with greater litter accumulation. Lower values detected in the disturbed sites may be due to high mortality and rapid turnover of fine roots[9].

The canopy openings alter edaphic environment such as soil moisture, soil temperature, light availability and nutrient cycling, which are ultimately reflected in fine root dynamics (Table 4)[3-5,9,24]. Ewel et al.[25] found no significant differences in fine root biomass 11 weeks following gap creation. A similar trend was reported in a low-land wet tropical forest of Costa Rica by Raich[2]. On the contrary, it has also been reported that fine root biomass and NPP in top 30 cm of the soil were much lower in openings in all sizes than under plants and declined steeply as opening size increased[3-5,24]. Lower levels of very fine and fine root biomass and NPP under canopy openings confirm and support the theory proposed by Wilczynski and Pickett[10] that the above-ground gap creates below-ground gap. This could be attributed to faster turnover rates[9]. Greater very fine and fine roots biomass and NPP under intact forests (closed canopies) could be attributed to greater litter accumulation along with higher tree density and basal area as suggested by Visalakshi[26].

Pattern of seasonal variations in root biomass and NPP (maximum during first rainy season and minimum in the dry season) observed in the present study is also in confirmation with others[26,27], and could be attributed to changes in edaphic environment such as soil moisture, soil temperature, litter accumulation and root turnover[5,9].

The present study thus reveals that disturbances such as annual wild fire and cattle grazing enhance very fine root biomass and NPP and also confirms the theory that the above-ground gap creates below-ground gap in deciduous forests.

1. Aber, J. D., Melillo, J. M., Nadelhoffer, K. J., McClaugherty, C. A. and Pastor, J., Oecologia, 1985, 66, 317–321.
2. Raich, J., Biotropica, 1980, 12, 231–232.
3. Sanford, R. L., J. Trop. Ecol., 1989, 5, 153–159.
4. Sanford, R. L., Oecologia, 1990, 83, 541–545.
5. Hook, P. B., Lauenroth, W. K. and Burke, I. C., J. Ecol., 1994, 82, 485–494.
6. Yin, X., Perry, J. A. and Dixon, R. K., For. Ecol. Manage., 1989, 27, 159–177.
7. Chandrashekara, U. M., Ph D Thesis, Jawaharlal Nehru University, New Delhi, 1991, p. 148.
8. Safford, L. O., Plant Soil, 1974, 40, 349–363.
9. Nadelhoffer, K. J., Aber, J. D. and Melillo, J. M., Ecology, 1985, 66, 1377–1390.
10. Wilczynski, C. J. and Pickett, S. T. A., J. Veg. Sci., 1993, 4, 571–574.
11. Jordan, C. F. and Escalante, G., Ecology, 1980, 61, 14–18.
12. Budowski, G., Turrialba, 1965, 15, 40–42.
13. Richards, P. W., The Tropical Rain Forests: An Ecological Study, Cambridge Univ Press, 1952, 4th edn, p. 450.
14. Staterson, K. A. and Vitousek, P. M., Can. J. Bot., 1984, 62, 823–829.
15. Smith, W. H., Bormann, F. H. and Liken, G. E., Soil Sci., 1968, 106, 471–473.
16. Ramakrishnan, P. S. and Ram, S. C., Vegetatio, 1988, 74, 47–53.
17. Hadley, E. B. and Kieckhefer, B. J., Ecology, 1963, 44, 389–395.
18. Pandey, A. N., Trop. Ecol., 1974, 15, 152–153.
19. Smith, D. H. and Schmutz, E. M., J. Range Manage., 1975, 28, 453–458.
20. Aerts, R., Bakker, C. and Caluwe, H. de., Biogeochemistry, 1992, 15, 175–190.
21. Lorenz, F. J. and Rogler, G. A., J. Range Manage., 1967, 20, 129–132.
22. Singh, J. S. and Yadava, P. S., Ecol. Monogr., 1974, 44, 351–376.
23. Sant, H. R. and Singh, R. S. P., Indian J. Ecol., 1982, 9, 197–202.
24. Silver, W. L. and Vogt, K. A., J. Ecol., 1993, 81, 729–738.
25. Ewel, J., Berish, C., Brown, B., Price, N. and Raich, J., Ecology, 1981, 62, 816–829.
26. Visalakshi, N., J. Biosci., 1994, 19, 103–116.
27. Singh, K. P. and Singh, R. P., Oikos, 1981, 37, 88–92.
28. Kershaw, K. A., Quantitative and Dynamic Ecology, Arnold, London, 1973, 2nd edn.
29. Mitchell, D. T., Coley, P. G. F., Webb, S. and Allsopp, N., J. Ecol., 1986, 74, 977–993.
30. Allen, S. E., Grimshaw, H. M., Parkinson, J. A. and Quarmby, G., Chemical Analysis of Ecological Materials, Blackwell, Oxford, 1974, p. 565.
31. Kangas, P., Biotropica, 1992, 24, 463–465.

