have multifunctional properties. The biomineralization process and its structural basis are well understood in metazoans including mollusks, corals, and vertebrates [5–7], but also in such protists as foraminifers and coccolithophores [8–10]. Mineralized structures are likewise found in many taxa of the unicellular Dinophyceae (Alveolata), where the potential to form calcareous structures is unique within the entire alveolates, it has been considered an apomorphic character trait supporting the monophyly of the Thoracosphaeraceae [17]. This assumption has gained some corroboration from molecular sequence data, although a number of (presumably secondarily) non-calcareous taxa might be also included in this group [18–20]. Molecular phylogenies segregate calcarious dinophytes into three main lineages, namely the E/Pe-clade (for Ensiculifera Balech, 1967 and Pentapharsodinium Indel. & A.R.Loebl.), the T/Pf-clade (for Thoracosphaera Kampter & Pfister Steid. & J.M.Burkh.), and Scrippsiella Balech ex A.R.Loebl. sensu lato (s.l.).

Since the early studies of dinophyte anatomy using transmission electron microscopy (TEM) [21–23], much progress has been made in understanding their complex and diverse organizations at the subcellular level. Much attention has been given to the ‘apical furrow’ system [24–25] and the flagellar apparatus [26–29], but the biomineralization process in calcarious dinophytes has not been the focus of such studies. Ultrastructure investigations into the subcellular components involved in biological processes such as the encystment of cells expand the basic data necessary for robust phylogenetic reconstructions.

Immature coccoid cells of Scrippsiella minima X.Gao & J.D.Dodge, in which the initial phase of mineralization takes place, are surrounded by two continuous matrices of unknown compounds that are delineated by an outer, middle, and inner unit...
crystal-like particles comparable to the shell [30,38]. Nevertheless, we have found vacuoles containing immature coccoid cells because they are the likely stages, in which knowledge in their Agenda Calcareous Dinophytes. We focus on [37] have identified this field as one of the most serious gaps in Th. heimii inouye and pienaar [31] have studied the crystallization process in small, cylindrical seed crystals are found in immature coccoid cells. The matrix is filled with numerous cytoplasmic vesicles (or vacuoles) likewise containing cylindrical fibrous material. In coccoid cells of Th. heimii, membrane [30]. It has been assumed that calcification proceeds within the outer matrix, starting at protrusions composed of fibrous material. In coccolid cells of Thoracosphaera heimii (Lohmann) Kampptner, a single matrix develops, delineated by an outer and inner unit membrane [31–32]. The matrix is filled with numerous large, regularly arranged crystals in mature coccolid cells, while small, cylindrical seed crystals are found in immature coccolid cells. Inouye and pienaar [31] have studied the crystalization process in Th. heimii in more detail and have discovered small and large cytoplasmic vesicles (or vacuoles) likewise containing cylindrical crystals. Such vacuoles may derive from the Golgi apparatus and actively transport seed crystals from the center towards the periphery of the cell. ‘Crystal-like bodies’ similar in appearance have been reported outside the calcareous dinoflagellates, including other Peridiniales [33–35] and Suessiales [24,36].

In this study, we investigate the ultrastructure of several calcareous dinoflagellates to document the subcellular structures that may play a role in biomineralization. Elbracht and colleagues [37] have identified this field as one of the most serious gaps in knowledge in their Agenda Calcareous Dinophytes. We focus on immature coccoid cells because they are the likely stages, in which such structures can be observed. The study of (even immature) coccolid cells is challenging, as fixatives frequently do not penetrate the shell [30,38]. Nevertheless, we have found vacuoles containing crystal-like particles comparable to Thoracosphaera [31] also in other species. We aim to contribute to a more complete understanding of the biomineralization process in calcareous dinoflagellates.

### Materials and Methods

#### Morphology

Fourteen calcareous dinoflagellate strains were collected and isolated from environmental samples (Table 1). They were cultivated in a climate chamber Percival I-36VL (CLF PlantClimatics; Emersacker, Germany) at 18°C or 23°C, 80 µmol photons m⁻² s⁻¹, and a 12:12 h light: dark photoperiod by using K-Medium without silicate [39] and 35 or 30 psu artificial seawater (hw marinemix professional: Wiegandt; Krefeld, Germany) at pH 8.2. Strains are currently held in the culture collections at the Institute of Historical Geology/Palaeontology (University of Bremen, Germany) and the Institute of Systematic Botany and Mycology (University of Munich) and are available upon request.

