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Cataposit 44 (Shipley) in water. After 10 min the fibers were recovered by filtration and suspended in a wash solution of Cataprep 404. Filtration was repeated and followed with 4 washes with 1 L of water. The final filter cake was freeze-dried to yield a fine light-gray powder. The electroless copper-plating bath was Fidelity 1025. Dry fibers were added at the concentrations specified in the text and subjected to continuous stirring. Once exhausted the reaction mixture was filtered, and the fibers washed copiously with water and freeze-dried.

Absolute density was determined by water displacement. A known mass of fibers was placed in a pre-weighed graduated cylinder. Water was added to a total known volume (bubbles were removed by subjecting the cylinder to vacuum). The volume of water was determined by re-weighing the cylinder. The volume occupied by the fibers was determined by subtracting the volume of water from the total volume.

An approximation of the conductivity of the metal plated onto cellulose was determined by use of a Spectra/Por cellulose membrane (Spectrum) as a surrogate material. Measurement of the conductivity of individual fibers is impractical, and the conductivity of bulk fiber is dominated by contact resistance. A membrane of 10 mm width and 8.5 cm long was plated with electroless copper essentially as described above. The final thickness was 30.5 μm, with a copper layer on both sides of less than 100 nm (as determined by weight gain). The resistance of this membrane was 3.5 Ω, which corresponds to a conductivity of $8.1 \times 10^5\ \Omega^{-1}\,m^{-1}$.

Sample composites were fabricated by adding the requisite mass of fibers to polyurethane LS-40 (BJB Enterprises) to yield the desired volume percentage. Moldings were made between two flat plates, with shims to determine the thickness. Samples were cured for 24 h at room temperature. Electromagnetic measurements were conducted with a Hewlett-Packard 8510 Network Analyzer and permittivities were calculated by the Nicholson-Ross technique [14]. Samples were 1.27 mm thick and toroidal, with an inner diameter of 3 mm and an outer diameter of 7 mm and measured in a coaxial cable arrangement.

DC conductivity of the composites was measured across the thickness of the samples (1.27 mm) between metal plates of 5 cm by 1.8 cm in dimension. Measurements were made with a Keithley 194 A Digital Multimeter. The limit of detection was approximately $3 \times 10^{-10}\ \Omega^{-1}\,m^{-1}$.

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Monodisperse Polyelectrolyte-Supported Asymmetric Lipid-Bilayer Vesicles**

By *Kiyofumi Katagiri* and *Frank Caruso**

It has been well known for more than thirty years that lipid molecules can spontaneously aggregate into spherically closed bilayer membranes in water.[1–7] These lipid vesicles, known as liposomes, have been extensively studied as cell-membrane models, drug carriers, and gene-transfection agents.[6,7] Although liposomes offer a number of advantages over other hollow particles with respect to physical properties, it is often difficult to control their size, monodispersity, and lipid-layer number (e.g., unilamellar or multilamellar) when conventional preparation methods (sonication,[6] rapid injection,[8] swelling,[9] reverse evaporation,[10] electroformation,[11] etc.) are employed. Additionally, the preparation of asymmetric bilayer vesicles, i.e., vesicles with different lipid compositions in the inner and outer layers, represents a significant challenge. In biological systems, cell membranes have an asymmetrical distribution of phospholipids: the aminophospholipids, such as phosphatidylserine, are enriched on the inner leaflet, while the outer leaflet has more phosphatidylcholine.[12] This asymmetry is a key feature of cell plasma membranes for exocytosis, intracellular fusion, lipid–protein interactions, and signal transduction.[13–16] A family of translocase enzymes (flippases and floppases) is dedicated to maintaining the correct distribution of lipids.[17] Therefore, the development of techniques for the preparation of artificial asymmetric lipid vesicles is challenging, not only for the construction of artificial cell-membrane models, but also for advances in the fabrication of drug- and gene-delivery systems. Partial symmetry can be achieved by altering the distribution of specific lipids.[12,18–20] For example, Smith and co-workers have reported synthetic translocases that facilitate the inward and outward translocation of phospholipids in liposomes and cell membranes.[12,18] On the other hand, there are only a few reports that describe the preparation of asymmetric liposomes consisting of com-