Contrasting ultrastructures in the eggshells of olive ridley turtles, *Lepidochelys olivacea*, from Gahirmatha, Orissa

G. Sahoo, B. K. Mohapatra, R. K. Sahoo and P. Mohanti-Hejmadi*

Regional Research Laboratory, Bhubaneswar 751 013, India
*Department of Zoology, Utkal University, Bhubaneswar 751 004, India

Normal and abnormal sized eggshells of olive ridley turtles, *Lepidochelys olivacea*, from Gahirmatha have been studied for ultrastructural characteristics. Fresh eggshells exhibit nodular shell units on the upper surface followed by a membranous network of fibres and a thin shell membrane. These structures are not seen in hatched eggshells as the calcium is resorbed during embryonic development. Abnormal shells do not show any such structure excepting disoriented fibres in its lower surface due to inadequate calcification.

THE eggshell is a mediating boundary and together with other features it contributes to the isolation of the embryo from the nesting environment. Modifications of the shell structure have probably evolved for functional

smoothness and evolutionary adaptations. It might reasonably be anticipated that the eggshells of marine turtles would show even greater structural variation concomitant with the laying habits, temperature and so on. The marine turtle eggshells fall within the category of 'softshelled' eggs[1,2]. Extensive studies have been carried out on the ultrastructure of green[3,4], leatherback[5-7], loggerhead[2] and Kemp's ridley[1] sea turtles. Although olive ridley, *Lepidochelys olivacea*, is the most abundant sea turtle and Gahirmatha (Orissa) is the world's largest rookery of this species[8], no report on its ultrastructural morphology is available in the literature. Here we describe briefly the eggshell ultrastructures of these turtles.

Eggshells from normal (both fresh and hatched; circular, 36.1–39.5 mm diameter) and abnormal (oblong, very small: 8.5 × 11.5 mm, 3.26 g; and very large: 46.5 × 80.0 mm, 58.32 g) eggs were collected from the Gahirmatha nesting rookery during the 1993 nesting season and were subjected to XRD (Philips PW-1700) and SEM (JEOL, JSM 35 CF) studies.

In general, the olive ridley turtle eggshell contains solely aragonite ($CaCO_3$) crystallites. This is in contrast to that of other marine turtles where traces of calcite and vaterite along with aragonite have been reported. In case of leatherbacks, all the three crystal forms of calcium carbonate (calcite, aragonite and vaterite)[5] and in green turtles, both calcite and aragonite are present[3].