Cultivated living cells were observed using an Olympus CX41 inverse microscope, equipped with a Kappa camera DX 20H-FW (supplied with Calypso software). For scanning electron microscopy (SEM) preparation, coccolid cells were desalinated in bidistillate water and air-dried on a glass slide that was fixed on a SEM stub (details are given in [40]). Samples were sputter-coated with platinum and documented with a LEO 438 VP (Zeiss) SEM.

#### Materials and Methods

### Table 1. Species list of TEM investigations (abbreviations: n.i., not indicated).

| Strain No. | Taxonomy | Species name with author | Locality | Lat. | Long. | Collector |
|------------|----------|--------------------------|----------|------|-------|-----------|
| GeoB 110  | Scrippsiella Calcispherion infusa Deflandre, 1949 | Mediterranean Sea (Spain) | 41°21'N | 3°0'1"E | n.i. |
| SZN 74    | Scrippsiella Calcispherion operatum Deflandre, 1947 | Mediterranean Sea (Italy) | 40°43'N | 14°10'E | Montresor |
| GeoB 34   | Scrippsiella Calcispherion aff. Operatum Deflandre, 1947 | Middle Atlantic | 08°30'N | 32°27'W | n.i. |
| tub²2     | Scrippsiella “Calcispherion” spec. | Eastern South Pacific (Chile) | 28°15'S | 78°00'W | n.i. |
| GeoB 38   | T/Pf Leonella granifera (D.Fütterer) Janofske & Karwath | Western Atlantic (Brazil) | 06°57'N | 47°54'W | n.i. |
| GeoB*61   | Scrippsiella Pennambulia tuberosa (Kamptner) Janofske & Karwath | South Atlantic (Brazil) | 11°32'S | 28°35'W | n.i. |
| GeoB 411  | Scrippsiella Scrippsiella trochoidea E Meier | Mediterranean Sea (Italy) | 41°15'N | 13°36'E | Gotschling, Zinßmeister & Söhner |
| GeoB*185  | Scrippsiella Scrippsiella trochoidea (F.Stein) A.R.Loebl. | Baltic Sea (Germany) | 54°22'N | 10°09'E | Meier |
| GeoB 188  | Scrippsiella Scrippsiella aff. Trochoidea (F.Stein) A.R.Loebl. | Mediterranean Sea (France) | 42°38'53"N | 3°08'E | Gotschling |
| GeoB 283  | Scrippsiella Scrippsiella aff. trochoidea (F.Stein) A.R.Loebl. | North Sea (Norway) | 63°28'N | 9°25'E | Gotschling & Petersen |
| GeoB 404  | Scrippsiella Scrippsiella aff. trochoidea (F.Stein) A.R.Loebl. | Mediterranean Sea (Italy) | 40°40'N | 14°46'E | Gotschling, Zinßmeister & Söhner |
| M34*25/5  | Scrippsiella Scrippsiella trochoidea (F.Stein) A.R.Loebl. | South Atlantic (Guyana) | 11°54'N | 57°48'90"W | n.i. |
| GeoB 228  | Scrippsiella Scrippsiella trochoidea var. aciculifera Montresor | Mediterranean Sea (Italy) | 40°07'N | 17°19'E | n.i. |
| GeoB 211  | T/Pf Thoracosphaera heimii (Lohmann) Kampptner | Eastern Mediterranean Sea | – | – | n.i. |

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buffer at pH 0 in a graded saccharose series (0.125 M, 0.05 M, 0.025 M, 0.01 M, without) each for 15 min and post-fixed with 1% osmiumtetroxide (Science Services; Munich, Germany) in 0.2 M cacodylate buffer. Following the instructions of the Embedding Medi Kit (Science Services), samples were dehydrated in a graded ethanol (Roth) or acetone (Roth) series (30%, 50%, 70%, 90%, 100%, 100%, 100%), and gradually infiltrated and embedded in Spurr’s resin [41].