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pletely different types of lipids on the inner and outer leaflets. The groups of Xiao[21] and Weitz[22] have employed the inverted-emulsion technique to prepare asymmetric vesicles with inner and outer monolayers composed of different amphiphiles. This technique affords asymmetric unilamellar vesicles with diameters that can be adjusted between 30 nm and 1 μm; however, relatively broad size distributions of the vesicles are obtained.

Herein, we report the preparation of monodisperse vesicles comprising asymmetric lipid bilayers supported on colloidal particles coated with polyelectrolytes (PEs). Both unilamellar and multilamellar lipid bilayers are formed. As schematically illustrated in Figure 1, the procedure for preparation of the PE-supported asymmetric bilayers involves three main steps: 1) application of the versatile and widely employed layer-by-layer (LbL) technique[23,24] to deposit oppositely charged PEs onto melamine formaldehyde (MF) particles with narrow size distributions;[25] 2) deposition of an "inner" lipid monolayer from organic solution onto the oppositely charged PE-multilayer coated particles; and 3) the formation of an "outer" lipid-monolayer coating by contacting small unilamellar vesicles (SUVs) with the hydrophobized (lipid-monolayer coated) colloids in water. We also demonstrate that multilayer asymmetric lipid coatings, interspersed with PEs, can be formed by repetition of these three steps. Poly(diallyldimethylammonium chloride) (PDDA) and poly(sodium 4-styrenesulfonate) (PSS) were employed as the polycation and polyanion, respectively, while dimethyldioctadecylammonium bromide (DDAB) and dihexadecyl phosphate, sodium salt (DHP) were used as the cationic and anionic lipids, respectively. Although a number of recent studies have focused on the formation of various lipid layers on PE-coated supports,[26–29] the preparation of asymmetric supported lipid-bilayer membranes (either on colloids or planar surfaces) via the LbL method has not been reported.

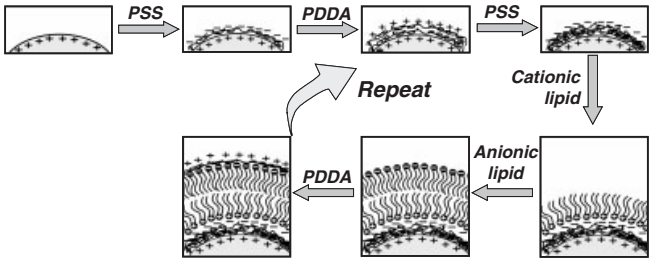

Figure 1. Schematic representation of the formation of PE-supported asymmetric lipid bilayer membranes via the layer-by-layer assembly method using poly(sodium 4-styrenesulfonate) (PSS) and poly(diallyldimethylammonium chloride) (PDDA). The PE layers and the outer lipid monolayer are deposited from aqueous solutions, while the inner lipid monolayer is adsorbed from an organic solvent.

Monodisperse asymmetric lipid-coated colloids were prepared by depositing two lipids (DDAB and DHP) which can form the inner and outer monolayers. Following formation of the PE/asymmetric lipid bilayer membranes, the core MF templates were removed by acid treatment to obtain vesicular particles. Alternatively, the lipid-coating processes could be conducted on preformed PE hollow capsules.