In a normal eggshell of the fresh kind, the outer inorganic surface exhibits a rough granular structure. The nodular shell units are very distinct which is in contrast to those reported for *Caretta caretta*[9] or *Dermochelys coriacea*[10] where they are ill-defined. Two types of structures are observed on this surface: micronodules which give the coarse granular appearance; and spicules which vary in size and are also arranged in different planes (Figure 1 a). The arrangement of the shell units is not compact and thus results in pore-like open spaces of varying sizes. Such type of pores are present in loggerhead[9] and Kemp's ridleys[1], but are absent in leatherbacks[10]. The pores are interrupted by the spicules and are meant for gaseous exchange and water absorption[11] which is essential during embryonic development. The inorganic surface is followed by a layer of inter-
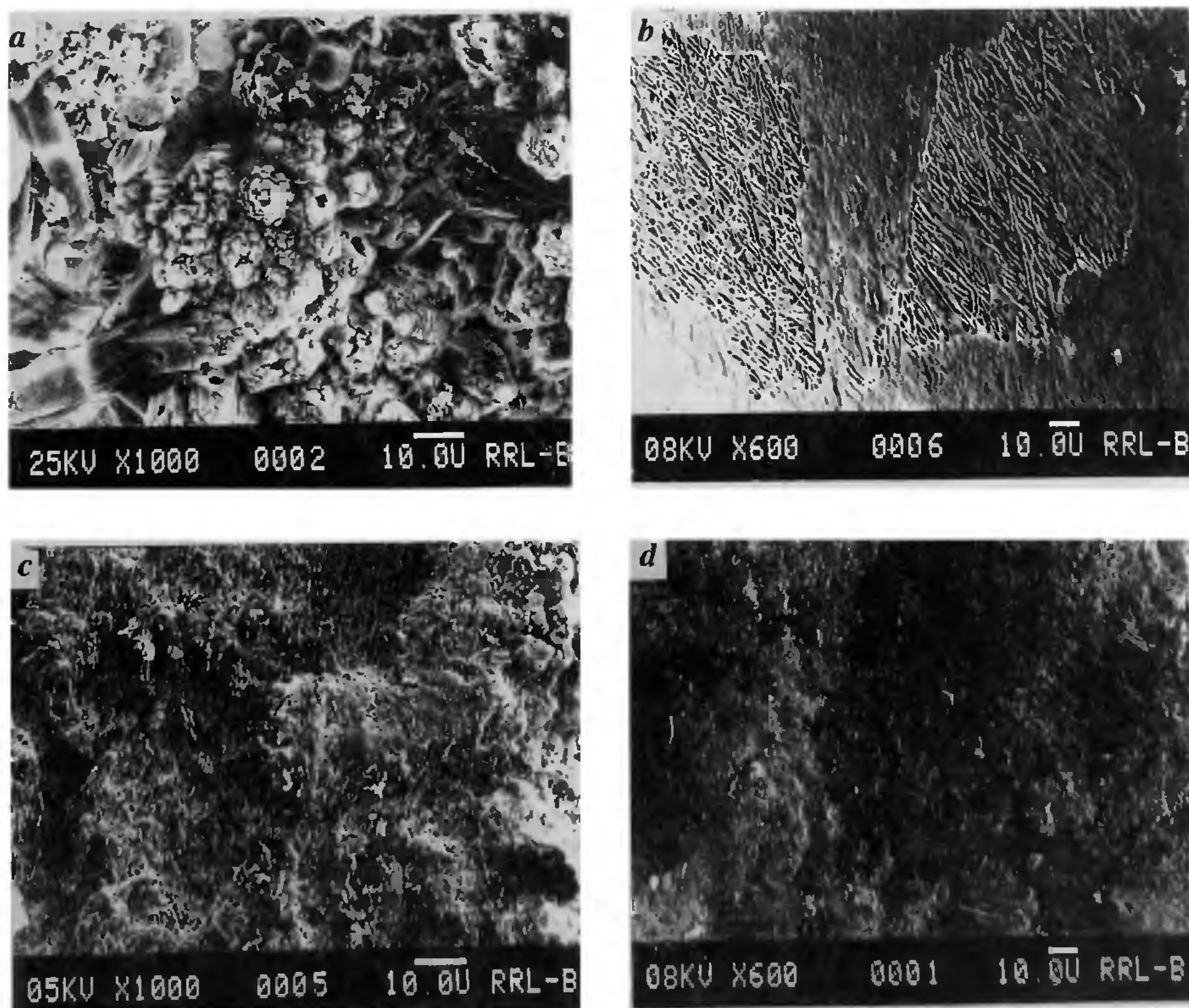

Figure 1 a–d. Electron micrographs of normal eggshells of olive ridley turtles. a, Upper surface of fresh eggshell showing loosely arranged crystallites and spicules. b, Lower surface of fresh eggshell showing the fibrilar network and a thin shell membrane covering it. c, Upper surface of hatched eggshell. d, Lower surface of hatched eggshell.

woven fibrils, which are organic in nature, frequently branched, ramifide, exhibit a netted structure (Figure 1 b) and these fibres are similar in composition to avian eggshells[12]. Specific positions of these fibres probably act as nucleation sites on which the calcification begins[3]. These fibres are flexible and stretch when the egg diameter increases during incubation. Below the fibres there is a very thin organic basement membrane through which the internal fibrilar network is visible in ventral view (Figure 1 b). In olive ridley, like other chelonians, the shell membrane is single-layered and not paired as observed in bird's eggshell[12].