The largest thecae and cells with a roundish form even as large as coccoid stages were selected for sectioning. Ultrathin sections were prepared with an ultra microtome (Leica EM UC6 Ultramikrotom). Sections were spread with 99% chloroform (Roth) and collected on copper 200 square mesh and 200 single bar grids (Plano) covered with collodium. Grids were stained with 1% aqueous uranylacetate (Plano) for 2 min and lead citrate (Plano) for 4 min [42]. TEM observations were done using a FEI Morgagni or a Zeiss EM 912.

Results

Cell morphology and life history

Leonella granifera (Fütterer) Janofske & Karwath and Th. heimii (both members of the T/Pf clade) produced mainly coccoid cells dividing vegetatively (thecate cells were rarely found under cultivation conditions). All investigated species of Scrippsiella s.l. (i.e., including also those of Calciodinellum Deflandre, 1949, Calciodinellum Deflandre, 1947, and Pernambugia Janofske & Karwath) developed golden brown, photosynthetically active thecate cells, which were always abundant under cultivation conditions because of vegetative division. The shape of the thecate cells was spherical to ovoid, with a rounded to conical apex. Early coccoid cells developed after shed of the theca by ecdysis and were darker than the thecate cells from the beginning. A red accumulation body was often already visible at this stage. Coccoid cells appeared first hyaline and darkened towards the brownish-opaque color at maturity after a few minutes.

Figure 1 shows a SEM image selection of the calcareous coccoid cell diversity investigated here with respect to their ultrastructure. Mature coccoid cells varied in size across species and were spherical to ovoid. The shell surface likewise differed between taxa, ranging from smooth without ornamental structures (Th. heimii: Fig. 1B) to reticulate (Calciodinellum aff. operosum Deflandre, 1947: Fig. 1F), spiny [Scrippsiella trochoidea (F.Stein) A.R.Loebl.: Fig. 1C,E], imperfectly intratabulate (Scrippsiella bicornata Zinssmeister, S.Soehner, S.Meier & Gottschling: Fig. 1A) to holotabulate (C. operosum: Fig. 1D). In some cases, remnants of an outer membrane covering the coccoid cell were present, and this membrane was always entirely intact in Th. heimii (Fig. 1B) and L. granifera.

Comparative ultrastructure

Cell ultrastructure was largely similar in organization among different calcareous dinophyte species (Figs. 2–4). Thecate cells were always smaller than coccoid cells within particular strains. Thecal plates were surrounded by a unit membrane (Figs. 2A, 4A,E), which was visible particularly at their boundaries (Figs. 2C, 4E). Chloroplasts, mitochondria, and other compartments such as trichocysts were likewise present in all of the cells examined.
In all of the thecate cells investigated (Fig. 2), many large chloroplasts were present in peripheral positions. Moreover, different types of pyrenoids were found within those cells that showed structural associations with the chloroplasts. Some large chloroplasts constituted a network, as they were connected by multiply-stalked pyrenoids (Figs. 2A,C–D, 3A–B). Additionally, starch grains adjacent to pyrenoids were present (Fig. 3A). Smaller chloroplasts showed internally fusiform, interlamellar pyrenoids (Figs. 2A,C, 3B). Some chloroplasts were neither attached to pyrenoids nor to starch grains and were particularly small in size. (Figs. 3A, 5E). The thylakoid lamellae were more or less parallel to each other, an arrangement that was occasionally perturbed by the presence of pyrenoids (in which case the lamella fibers led into the pyrenoids). The thylakoids consisted of two to four lamellae (Figs. 2A, 3C, 4G).

Oval to elongated mitochondria with tubular cristae and surrounded by two unit membranes were visible (Fig. 3D). They were numerous and distributed all over the cytoplasm of thecate
cells (Fig. 2A,C). The globose dinokaryon surrounded by two unit membranes was located close to the center of the cell and always showed condensed, rod-shaped chromosomes (Figs. 2A–B,D,E). The Golgi apparatus was located in the center of the cell close to the dinokaryon and consisted of a stack of few, flattened cisterns of the dictyosome. Golgi-derived vesicles were likewise visible near the dictyosome (Fig. 4G). A roundish accumulation body (Fig. 2B) was often present in large thecate cells and was always developed in coccoid cells.