Multilayer film formation on colloids was monitored by microelectrophoresis measurements. Figure 2a shows the ζ-potential as a function of PE and lipid layer number for the positively charged MF particles coated with PSS/PDDA layers and a DDAB/DHP asymmetric bilayer. The positively charged (uncoated) MF particles (diameter = 1.09 μm) yielded a ζ-potential of ca. +55 mV in water. The presence of PSS on the particle surface (layer number 1) causes a reversal in ζ-potential to about –45 mV. Alternating ζ-potentials were obtained with the subsequent deposition of cationic PDDA and anionic PSS, suggesting stepwise formation of PSS/PDDA/PSS on the particles. At layer number 4, the cationic DDAB was adsorbed onto the PSS-terminated particles. These coated particles yielded a ζ-potential of close to 0 mV. (Ethanol was used for a dispersant in this case.) The sign of the ζ-potential is in agreement with that expected for the formation of a DDAB layer on the particles, with the hydropho-

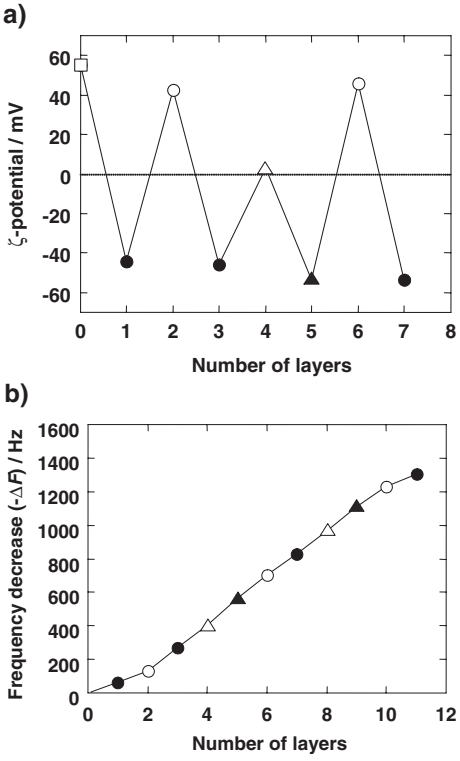

Figure 2. a) ζ-potential and b) quartz crystal microbalance (QCM) frequency decrease ($-\Delta F$) for the LbL deposition of PEs and lipids: bare MF core (□); PSS (●, 1.0 mg mL^{-1} in 0.5 M NaCl); PDDA (○, 1.0 mg mL^{-1} in 0.5 M NaCl); DDAB (△, 1.0 mM in EtOH); DHP (▲, 1.0 mM in H_2O). ζ-potential measurements were conducted in deionized water, except for the 4th layer (inner lipid coating), where the particles were suspended in ethanol. QCM frequencies were recorded in air.

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bic alkyl tails of DDAB extending outwards from the particle surface. We experimentally verified that PSS and DDAB molecules in bulk solution associate to form a complex in ethanol solution (data not shown). At layer number 5, the ζ-potential shifts to negative values (–53 mV) since anionic DHP was deposited on the surface. In this case, a DHP layer forms on the DDAB-terminated particles via hydrophobic interactions, with the hydrophilic head groups of DHP forming the outer surface of the particles. Alternating ζ-potentials are obtained with further deposition of PDDA/PSS on the DHP-terminated particles. The above data suggests that cyclic multilayer formation of PDDA/PSS/DDAB/DHP can be carried out. ζ-potential measurements also showed that the system with DHP as the inner leaflet and DDAB as the outer leaflet could be prepared on the PE-coated particles (data not shown).

In order to quantify the amount of PE and lipid deposited with each adsorption step, a quartz crystal microbalance (QCM) was employed to follow the assembly process. The QCM frequency, $-\Delta F$, decreases proportionally with an increase in mass deposited on the crystal surface according to the Sauerbrey equation.[30,31] QCM electrodes were alternately immersed for 20 min in aqueous PSS and PDDA solutions (each containing 0.5 M NaCl), an ethanolic solution of DDAB, and a SUV dispersion of DHP, with intermediate washing and drying under a nitrogen stream following deposition of each layer. This process was periodically interrupted to measure the QCM resonance frequency. The values of $-\Delta F$ observed at each step of the assembly of the PSS/PDDA/$DDAB_{inner}$/DHP_{outer} system are shown in Figure 2b. Regular film growth was observed for this system, and the average $-\Delta F$ measured for the lipid adsorption steps was ca. 130 Hz. QCM responses of assembled lipid films can be enhanced by hydration of the lipid films.[32] When taking this effect into account, the measured value is in good agreement with that calculated for a hexagonally packed lipid monolayer of DDAB or DHP ($-\Delta F \sim 100$ Hz).[33] This suggests that both the DDAB and DHP were mainly deposited as monolayers.