The outer surface of a hatched eggshell, even at higher magnification, does not show any organized structure and is rather homogenous with fused granules (Figure 1 c). The surface is obscured by minute spherical or amorphous masses. Partial alteration of hatched eggshells of *Caretta caretta* has also been reported by Schleich and Kastle[2]. This sort of disruption of structures is due to the resorption of calcium from the shell.

Similar alteration in eggshell ultrastructure in developing embryos due to utilization of calcium reserve has been reported in domestic fowl[13] and leathery (*Dermochelys coriacea*)[14] and softshell turtles (*Trionyx spiniferus*)[15]. The fibrilar network of the inner surface is not visible as it is covered by a dense, structureless thick layer (Figure 1 d).

The abnormality of the eggs is mainly in the form and extreme size, small or large or in the form of two or more eggs joined together[16]. These eggs never hatch. There is no report on the structure of abnormal eggshells of any marine turtle. In case of abnormally small eggs, the calcification of the shell is incomplete and present in the form of a very thin layer through which the inner surface is clearly visible (Figure 2 a). No crystal growth is discernible on its upper inorganic surface. The lower organic surface consists of widely spaced fibres and microglobules without the basement membrane. The fibres are of uneven thickness, irregularly arranged and show branching (Figure 2 b). Tiny globules
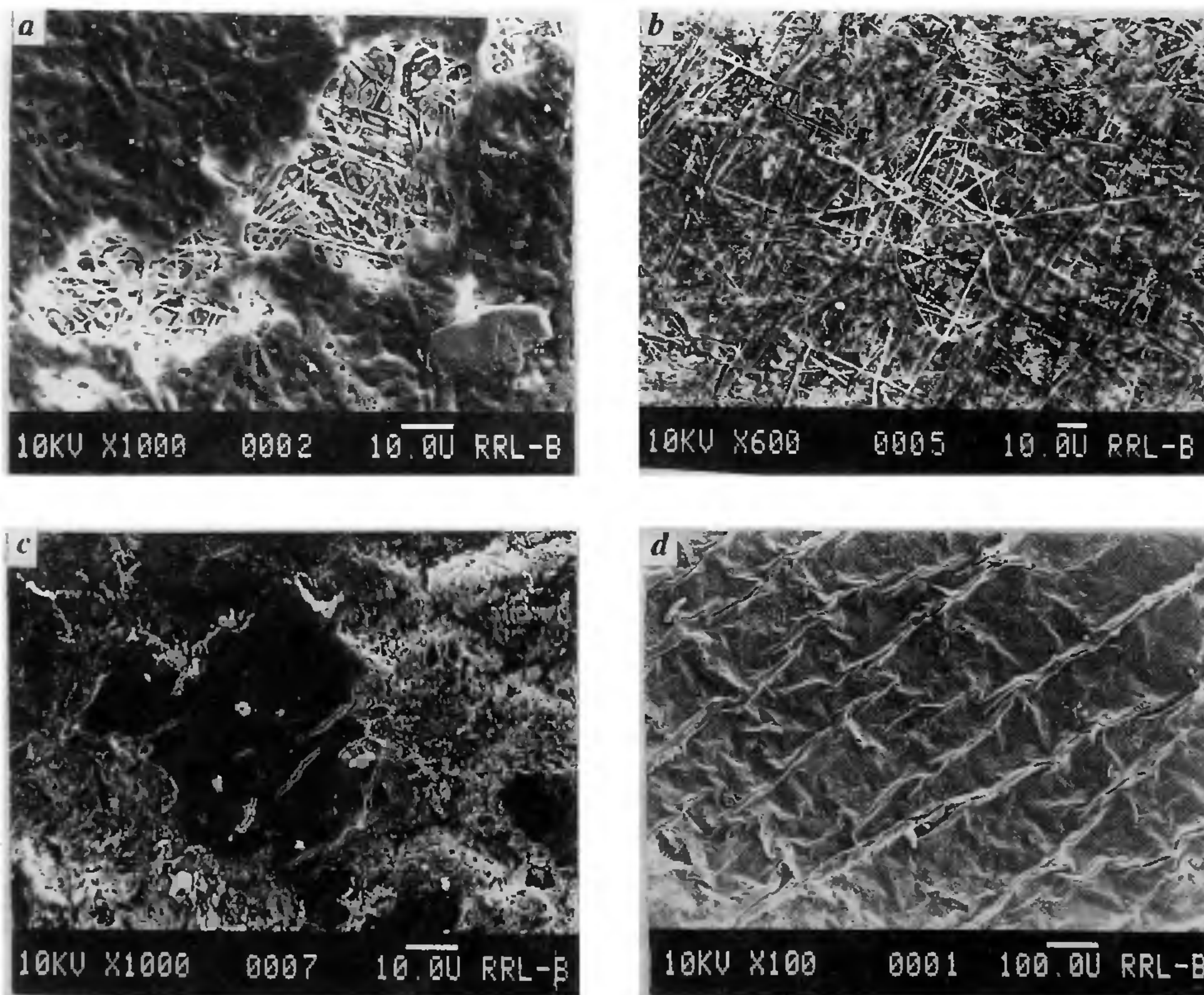

