Burkholderia cenocepacia-induced delay of acidification and phagolysosomal fusion in cystic fibrosis transmembrane conductance regulator (CFTR)-defective macrophages

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Received 11 August 2008
Revised 12 September 2008
Accepted 17 September 2008

INTRODUCTION

Cystic fibrosis (CF) is a lethal recessive human genetic disorder caused by mutations in the gene encoding a cAMP-regulated chloride channel (cystic fibrosis transmembrane conductance regulator; CFTR) that is predominantly expressed on the apical membrane of epithelial cells (Boucher, 2007; Cheng et al., 1990; Riordan et al., 1989). The airways of CF patients become chronically infected with a restricted subset of opportunistic soil bacteria belonging to the Pseudomonas and Burkholderia genera (Govan et al., 1996; Govan & Deretic, 1996). Chronic infection leads to progressive airway tissue destruction, pulmonary dysfunction, and death. Burkholderia cepacia complex (Bcc) strains have become important human opportunistic pathogens (Coenye & Vandamme, 2003; Mahenthiralingam et al., 2005), particularly in young adult patients with CF. B. cenocepacia is one of the most prevalent and pervasive Bcc species in Canada and other parts of the world (Speert et al., 2002). Bcc and non-Bcc species of Burkholderia are characterized by their metabolic diversity and ability to adapt to varying environmental conditions, including nutrient limitation, antibiotics, antimicrobial peptides and toxic compounds (Coenye & Vandamme, 2003).

Survival and persistence within host cells and tissues are believed to play a key role in the adaptation of Burkholderia species to multiple niches, including roots and other plant tissues, free-living cells in the environment, and humans.

Abbreviations: Bcc, Burkholderia cepacia complex; BcCV, Burkholderia cenocepacia-containing vacuole; CF, cystic fibrosis; CFTR, cystic fibrosis transmembrane conductance regulator; eGFP, enhanced green-fluorescent protein; mRFP1, monomeric red-fluorescent protein 1; TMR–dextran, tetramethylrhodamine–dextran.
Indeed, *Burkholderia* species can be found as obligate intracellular endosymbionts in pathogenic fungi (Partida-Martinez & Hertweck, 2005), as symbiotic and endophytic bacteria inhabiting leaf galls, roots and other tissues of plants (Balandreau & Mavingui, 2006; Van Oevelen et al., 2002), and within free-living amoebae (Inglis et al., 2000; Lamothe et al., 2004; Landers et al., 2000; Marolda et al., 1999). Direct evidence of ex vivo intracellular survival of Bcc isolates in mammalian cells has been demonstrated in the human respiratory epithelial cell line A549 and in cultures of primary lung epithelial cells (Burns et al., 1996; Keig et al., 2001, 2002; Sajjan et al., 2006). Although it is unclear whether intracellular survival in epithelial cells is relevant in vivo, the capacity of Bcc strains to penetrate epithelial cells correlates with mouse infectivity (Chiu et al., 2001; Cieri et al., 2002). Furthermore, *Burkholderia pseudomallei* (Jones et al., 1996) and *Burkholderia mallei* (Ribot & Ulrich, 2006) survive and replicate in macrophage cell lines, unlike Bcc strains, which survive with minimal or no replication (Lamothe et al., 2007; Martin & Mohr, 2000; Saini et al., 1999). These differences may be due to the ability of *B. mallei* and *B. pseudomallei* to escape from the phagocytic vacuole shortly after phagocytosis and replicate in the cytosol, a process that requires bacterial effectors secreted by type III secretion systems (Stevens et al., 2002, 2003). In contrast, phagocytized Bcc strains reside in bacteria-containing vacuoles (BcCVs) that at least for the initial hours post-infection do not acidify and avoid fusion with lysosomes by a type III-independent mechanism (Lamothe et al., 2007). The bacterial determinants and the host cell targets involved in the mechanism underlying Bcc-mediated altered phagosome maturation still remain unknown.