The deposition of lipid membranes onto PE-coated particles was also examined by fluorescence microscopy. A fluorescence microscopy image of the particles comprising MF (diameter = 2.98 μm) cores coated with PSS/PDDA/PSS/DDAB/DHP/PDDA/PSS is shown in Figure 3a. *N*-(7-Nitro-2,1,3-benzoxadiazol-4-yl)-1,2-dipalmitoyl-*sn*-glycero-3-phosphoethanolamine (NBD-DPPE) was employed as the anionic lipid probe dye (3 mol-%) in the anionic DHP layer. Homogeneous fluorescence is seen originating from the surface of the PE/lipid-coated particles. Similar images were obtained for particles coated with an additional one or two PDDA/PSS/DDAB/DHP layers, and for the reverse lipid system (i.e., PSS/PDDA/DHP/DDAB). This proves, within the limits of resolution of the fluorescence microscopy technique, the presence and homogenous distribution of lipids on the particle surface. Moreover, similar images were obtained for the PE-supported asymmetric bilayer vesicles after removal of the MF core (images not shown). This suggests that the core-removal process did not cause macroscopic destruction of the

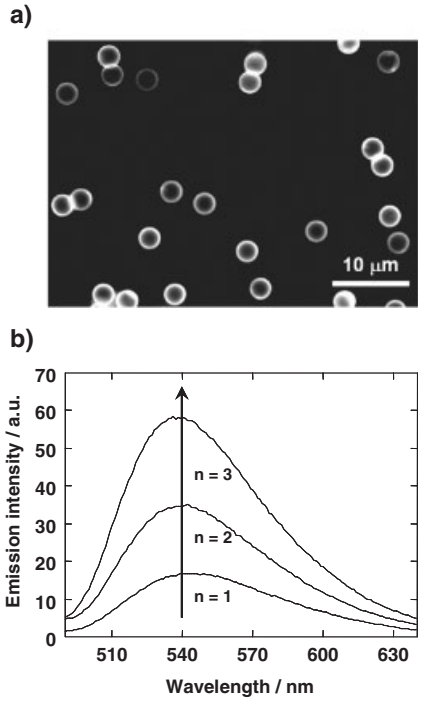

Figure 3. a) Fluorescence microscopy image of MF particles (2.98 μm diameter) coated with a PSS/PDDA/PSS/DDAB/DHP/PDDA/PSS multilayer, and b) fluorescence spectra of a dispersion of MF particles coated with PSS/PDDA/PSS/$(DDAB/DHP/PDDA/PSS)_n$ ($n = 1$, 2, and 3). All samples contain 3 mol-% NBD-DPPE in the DHP layer. Excitation: 470 nm.

PE/lipid-bilayer membrane. Figure 3b shows fluorescence spectra of the MF particles coated with PSS/PDDA/PSS/$(DDAB/DHP/PDDA/PSS)_n$ multilayers (where $n = 1$, 2, or 3) dispersed in water. Again, NBD-DPPE was employed as the probe dye (ca. 3 mol-%) in the DHP monolayer. NBD-DPPE emission with a maximum wavelength of 540 nm was observed for each system. The linear increase in the maximum emission intensity observed for the $n = 1$, 2, and 3 coatings indicates that multiple asymmetric bilayers were deposited. It is noted that when a SUV dispersion of DHP was directly contacted with the PSS-terminated MF particles, no emission was detected. This confirms that the DHP layer deposits onto the particles when DDAB forms the outermost layer. The above data reveal that not only single asymmetric bilayers can be formed, but that multiple-bilayer membranes with controlled lipid layer number can be prepared by the LbL technique. This potentially represents an alternative and facile means to control the permeability properties of PE multilayer-based thin films and capsules.[25d]