Figure 2 a–d. Electron micrographs of abnormal eggshells of olive ridley turtles. a, Upper surface of a very small egg shell. Note how a very thin, almost transparent, calcareous layer covers the fibrous network. b, Lower surface of a very small eggshell. Disoriented fibrils with microglobules are seen. c, Upper surface of a very large eggshell. Remnants of calcareous layer (black) is seen over a spongy mass. Pores appear thin and elongated due to interconnection. d, Lower surface of a very large eggshell in which the parallel bifurcated fibrils are arranged in a trellis pattern.

are associated with these fibres and seem to be the nucleation centres for crystal growth. The abnormally large eggshells are also without any crystal or nodular structure on its outer surface. Remnants of a calcareous layer as small patches are seen over a spongy mass (Figure 2 c). Pores are interconnected and appear elongated. The fibrilar network in the lower surface is remarkable. The fibres are bifurcated and run more or less parallel to each other in the form of a trellis pattern (Figure 2 d). These parallel fibres are further perpendicularly crossed by similar fibrils at regular interval.

Thus, the olive ridley eggshells, though appearing alike externally, show contrasting ultrastructures. Dissimilar structure is observed not only amongst normal and abnormal shells but also between fresh and hatched ones. Fresh shells consist of an outer inorganic calcareous layer with nodular units and an inner organic layer which is represented by a profuse mat of fibrilar network. The eggshells undergo drastic ultrastructural changes during incubation like those of Aves and other turtles as a result of resorption of calcium from the shell. The abnormal eggs do not show any crystalline structure probably due to improper growth and inadequate calcium deposition.

1. Hirsch, K. F., *Copeia*, 1983, **2**, 382–397.
2. Schleich, H. and Kastle, W., *Reptile Egg Shells*, SEM Atlas, G. S. Verlag, Stuttgart, 1988, pp. 119.
3. Solomon, S. E. and Baird, T., *J. Exp. Mar. Biol. Ecol.*, 1976, **22**, 145–160.
4. Solomon, S. E. and Baird, T., *J. Exp. Mar. Biol. Ecol.*, 1977, **27**, 83–92.
5. Chan, E. H. and Solomon, S. E., *Anim. Technol.*, 1989, **40**, 91–102.
6. Solomon, S. E. and Watt, J. M., *Anim. Technol.*, 1985, **36**, 19–27.
7. Solomon, S. E. and Tippett, R., *Anim. Technol.*, 1987, **38**, 73–79.
8. Bustard, H. R., Tiger Paper, 1976, 3.
9. Packard, M. J. and Hirsch, K. F., *Scanning Electron Microscopy*, 1986, **IV**, 1581–1590.
10. Chan, E. H., Saleh, H. U. and Liew, H. C., *Pertanicka*, 1985, **8**, 265–271.
11. Tracy, C. R., Packard, G. C. and Packard, M. J., *Physiol. Zool.*, 1978, **51**, 378–387.
12. Becking, J. H., *The Ibis*, 1975, **117**, 143–151.
13. Johnston, P. M. and Comar, C. L., *Am. J. Physiol.*, 1955, **183**, 365–370.
14. Simkiss, K., *Comp. Biochem. Physiol.*, 1962, **7**, 71–79.
15. Packard, M. J. and Packard, G. C., *J. Morphol.*, 1979, **159**, 131–144.
16. Dash, M. C. and Kar, C. S., *Turtle Paradise Gahirmatha*, Interprint, New Delhi, 1990, pp. 295.