In *S. aff. trochoidea*, a remarkably high number of lysozymes (Fig. 6C,D) were scattered throughout the cytoplasm and were even found within vesicles. The pusular system was formed by tubules of about 50 to 100 nm (Figs. 4A,H), and vesicles leading close to the flagellar base. Under the plasmalemma, cellulose plates were present that overlapped at their boundaries (Figs. 4A–E). The apical furrow system consisted of the apical pore plates, and the pore itself was covered by one such plate (Fig. 4A). The sulcal region consisted of few overlapping sulcal plates (Fig. 4C). A roundish accumulation body (Fig. 2B) was often present in large thecate cells and was always developed in coccoid cells.

Before encystment and particularly in cells of the coccoid stage, size and number of chloroplasts decreased. Moreover, the number of starch grains and lipid droplets (both with a storage function) increased during encystment (Figs 2B,D) and in coccoid cells (Fig. 2E). The calcitic crystals deposited in the single matrix surrounding the cell were not preserved after the treatment with uranylacetate and lead citrate and were therefore recognized as empty space in the thin sections.

Crystal-like particles and biomineralization

In nine of fourteen strains (corresponding to at least eight species), crystal-like particles were detected in encysting cells. They were found within cytoplasmic vacuoles, which were peripherally located in the cell body. Such vacuoles were variable in size and were particularly large in *Calciodinellum aff. operosum* (Fig. 5C), *C. aff. operosum* (Fig. 5F), and *S. aff. trochoidea* (Fig. 6C–D). The crystal-like particles in the vacuoles were between 50 to 380 nm in length and 24 to 86 nm in width. They had an elongated and cylindrical shape and were irregularly scattered with sometimes large gaps between each other. Only *C. operosum* (Fig. 5G) showed large, densely arranged crystal-like particles of varying size (330 to 1200 nm long and 70 to 470 nm wide) and shape, ranging from rod-like to cylindrical to square-cut. Crystal-like particles of *Pernambugia tuberosa* (Kampffner Janoske & Karwath (Fig. 5D), *S. bicarinata* (Fig. 5E), *S. trochoidea* (Fig. 6A–B), *S. trochoidea var. aciculifera* Montresor (Fig. 5A), and *Th. heimii* (Fig. 5B) had an ovoid to barrel-like shape with blunt edges. In *C.
Figure 4. Ultrastructural traits in detail. (TEM). A. ‘Apical furrow’ system of Scripsiella trochoidea (GeoB 377, note the numerous vesicles under the cell surface). B. Overlapping thecal plates of Scripsiella trochoidea (GeoB 188, note the outer protrusions of overlapping theca plates). C. Overlapping theca plates in the sulcal region of “Calciodinellum” spec. (tub*2). D. Strand of peripheral microtubules in Scrippsiella bicarinata (GeoB 411, note the multiple membranes under the cell surface). E. Thecal plate boundary of Scripsiella trochoidea (GeoB 377, note the detached outer unit membrane). F. Trichocysts of Scripsiella trochoidea var. aciculifera (GeoB 228). G. Subcellular organization of Scrippsiella bicarinata (GeoB 411, note the longissession of a trichocyst and the rough endoplasmatic reticulum indicated by an arrow). H. Golgi apparatus of Scrippsiella trochoidea (M34*25/5). J. Pusule of Scrippsiella bicarinata (GeoB 411, note the multiple membranes under the cell surface). Abbreviations: cb, crystal-like particle; ch, chloroplast; cp, cover plate; d, dictyosome, fb, microtubular fiber; gv, Golgi-derived vesicle; me, unit membrane; mt, mitochondrion; po, pore plate; pv, pusular vesicle; py, pyrenoid; re, rough endoplasmatic reticulum; st, starch grain; tr, trichocyst; th, thecal plate. Scale bars: A to C and E to G 1 μm, D, H, J 0.1 μm.

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infula (Fig. 5C), C. aff. operosum (Fig. 5F), and S. aff. trochoidea (Fig. 6C–D), the shape of the crystal-like particles was square-cut.