We employed a fluorescence quenching assay to determine the distribution of probe lipids between the inner and outer leaflets of the bilayer membrane.[19,34] NBD-DPPE was again used as a probe lipid in the DHP monolayer. Sodium hydrosulfite ($Na_2S_2O_4$) was adopted as a fluorescence quencher for the NBD-derivative probe. The NBD group is reduced to a 7-amino-2,1,3-benzoxadiazol-4-yl group (ABD group) with $Na_2S_2O_4$.[19] The fluorescence intensity of ABD derivatives is

less than 0.2 % compared with NBD-derivatives. The fluorescence emission of the asymmetric lipid-bilayer-coated particle dispersion was measured before and after addition of $Na_2S_2O_4$ solution. The addition of the quencher to the dispersion causes a reduction in the emission of only the probe on the outer monolayer of the membrane, since the quencher molecule cannot diffuse across the lipid-bilayer membrane.[19,22] Figure 4 shows the relative fluorescence intensity as a function of time for reaction of $Na_2S_2O_4$ with the asymmetric lipid-bilayer-coated particles. In the case of the DHP

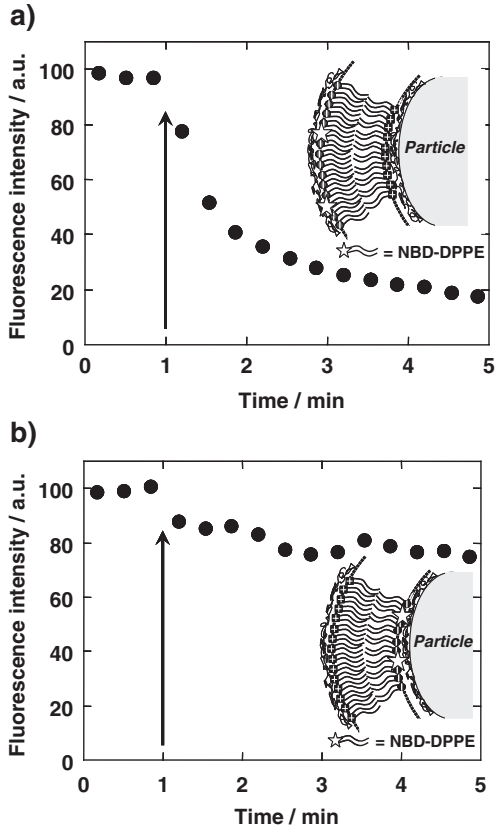

Figure 4. Time course of fluorescence intensity of PE/asymmetric-lipid-bilayer-coated MF particles. The addition of $Na_2S_2O_4$ is indicated by the arrow (ca. 1 min). a) MF particles (2.98 μm diameter) coated with a PSS/PDDA/PSS/DDAB/DHP/PDDA/PSS multilayer. NBD-DPPE is embedded in the outer layer of DHP. b) MF particles (2.98 μm diameter) coated with a PSS/PDDA/DHP/DDAB/PSS/PDDA multilayer. NBD-DPPE is embedded in the inner layer of DHP. All samples contain 3 mol-% NBD-DPPE in the DHP layer. Excitation = 470 nm; emission = 540 nm.