The ability of Bcc strains to colonize and infect the mucosal airways of CF patients is not well understood. Recent work has suggested that phagosomes of CFTR-defective alveolar macrophages and neutrophils exhibit a constitutive acidification delay that could contribute to alveolar macrophage dysfunction, presumably facilitating infection by CF-related pathogens such as *Pseudomonas aeruginosa* (Di et al., 2006). However, an independent study using fluorescence ratio imaging to measure the endosomal pH against an internal standard unequivocally demonstrated that phagolysosomal acidification in alveolar macrophages is CFTR-independent (Haggie & Verkman, 2007), although these investigators did not examine directly the ability of CFTR macrophages to clear an intracellular infection. In agreement with the latter study, we demonstrate here that uninfected CFTR-defective macrophages or normal macrophages treated with a CFTR-specific inhibitor display normal acidification. However, following infection with *B. cenocepacia*, BcCVs from CFTR-defective macrophages exhibit a more pronounced delay in acidification and phagolysosomal fusion than that observed with normal macrophages. The CFTR-associated phagosomal maturation defect was absent in macrophages exposed to heat-inactivated *B. cenocepacia* and macrophages infected with a non-CF pathogen such as *Salmonella enterica*, a bacterium that once it becomes intracellular rapidly traffics to acidic compartments that acquire lysosomal markers. We conclude that CFTR somehow contributes to the clearance of the intracellular infection and further maturation of the BcCVs, but that this process is specific for *B. cenocepacia* and perhaps other pathogens that chronically infect the airways of CF patients.

**METHODS**

**Reagents and antibodies.** Dulbecco’s modified Eagle medium (DMEM) and fetal bovine serum (FBS) were purchased form Wisent. Dextran tetramethylrhodamine, dextran Oregon green 514 and dextran FITC (molecular mass 10 000 Da) were from Molecular Probes. The primary monoclonal rat anti-mouse LAMP-1 (ID4B) antibody was from the Developmental Studies Hybridoma Bank. Polyclonal goat anti-mouse EEA1 (IgG) was from Santa Cruz Biotechnology. The secondary Alexa 488-conjugated chicken anti-rat and chicken anti-goat antibodies were purchased from Molecular Probes. Concanaamycin A and CFTR	extsubscript{mut}-172 were from Sigma.

**Bacterial strains, macrophages and culture conditions.** *B. cenocepacia* J2315 is a prototypic strain of the highly transmissible ET12 clone (Govan et al., 1993; Johnson et al., 1994). *S. enterica serovar Typhimurium* LT2 was from our culture collection. Bacteria were grown at 37 °C in Luria–Berti (LB) medium with agitation. *B. cenocepacia* carrying the arabinose-inducible plasmid pMLBAD-eGFP (Lefebre & Valvano, 2002), which expresses the enhanced green-fluorescent protein (EGFP), was grown with 2 % (w/v) arabinose and a final concentration of 100 μg trimethoprim ml	extsuperscript{−1}. *B. cenocepacia* carrying pJR1 (Lamothe et al., 2007), which expresses the monomeric red-fluorescent protein 1 (mRFP1), was grown in the presence of 100 μg trimethoprim ml	extsuperscript{−1} and *S. enterica* carrying pRSET-mRFP1 (Campbell et al., 2002) was grown in the presence of 100 μg ampicillin ml	extsuperscript{−1}. RAW 264.7 macrophages were derived from BALB/c mice (TIB-71, ATCC). The murine cftr	extsuperscript{−/−} and cftr	extsuperscript{+/+} macrophage cell lines were obtained from Dr D. Radzioch, McGill University. Bone marrow-derived macrophages from the CF-neo ablution were generated by stable transfection of the retroviral recombinant J2 construct expressing part of the v-raf and v-myc genes, as described previously (Blasi et al., 1989). The cell lines were derived from the bone marrow of littermate control mice (born from the same carrier of the CF-neo ablution vector parents). The cftr gene knockout mice and their littermate cftr parental controls were on the same C57BL/6 genetic background. The ablution of the cftr gene in the cftr	extsuperscript{−/−} macrophages was confirmed in our laboratory by PCR amplification of an internal fragment of the murine cftr gene exon 10 using primers CFE10NT (5′-GGAAATTTATTCCAGGAGTGAGAGCT-3′) and CFE10CT (5′-GCTTACTGAGAAGGTGAAGCT-3′). Cells were maintained in DMEM with 10 % FBS and grown at 37 °C in a humidified atmosphere under 5 % carbon dioxide.