The crystal-like particles showed some structural association to the vacuolar membrane (in, e.g., P. tuberosa: Fig. 5D and S. trochoidea: Fig. 6A–B) and/or internal membranes of the vacuoles (in, e.g., S. bicarinata: Fig. 5E and Th. heimii: Fig. 5B). In S. aff. trochoidea (Fig. 6C–D) and C. operosum (Fig. 5G), the crystals seemed to be present in higher number and surrounded by dense material of unknown origin, possibly unit membranes. A connection between membranes and crystal-like particles was not detected in C. infula (Fig. 5C) and C. aff. operosum (Fig. 5F), but it was not clear whether this was a distinctive character.

Discussion

Ultrastructure

Ultrastructure studies of coccoid dinoflagellate cells are still rare because of many methodological problems [30,39]. Nevertheless, they have great importance in better understanding the precise biological function of this specific developmental stage and gaining more basic data for phylogenetic reconstructions. Ecological stress such as temperature and light [43] and reduced availability of iron or nutrients may interfere with the interpretation of cellular ultrastructure [44–45]. We have aimed to avoid such bias by investigating cells held under constant culture conditions. Most of the cells studied here have shown subcellular details such as a reduced number of chloroplasts and an increased number of starch grains and lipid drops. These features have been interpreted
as typical indicators for encysting cells or (early) coccoid cells, respectively [27,30,38,46].

The number of layers constituting the shell of coccoid cells might have some phylogenetic significance [47]. *Thoracosphaera* [31–32] and *Leonella* Janofske & Karwath have a single matrix surrounding the cell (delineated by two unit membranes), while all extant species of *Scrippsiella* (including *S. minima* [30]) and its putative relatives such as *Calciodinellum* and *Pernambugia* share two layers (delineated by three unit membranes). These two types of shell architecture may correlate with molecular phylogenies, in which *Scrippsiella s.l.* and the T/Pf-clade represent two distinct lineages of the Thoracosphaeraceae [19–20]. As most peridinioid dinophytes exhibit the two-layer type [48–49], it is most plausible to assume that the one-layer type is apomorphic and today restricted to the T/Pf-clade. For future research, it is tempting to investigate the number of layers in coccoid cells of other members of the T/Pf-clade such as *Pfiesteria*.

For *S. minima*, Gao and co-workers [30] concentrated on mucilaginous material between the inner and the middle unit membranes surrounding the coccoid cell. This material is raised to form protrusions that are calcified in later stages of development. The presence of vacuoles including crystal-like particles has been previously reported from *Scrippsiella sweeneyae* Balech ex A.R.Loebl. [51] and *Th. heimii* [31], and the latter authors have assumed a crucial role of those structures during biomineralization. A striking observation of the present study is that vacuolar crystal-like particles are abundant among calcareous dinophytes (records for seven more species). They have also been documented in *Tyrannodinium edax* (A.J.Schill.) Calado (= *Peridinium berolinense* Lemmerm. [35], a non-calcareous member of the T/Pf-clade [52]). However, vacuolar crystal-like particles have been sporadically reported (albeit under different names) from dinophytes (and also in thecate cells), but never have they been the focus of ultrastructural studies in a comparative approach. They have been found in the Gonyaulacales [53], Peridiniales [33–34], and

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**Figure 6. Vacuolar crystal-like particles.** (TEM). A: *Scrippsiella trochoidea* (GeoB*185*, early coccoid cell, the theca is still attached; note the intravacuolar vesicles). B. *Scrippsiella aff. trochoidea* (GeoB*185*). C. *Scrippsiella aff. trochoidea* (GeoB 283, note the lysozymes as intravacuolar membrane whors). D. *Scrippsiella aff. trochoidea* (GeoB 283, note the lysozymes as intravacuolar membrane whors). Abbreviations: cb, crystal-like particle; ch, chloroplast; ix, inner matrix; ly, lyosme; me, unit membrane; th, thecal plate. Scale bars: 1 μm.

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Suessiales [24,36,38,54], but their variation in size, shape, and subcellular distribution makes an overall homology unlikely.