layer as the outer leaflet, a rapid decrease in fluorescence (80 % within 4 min) was observed upon the addition of $Na_2S_2O_4$ (Fig. 4a). This indicates that a majority of the NBD-DPPE resides in the outer leaflet of the DHP layer. In contrast, the fluorescence intensity decreased by less than 20 % upon the addition of $Na_2S_2O_4$ when NBD-DPPE was incorporated in the DHP inner layer in the DHP/DDAB system (Fig. 4b). These results confirm the formation of an asymmetric bilayer and are similar to those reported by Weitz and co-workers for asymmetric lipid bilayers prepared by the inverted-emulsion template method.[22] The asymmetry of the lipid bilayers was retained even after 72 h, as assessed by the fluorescence-quenching assay which revealed no changes in the emission properties of the two systems shown in Figure 4. This suggests the absence of lipid flip-flop in these systems. The formation of PE/lipid complexes in the multilayer membrane is likely to be responsible for preventing lipid flip-flop. It was also observed that the emission intensity of NBD-DPPE did not change upon the addition of a solution of the detergent Triton X-100 (TX-100), which is widely used as an agent for lysing lipid membranes.[5,35] Previous work has shown that probe molecules embedded in an inner lipid leaflet are readily quenched when the asymmetric lipid bilayer was lysed by TX-100.[22] In our system, the resistance of the membrane to TX-100 is most probably due to the enhanced stability arising from the PE layers that encase both the inner and outer lipid-bilayer membrane. The PEs act as a supporting skeleton, reinforcing the morphological stability of the lipid membrane by complexation with the lipid molecules.

In conclusion, we have demonstrated the preparation of monodisperse PE-supported asymmetric lipid bilayers by LbL-colloid templating. The diameters of the PE-supported lipid-bilayer vesicles correspond closely to those of the original template core particles, thereby providing a means to control their size. Notably, both unilamellar and multilamellar vesicles (with tailored layer numbers) are readily obtained via this technique. The asymmetry retained by the lipid bilayers is attributed to their complexation with the PE support, which also enhances the bilayer morphological stability against surfactants. The ability to control the type of lipid in each leaflet to form asymmetric lipid bilayers opens new opportunities for the construction of artificial cell-membrane models or drug-delivery systems with designed compositions and structures. Furthermore, the possibility to orient dipole moments of dye molecules embedded within asymmetric lipid bilayers potentially makes these systems interesting for nonlinear optical (including second-harmonic generation) studies.[36,37] Our current work is focused on the preparation of free-standing liposomes of asymmetric bilayers by employing decomposable PEs.

Experimental

Materials: Unless otherwise stated, all reagents and chemicals were obtained commercially and used without further purification. Positively charged, weakly crosslinked melamine formaldehyde (MF) particles (with diameters of 1.09 μm or 2.98 μm) were purchased from Microparticles GmbH, Berlin, Germany. Poly(diallyldimethylammonium chloride) (PDDA, weight-average molecular weight $M_w = 100\,000$–$200\,000$ g mol^{-1}), poly(sodium 4-styrenesulfonate) (PSS, $M_w = 70\,000$ g mol^{-1}), dihexadecyl phosphate, sodium salt (DHP), dimethyldioctadecylammonium bromide (DDAB), *N*-(7-Nitro-2,1,3-benzoxadiazol-4-yl)-1,2-dipalmitoyl-*sn*-glycero-3-phosphoethanolamine (NBD-DPPE), Triton X-100 (TX-100), 2-(cyclohexylamino)ethanesulfonic acid (CHES), sodium 3-mercapto-1-propanesulfonate, and

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sodium hydrosulfite ($Na_2S_2O_4$) were purchased from Sigma–Aldrich. Ethanol (EtOH), tetrahydrofuran (THF), chloroform, sodium hydroxide, and hydrochloric acid were obtained from Merck. EtOH was distilled before use. The water used in all experiments was prepared in a Millipore Milli-Q Synergy purification system and had a resistivity of 18.2 MΩ cm.

Liposome Preparation: Liposomal dispersions of anionic DHP and cationic DDAB were prepared as follows. The lipids were dissolved in $CHCl_3$ and films were formed on the inside of vials by drying with a N_2 stream. The films were dried under vacuum overnight. Hydration of the lipid films was then performed at room temperature with deionized water. The concentration of lipid was fixed at 1.0 mM. Multilamellar vesicles were generated using a vortex mixer, and these liposomal dispersions were then ultrasonicated with a bath-type sonicator (70 W) for 2 h at a temperature above the phase-transition points of the lipids to obtain small unilamellar vesicles (SUVs).