**Macrophage infection assays.** Bacterial strains were washed twice and resuspended in DMEM 10 % FBS. Heat-inactivated bacteria were obtained by incubation at 60 °C for 25 min prior to infection. Under these conditions we have noticed 100 % bacterial cell death upon plating in Luria broth medium. Bacteria were added to macrophage cells grown on glass coverslips at the m.o.i. indicated in each experiment, centrifuged for 1 min at 300 g and incubated at 37 °C with 5 % carbon dioxide. Infected macrophages were washed three times with 1 × PBS (Wiset) and observed under a fluorescence microscope or reincubated at 37 °C under 5 % carbon dioxide in DMEM containing 10 % FBS. In a previous study (Lamothe et al., 2007), we have investigated the internalization of *B. cenocepacia* strain J2315 in the mouse macrophage-like cell line RAW 264.7, and determined that an m.o.i. of 30–50 allows us to find a reasonable proportion of infected macrophages with a sufficient number of...
in intracellular bacteria for analysis. Therefore, unless otherwise indicated, all the experiments in this study were performed using an m.o.i. of 30.

**Live fluorescence microscopy.** For lysosome labelling, macrophages were incubated at 37 °C with 5% carbon dioxide in the presence of 250 μg ml⁻¹ tetramethylrhodamine–dextran (TMR–dextran) for 2 h and chased for 1 h in DMEM with 10% FBS. Labelling of the endosomal pathway was done by incubating macrophages with *B. cenocepacia* J2315 for 2 h, followed by a 1 h chase to allow maturation of the BcCVs, and a final incubation with 250 μg TMR–dextran ml⁻¹ for an additional 2 h. External TMR–dextran was removed by serial washes with PBS and the macrophages were immediately visualized. Images were acquired using an Axioscope 2 (Carl Zeiss) microscope with a ×100 oil immersion objective coupled to a Qimaging (Burnaby) cooled charged-coupled device camera.

**Immunofluorescence and confocal microscopy.** Cells were fixed in 4% paraformaldehyde (v/v) for 30 min at room temperature and incubated for 10 min with 100 mM glycine in 1× PBS. Cells were then permeabilized with 0.1% Triton X-100 (v/v) and blocked with 5% milk powder (v/v) for 1 h at room temperature. Permeabilized cells were incubated with primary antibodies, followed by secondary antibodies in 5% milk powder for 1 h each at room temperature. Coverslips were mounted onto glass slides using fluorescent mounting medium (DakoCytonation). All secondary antibodies were used at a dilution of 1:1000, rat anti-LAMP-1 at 1:50 and goat anti-EEA1 at 1:100. Confocal microscopy was performed using a Zeiss LSM 510 laser scanning confocal microscope and a ×100 oil immersion objective. In each experiment, the colocalization of EEA1 and BcCVs was done by incubating macrophages with primary antibodies, followed by secondary antibodies and a ×100 oil immersion objective. In each experiment, the colocalization of EEA1 and BcCVs was determined by examining 21 fields at ×100 and counting the number of BcCVs surrounded by a ring-like structure per macrophage cell.