**Chemistry and Function**

Analytical chemistry of the crystal-like particles might likewise be indicative of their independent evolutionary origin, although the precise molecular composition based on ultimate analyses is rarely investigated. In the Suessiales, the vacuolar crystals with a characteristic rectangular shape are composed of calcium oxalate [55], while the bi-rhombohedral particles found in the Gonyaulacales contain guanine and other as yet unidentified components [56]. Conversely, the mature shell of calcareous dinophytes is composed of calcite elements, as it has been determined for S. trocholaesa [12] and Th. heimi [57]. Inouye and Fernaæ [31] have shown that the vacuolar crystal-like particles are sensitive to acid, and there is no reason to assume that they are not calcitic. For future research, the precise molecular composition of such particles found outside the Thoracosphaeraceae (in, e.g., Gephyrocapsa oceanica [34] and Peridinopsis Lemmerm., [33]) is essential to reliably determine whether they are homologous across the Peridiniales.

Multiple functions of vacuolar crystal-like particles have been discussed. In the Suessiales, the mature elements are characteristically brick-like [24,36,58–59] and are associated with an eyespot in a regular arrangement of one to several rows [60–61]. Eyespots effectively absorb and reflect blue-green laser light [62] and are structurally connected to the flagellar apparatus. The support of these structures in locomotion has been therefore suggested [63]. More generally, the vacuoles containing crystal-like particles have been variously interpreted to be involved in the detoxification of the dinophyte cell [38,53,64]. Many calcifying organisms have access to corresponding physiological pathways to compose their aragonite and calcite structures [65–66]. Calcareous dinophytes have access to corresponding physiological pathways to detoxification of the dinophyte cell [38,53,64]. Many calcifying organisms have access to corresponding physiological pathways to compose their aragonite and calcite structures [65–66]. Calcareous dinophytes may have thus modified the potential for calcification by elevating their intracellular pH. Proc Natl Acad Sci USA 106: 15374–15378.

**Conclusion**

Compared to calcareous dinophytes, biomineralization in other unicellular organisms, such as the foraminifers and coccolithophores, has been more thoroughly investigated [9–10]. Foraminifers show principle differences in this process, as needle-like seed crystals are formed in vacuoles prior to the calcification of the shell (milolid species), or not (hyaline species) [8]. At the ultrastructure level, the milolid type somewhat resembles what is demonstrated for (calcareous) dinophytes in this and other studies. In coccolithophores, the crystallization process leading to the mature coccoliths takes place in Golgi-derived vesicles [10] moving from the cell center to the periphery.

The assumption that calcareous dinophytes have a similar calcification mechanism as coccolithophores has been postulated by Tangen and colleagues [32]. It is generally accepted that biominalization in calcareous dinophytes also takes place under strong control at the cellular level [13,70–71]. Tabulation patterns that are reflected in the shell of the cocoid cells in at least some members of the Thoracosphaeraceae indicate that biominalization is linked to amphiboles vesicles constituting the thecal plates. In cocoid cells at an early developmental stage, calcitic seed crystals are formed in vesicles that probably derive from the Golgi apparatus. Such vesicles are transported to the cell periphery, and the seed crystals are deposited in the outer (or only) matrix surrounding the cocoid cell. They may accumulate at collection sites, where they are visible as protrusions of mucilaginous material (functioning as ‘skeletons’: [30]). However, it remains unclear at present how the seed crystals pass through the inner matrix in Scyphophora and its relatives. More research on the life history, ultrastructure, and physiology is necessary, and high-spatial resolution analyses such as NanoSIMS, Raman spectroscopy, soft X-ray microscopy, and atomic force microscopy may be promising approaches in developing a comprehensive scenario for biominalization in calcareous dinophytes.

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**Author Contributions**

Conceived and designed the experiments: CZ MG. Performed the experiments: CZ FK. Analyzed the data: CZ HK GT. Contributed reagents/materials/analysis tools: CZ HK GT FK MG. Wrote the paper: CZ MG.