Polyelectrolyte (PE) and Lipid Coating on Colloids: MF particles were alternately suspended in an aqueous PSS solution (1 mg mL^{-1}) containing 0.5 M NaCl and an aqueous PDDA solution (1 mg mL^{-1}) containing 0.5 M NaCl. After each PE-adsorption step, the samples were washed three times with deionized water. The coating procedure has been detailed elsewhere [25]. The aqueous dispersions of PE-coated MF particles were centrifuged (2000*g*, 3 min), and then the supernatant water was removed. The samples were washed three times with deionized water. Lipid-monolayer deposition on the PE layers (inner leaflet) was performed using lipid solutions in organic solvents. Ethanol and THF were used for deposition of DDAB and DHP, respectively. The lipid concentration was fixed at 1.0 mM. The PE-coated MF particles were suspended in an oppositely charged lipid solution with organic solvent, incubated for 20 min, and then the samples were washed three times with organic solvent. The particles were then centrifuged (2000*g*, 3 min) and the organic solvent was removed. Deposition of the outer lipid layer was performed by incubating for 20 min in a SUV dispersion of a different lipid from the one used to form the inner leaflet. Following incubation, the samples were washed three times with deionized water.

Hollow-Capsule Fabrication: Core dissolution of MF was accomplished by exposure of the coated particles (suspended in 0.5 mL water) to 0.1 M HCl solution (1 mL) for 30 min. The hollow capsules were centrifuged and redispersed in deionized water. To ensure complete core removal, this process of HCl exposure/redispersion was repeated a further two times. Finally, the hollow capsules were washed three times with deionized water.

Microelectrophoresis: PE and lipid coating of the particles were qualitatively monitored by measuring the ζ-potentials of the coated particles with an electrophoretic light scatterer equipped with a laser Doppler system (Malvern, Zetasizer 2000). An average of five measurements at the stationary level was taken for each data point. The ζ-potential measurements were conducted in deionized water, except for lipid-monolayer coated (hydrophobized) particles, in which the particles were suspended in EtOH.

Quartz Crystal Microbalance (QCM) Measurements: QCM electrodes (Kyushu Dentsu Co. Ltd., Japan) were used in a home-built system [31b] to follow the layer-by-layer (LbL) deposition of the PEs and lipids on planar surfaces. The AT-cut QCM resonator was covered by vapor-deposited gold on both faces, and the resonance frequency was 9 MHz. The QCM frequency ($-\Delta F$) decreases proportionally with increasing mass [30,31]. Since the QCM frequency response for lipid membranes can drastically change at the lipid phase-transition temperature when measured in water [38], we recorded the QCM frequency changes in air and below the phase-transition temperatures of the lipids. A precursor film of a self-assembled monolayer of sodium 3-mercapto-1-propanesulfonate was followed by five polyelectrolyte layers deposited on the QCM resonator by the alternate assembly of PDDA and PSS. All experiments were conducted at 20 °C, and QCM frequencies were recorded in air.

Fluorescence Measurements: A Cary Eclipse fluorescence spectrophotometer (Varian) was used for fluorescence spectra measurements. Fluorescence micrographs were acquired with an Olympus IX71 microscope. 3 mol-% NBD-DPPE was used as the lipid fluorescent probe. The excitation wavelength was set at 470 nm.

Fluorescence Quenching Assay for Measuring Lipid Asymmetry: The assay was employed to confirm the distribution of the lipid fluorescent probe between the inner and outer monolayers. The coated particles were prepared with 3 mol-% of NBD-DPPE on either the inner or outer leaflet. The particles were suspended in 2 mL of buffer solution (5 mM CHES at pH 9.0). The fluorescence emission of these solutions was recorded over time at an excitation wavelength of 470 nm and an emission wavelength of 540 nm. After a constant baseline fluorescence was obtained (1 min), 50 μL of quencher solution (1 M $Na_2S_2O_4$ in 5 mM CHES at pH 9.0) was added, and the reaction was followed by monitoring the decrease in the fluorescence emission of NBD-DPPE. 50 μL of TX-100 solution (50 mM) was added after the fluorescence reached a plateau (~4 min after the addition of $Na_2S_2O_4$).