**Analysis of endosomal pH.** Macrophages were grown on coverslips in DMEM with 10% FBS to a confluence of 60%. Monolayers were then incubated simultaneously with 250 μg TMR–dextran ml⁻¹ and 250 μg ml⁻¹ FITC-conjugated dextran for 2 h or overnight at 37 °C. External dextran was removed by three washes of 2 ml PBS and macrophages were reincubated for 2 h in DMEM 10% FBS to chase dyes to lysosome compartments. In control experiments, macrophages were preincubated for 20 min with 250 nM concanamycin (to inhibit the activity of the vacuolar ATPase; Whytisde et al., 2005) or 10 μM CFTRinh-172, a thiazolidinedione compound that specifically inhibits CFTR ion channel activity and has no other known toxic effects for rodent cells in culture (Ma et al., 2002; Taddei et al., 2004). To activate macrophages, monolayers were incubated with 500 ng ml⁻¹ of *B. cenocepacia* J2315 LPS. Pictures were taken using a Zeiss LSM 510 laser scanning confocal microscope with a ×100 oil immersion objective, and laser power remained constant between each experiment. Fluorescence intensities were analysed using LSM Image Examiner software and presented as fluorescence ratios (FITC/TMR).

**Statistical analyses.** The statistical analysis was done using GraphPad Prism version 4-03 (GraphPad Software) by two-way ANOVA and Student’s t test, depending on the experiment. Data were collected by counting the BcCVs per macrophage cell over 21 fields in triplicate.

### RESULTS AND DISCUSSION

**CFTR dysfunction does not alter the early maturation of BcCVs**

To characterize the nature of the compartment occupied by intracellular *B. cenocepacia* in macrophages with altered CFTR function we used three murine macrophage cell lines: RAW 264.7 macrophages, derived from BALB/c mice, and a pair of congenic *cfr*⁺/+ and *cfr*−/− macrophage cell lines derived from C57BL/6 mice. CFTR function in RAW 264.7 macrophages was pharmacologically abrogated with treatment with CFTRinh-172. We performed all of these experiments with *B. cenocepacia* strain J2315, the index case clinical isolate of the ET12 epidemic clone that has been associated with severe lung deterioration in CF patients (Govan et al., 1993). Previously, our laboratory demonstrated that strain J2315 is poorly internalized by RAW 264.7 macrophages. However, m.o.i. values of 30–50 result in a reasonable proportion of infected macrophages and are sufficient for microscopic analysis (Lamothe et al., 2007).

Also, in previous work, we have shown that the phagosomes of RAW 264.7 macrophages containing live *B. cenocepacia* J2315 remain functionally connected to the endocytic pathway, as revealed by the continued access of endocytic tracers, such as fluorescent dextran, to the BcCVs (Lamothe et al., 2007). It should be noted that lack of intracellular bacterial replication and the failure to kill extracellular bacteria prevent us from obtaining viable counts in macrophage infection experiments (Lamothe et al., 2007; Saini et al., 1999). Therefore, to examine the effect of the CFTR status on the intracellular behaviour of *B. cenocepacia*, we conducted single-cell analyses using a variety of markers that correspond to various stages of phagosomal maturation. We first investigated whether BcCVs in macrophages devoid of CFTR function also remain connected with the endocytic pathway. Macrophages were incubated with live *B. cenocepacia* for 2 h, allowed to mature for 1 h and then incubated with TMR–dextran (see Methods). BcCVs in both *cfr*⁺/+ and *cfr*−/− macrophages, as well as BcCVs from infected RAW 264.7 macrophages that were pre-incubated with CFTRinh−172, incorporated fluorescent dextran (Fig. 1a). Quantitative analysis demonstrated that on average 75% of BcCVs accumulated external dextran with no statistically significant differences among the cell lines, irrespective of CFTR function (Fig. 1b). Therefore, the CFTR functional status does not affect the interactions of BcCVs with newly formed endocytic vacuoles. We also demonstrated previously that the EEA1 protein, a marker for the sorting vesicles of the endocytic pathway, is transiently recruited previously that the EEA1 protein, a marker for the sorting vesicles of the endocytic pathway, is transiently recruited
Fig. 1. BcCVs remain functionally associated with incoming endosomes. (a) eGFP fluorescent B. cenocepacia J2315, TMR–dextran and merged pictures of BcCVs from cftr+/+ macrophages (top panels), cftr−/− macrophages (middle panels) and RAW 264.7 macrophages exposed to 10 μM of CFTRinh-172 (bottom panels). All experiments were conducted at an m.o.i. of 30, which we have previously determined to be optimal conditions for phagocytosis of B. cenocepacia (Lamothe et al., 2007). Bar, 11 μm. (b) Percentage of BcCVs accumulating external dextran in cftr+/+ (white bar), cftr−/− (black bar) and CFTRinh-172-treated (hatched bar) RAW 264.7 macrophages. The values represent the mean and SD of three experiments in which 21 fields were examined at ×100. Error bars, SEM.