**References**

1. De Yoreo JJ, Vekilov PG (2003) Principles of crystal nucleation and growth. Rev Mineral Geochem 54: 57–94.
2. Knoll AH (2003) Biominalization and evolutionary history. In: DowE PM, De Yoreo JJ, Weiner S, editors. Biominalization. Washington, DC: Mineralogical Society of America. pp 329–336.
3. Murdock DJE, Donoghue PCJ (2011) Evolutionary origins of animal skeletal biominalization. Cells Tissues Organs 194: 98–102.
4. Raven JA, Giordano M (2009) Biomineralization by photosynthetic organisms: Evidence of coevolution of the organisms and their environment? Geobiology 7: 140–154.
5. Bauerlein E (2004) Biomineralization. Progress in biology, molecular biology and application. Weinheim: Wiley.
6. Mann S (2001) Biomineralization: Principles and concepts in bioinorganic materials chemistry. Oxford: Oxford University Press.
7. Simkis K (1989) Biomineralization. San Diego: Academic Press.
8. de Nooijer LJ, Toyofuku T, Kitazato H (2009) Foraminifera promote calcification by elevating their intracellular pH. Proc Natl Acad Sci USA 106: 15374–15378.
9. Young JR, Davis SA, Bown PR, Mann S (1999) Coccolith ultrastructure and biomineralisation. J Struct Biol 126: 195–215.
10. Young JR, Heinricken K (2003) Biominalization within vesicles: The calcite of coccoliths. In: DowE PM, De Yoreo JJ, Weiner S, editors. Biominalization. Washington, DC: Mineralogical Society of America. pp 189–213.
11. Janoskie D (1996) Ultrastructure types in recent “calciophores”. Bull Inst Oceanogr Monaco Né: spéciale 14: 295–303.
12. Wall D, Guillaud RRL, Dale B, Swift E, Watebe N (1970) Calcitic resting cysts in Proxillina trochoidea (Stein) Lemmermann, an autotrophic marine dinoflagellate. Phycologia 9: 151–156.
13. Meier S, Engemann N, Gotschlich M, Kohring R (2009) Die Bedeutung der Struktur der Zystenwand Kalkiger Dinoflagellaten (Thoracosphaeraceae, Dinophyceae). Ber paläoabil Abh 10: 245–256.
14. Bolli HM (1974) 39. Jurassic and Cretaceous Calcisphaerulidae from DSDP Leg 27, eastern Indian Ocean. Init Rep Deep Sea 27: 843–907.
15. Deflandre G (1946) Les Calcisphacées. Dinoflagellés à thèque calcaire. Le Botaniste 34: 191–219.
16. Keupp H (1981) Die kalkigen Dinoflagellaten-Zysten der borealen Unter-Kreide (Unter-Hauterivium bis Unter-Albium). Facies 6: 1–190.
35. Wedemayer GJ, Wilcox LW (1984) The ultrastructure of the freshwater, 
33. Calado AJ, Moestrup Ø (2002) Ultrastructural study of the type species of 
29. Roberts KR (1989) Comparative analyses of the dinoflagellate flagellar 
28. Hansen G, Moestrup Ø (2005) Flagellar apparatus and nuclear chambers of the 
40. Zinssmeister C, Soehner S, Facher E, Kirsch M, Meier KJS, et al. (2011) Catch 
39. Keller MD, Selvin RC, Claus W, Guillard RRL (1987) Media for the culture of 
37. Elbra¨chter M, Gottschling M, Hildebrand-Habel T, Keupp H, Kohring R, et al. 
19. Gottschling M, Keupp H, Plotner J, Knop R, Willems H, et al. (2005) Phylogeny 
18. Craveiro SC, Calado AJ, Daugbjerg N, Hansen G, Moestrup Ø (2011) 
24. Craveiro SC, Moestrup Ø, Daugbjerg N, Calado AJ (2010) Ultrastructure and 
21. Dodge JD (1971) Fine structure of the Pyrrophyta. Bot Rev 37: 481–508. 
46. Chapman DV, Dodge JD, Heaney SI (1982) Cyst formation in the freshwater 
69. Nehring S (1994) 
68. Hesse K-J, Tillmann U, Nehring S, Brockmann U (1996) Specific factors 
64. Pokorny KS, Gold K (1973) Two morphological types of particulate inclusions 