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[33] The Sauerbrey relationship between adsorbed mass, M [g], and frequency change, ΔF [Hz], in this system is $-\Delta F = 1.83 \times 10^8 \times M/A$, where A is the apparent area of the QCM. By using the limiting molecular area A' of the lipid molecule, 0.4 nm^2, and the molecular weight M_w of DHP and DDAB, 568.8 and 631.0 g mol^{-1}, respectively, the adsorbed mass for a monolayer can be calculated according to $M = (2A/A')/(N_A \times M_w)$, where N_A is Avogadro's number.
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A Disordered Carbon as a Novel Anode Material in Lithium-Ion Cells**

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In recent years, interest in lithium-ion batteries has increased constantly. Due to the high value of energy density and the long cycle life, these batteries have become the power sources of choice for consumer electronic devices such as cellular phones and laptop computers. In addition, lithium-ion batteries are expected to be a major breakthrough in the hybrid vehicle field. Despite their impressive commercial success, lithium-ion batteries need to be further improved in order to meet ongoing market innovations. Accordingly, extensive research and development effort is presently directed towards the characterization of new, improved-performance electrode and electrolyte materials.

With respect to the cathode, the main goal is to replace the present cobalt-based oxides with cheaper and more environmentally compatible materials. A promising candidate is the olivine-type lithium iron phosphate, which is presently attracting a great deal of attention.[1]

The current trend in the electrolyte area is to replace the conventional liquid electrolytes with more reliable and more easily processable polymer electrolytes. Important results have been achieved with gel-type polymer electrolytes,[2] and, in prospect, with morphologically modified, nanocomposite polymer electrolytes.[3]

At present, graphite is widely used as the anode in commercial cells due to its facile lithium insertion–removal staging electrochemical process and its low cost. However, the lithium storage capacity of graphite is limited to 372 mAh g^{-1}, as associated with its maximum LiC_6 stage.[4] Very appealing alternative anode materials are lithium metal alloys, owing to their impressively high values of theoretical specific capacity, which exceed that of graphite by orders of magnitude.[5] However, although great progress has been achieved recently, for example by moving to nanostructures,[6,7] the cycle life of these anodes is still limited by the volume changes that accompany the lithium acceptance–lithium release process.[8]

Thus, in the immediate search for high-capacity anode materials, attention may be more profitably directed to disordered carbons, where up to two lithium layers per carbon layer are envisioned, corresponding to a consistent enhancement of specific capacity with respect to that of graphite.[9] Following this line, we have recently described the synthesis and characterization of a class of disordered carbons prepared by a new concept involving the pyrolysis of oligophenylene precursors such as hexa(phenyl)benzene (HPB) and hexakis(*p*-bromophenyl)-benzene (HPB-Br).[10] Preliminary tests have shown that these carbons are electrochemically active and that they can be cycled in a lithium cell with a stable capacity of the order of 500 mAh g^{-1}.[10]

In this communication we report a further electrochemical characterization of one of these new disordered carbon electrodes, i.e., the HPB-derived material, and its use as a novel, advanced anode in a lithium-ion battery.

Figure 1 shows the X-ray diffraction (XRD) pattern of the as-prepared, pyrolyzed HPB carbon sample. The 002 major peak at $2\theta \sim 27°$, which is typical of graphite, is clearly shown, confirming the graphene-like structure of the sample. The broad peak at higher angle ($2\theta \sim 43°$) is due to convolution of the [10] *hk* bi-dimensional line and the weak 004 reflection. As discussed by Inagaki,[11] the appearance of *hk* lines is due

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