Fig. 2. BcCVs transiently accumulate the early endosomal marker EEA1. cftr+/+ (top panels) and cftr−/− (bottom panels) macrophages were infected with live B. cenocepacia J2315 expressing mRFP1 (red fluorescence) at an m.o.i. of 50. Immunolabelling was performed using goat anti-EEA1 and a secondary anti-goat antibody conjugated to AlexaFluor 488 (green fluorescence). Images were captured with a 510 LSM confocal microscope and ×100 objective. Bar, 6 μm.
lysosome marker LAMP-1 is clearly detected in the membrane surrounding BcCVs, forming a typical ring-like fluorescent structure (Lamothe et al., 2007). In contrast, at earlier time points, there is a redistribution of the LAMP-1-rich compartment, as demonstrated by images showing clumps of LAMP-1 fluorescence surrounding intracellular bacteria with no typical ring formation (Lamothe et al., 2007). We investigated whether CFTR function is required for this late maturation step by monitoring the temporal accumulation of LAMP-1 into BcCVs of CFTR-defective macrophages. As expected according to our previous study (Lamothe et al., 2007), BcCVs of CFTRinh-172-treated and -untreated RAW 264.7 cells, as well as BcCVs of cftr<sup>+/+</sup> and cftr<sup>−/−</sup> macrophages, did not accumulate LAMP-1 between 1 and 4 h post-infection. However, at 8 h post-infection, 80 ± 2% of BcCVs from cftr<sup>+/+</sup> macrophages acquired LAMP-1, giving rise to typical ring-like structures (Fig. 3a, top-middle panel, arrowheads, and Fig. 3b). In contrast, rings of LAMP-1-positive material were observed in fewer than 32 ± 3% of BcCVs in cftr<sup>−/−</sup> cells (P<0.001; Fig. 3a, centre panel, and Fig. 3b). At 8 h post-infection, several BcCVs in cftr<sup>+/−</sup> macrophages also demonstrated a uniform red fluorescent background (Fig. 3a, top-left panel, arrows). We have previously shown that the escape of mRFP1 from the bacterial cytoplasm to the lumen of the BcCVs occurs after phagolysosomal fusion (Lamothe et al., 2007), indicating loss of bacterial cell envelope integrity. This escape of mRFP1 was not observed in BcCVs of infected cftr<sup>−/−</sup> macrophages (Fig. 3a, middle-left panel), suggesting that bacteria in these vesicles still have an intact cell envelope. RAW 264.7 macrophages incubated with CFTRinh-172 also did not reveal a significant number of typical rings of LAMP-1 colocalization with BcCVs at 8 h post-infection (Fig. 3b). However, clumps of LAMP-1-positive material were found colocalizing with BcCVs (Fig. 3a, lower panels). We have previously observed this

![Fig. 3. Colocalization of LAMP-1 to BcCVs.](http://mic.sgmjournals.org)
phenomenon in untreated RAW 264.7 macrophages infected with live *B. cenocepacia* just prior to ring formation (Lamothe *et al.*, 2007), but at 1–3 h post-infection. We also observed in some of the BcCVs the release of mRFP1 to the vacuolar lumen (Fig. 3a, lower-left panel, arrows), suggesting that phagolysosomal fusion had begun to take place. Therefore, these results suggest that in the presence of CFTRinh-172 there is also a significant delay in the maturation of the BcCVs, albeit less pronounced than that found with *cftr*<sup>−/−</sup> macrophages.