61. Moestrup Ø, Daugbjerg N (2007) On dinoflagellate phylogeny and classification. 
56. DeSa R, Hastings JW (1968) The characterization of scintillons: Bioluminescent 
52. Calado AJ (2011) On the identity of the freshwater dinoflagellate 
58. Horiguchi T, Pienaar RN (1994) 
57. Gussone N, Zonneveld K, Kuhnert H (2010) Minor element and Ca isotope 
56. DeSa R, Hastings JW (1968) The characterization of scintillons: Bioluminescent 
50. Monnet B (1993) Wechselseitige Beziehungen organischer und kalzitischer 
43. Sgrosso S, Esposito F, Montresor M (2001) Temperature and daylength regulate 
45. Lewis J (1988) Cysts and sediments: Gonyaulax polyedra (Dinophyceae): Effects of iron stress. J Phycol 5: 721–731. 
37. Gussone N, Zonneveld K, Kuhnert H (2010) Minor element and Ca isotope 
568–585. 
49. Pitt M, Hohnadel J (1971) Ultrastructure of Calcareous Dinophytes. 
19. Gottschling M, Keupp H, Plotner J, Knop R, Willems H, et al. (2005) Phylogeny 
21. Dodge JD (1971) Fine structure of the Pyrrophyta. Bot Rev 37: 481–508. 
43. Sgrosso S, Esposito F, Montresor M (2001) Temperature and daylength regulate 
17. Wall D, Dale B (1968) Modern dinoflagellate cysts and evolution of the 
18. Craveiro SC, Calado AJ, Daugbjerg N, Hansen G, Moestrup O (2011) 
19. Gottschling M, Keupp H, Plotner J, Knop R, Willems H, et al. (2005) Phylogeny of 
20. Bibby BT, Dodge JD (1974) Fine-Structure of chloroplast nucleoid in 
59. Moeller KJ, Lehnert EJ, Tillmann U, Nehring S (1998) Specific factors 
29. Roberts KR (1989) Comparative analyses of the dinoflagellate flagellar 
20. Bibby BT, Dodge JD (1974) Fine-Structure of chloroplast nucleoid in 
40. Zinssmeister C, Soehner S, Facher E, Kirsch M, Meier KJS, et al. (2011) Catch 
50. Monnet B (1993) Wechselseitige Beziehungen organischer und kalzitischer 
21. Dodge JD (1971) Fine structure of the Pyrrophyta. Bot Rev 37: 481–508. 
19. Gottschling M, Keupp H, Plotner J, Knop R, Willems H, et al. (2005) Phylogeny 
21. Dodge JD (1971) Fine structure of the Pyrrophyta. Bot Rev 37: 481–508. 
19. Gottschling M, Keupp H, Plotner J, Knop R, Willems H, et al. (2005) Phylogeny 
20. Bibby BT, Dodge JD (1974) Fine-Structure of chloroplast nucleoid in 
40. Zinssmeister C, Soehner S, Facher E, Kirsch M, Meier KJS, et al. (2011) Catch 
50. Monnet B (1993) Wechselseitige Beziehungen organischer und kalzitischer 
21. Dodge JD (1971) Fine structure of the Pyrrophyta. Bot Rev 37: 481–508. 
41. Schrader K, Beblo G, Kubiszewski J, Tyczynski S, Wolinska M (1987) Cyst formation in the marine 
59. Moeller KJ, Lehnert EJ, Tillmann U, Nehring S (1998) Specific factors 
19. Gottschling M, Keupp H, Plotner J, Knop R, Willems H, et al. (2005) Phylogeny 
50. Monnet B (1993) Wechselseitige Beziehungen organischer und kalzitischer 
21. Dodge JD (1971) Fine structure of the Pyrrophyta. Bot Rev 37: 481–508. 
41. Schrader K, Beblo G, Kubiszewski J, Tyczynski S, Wolinska M (1987) Cyst formation in the marine 
59. Moeller KJ, Lehnert EJ, Tillmann U, Nehring S (1998) Specific factors 
19. Gottschling M, Keupp H, Plotner J, Knop R, Willems H, et al. (2005) Phylogeny 
21. Dodge JD (1971) Fine structure of the Pyrrophyta. Bot Rev 37: 481–508. 
19. Gottschling M, Keupp H, Plotner J, Knop R, Willems H, et al. (2005) Phylogeny 
21. Dodge JD (1971) Fine structure of the Pyrrophyta. Bot Rev 37: 481–508. 