The pronounced maturation delay observed with *cftr*<sup>−/−</sup> and CFTRinh-172-treated RAW 264.7 macrophages, as judged by the ability of BcCVs to accumulate LAMP-1, was not noticeable when these cells were exposed to heat-inactivated *B. cenocepacia*. Indeed, BcCVs carrying heat-inactivated *B. cenocepacia* rapidly fused with LAMP-1-rich compartments at 30 min post-infection in the three cell types tested (Fig. 4, see arrowheads). Quantitative analysis revealed that on average 80% of BcCVs colocalized with LAMP-1-positive material forming ring-like structures in all of the cell types. Therefore, BcCVs containing nonviable *B. cenocepacia* in *cftr*<sup>−/−</sup> macrophages or in CFTR-inhibited cells trafficked normally into LAMP-1-rich vacuoles, as we have previously found in normal RAW 264.7 macrophages (Lamothe *et al.*, 2007). These results argue that the absence of CFTR expression in *cftr*<sup>−/−</sup> cells or the pharmacological abrogation of CFTR function by CFTRinh-172 exaggerates the *B. cenocepacia*-mediated traffic delay of the BcCVs into the lysosomes.

**A dysfunctional CFTR has no impact on the maturation of vacuoles containing *S. enterica***

To assess whether the absence of a functional CFTR protein has any effect on the trafficking of other intracellular bacteria, we infected *cftr*<sup>−/−</sup> macrophages with *S. enterica* strain LT2. *Salmonella* is a bona fide intracellular pathogen that promotes its own uptake, and since it has been shown that a high bacterial inoculum may lead to macrophage cell death induced by intracellular *Salmonella* (Fink & Cookson, 2007; Monack *et al.*, 1996; Rupper & Cardelli, 2008), we used a lower m.o.i. for these experiments. It has been reported that the bacteria-containing vacuoles of RAW 264.7 cells infected with *S. enterica* acquire LAMP-1 relatively early after infection (Beuzón *et al.*, 2000; Rathman *et al.*, 1997). At 4 h post-infection, phagosomes containing live *S. enterica* from *cftr*<sup>−/−</sup> and RAW 264.7 cells incubated with CFTRinh-172 accumulated LAMP-1 (Fig. 5). These results indicate that the delay in LAMP-1 accumulation is specific to the vacuoles containing live *B. cenocepacia* and is not a general defect with any viable intracellular bacteria.

**Uninfected CFTR-defective macrophages display normal phagolysosomal acidification**

Internalized *B. cenocepacia* within macrophages reside in poorly acidified vacuoles before fusion with lysosomes (Lamothe *et al.*, 2007). To assess the state of acidification of vacuoles carrying live *B. cenocepacia* in *cftr*<sup>−/−</sup> macrophages, infected cells were incubated with LysoTracker Red (Invitrogen). This fluorescent probe accumulates preferentially within the most acidic compartments of the cell due to

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**Fig. 4.** Vacuoles containing heat-inactivated *B. cenocepacia* rapidly accumulate LAMP-1 in the absence of CFTR function. *cftr*<sup>+/+</sup>, *cftr*<sup>−/−</sup> and CFTRinh-172-treated RAW 264.7 macrophages were infected with heat-inactivated *B. cenocepacia* expressing mRFP1 (red fluorescence) at an m.o.i. of 30 and fixed at 30 min post-infection. Immunolabelling was performed using rat anti-LAMP-1 and a secondary anti-rat antibody conjugated to AlexaFluor 488 (green fluorescence). Arrowheads point to typical ring-like structures denoting the colocalization of LAMP-1 with the BcCVs, which is observed in the three types of macrophages. Images were captured with a 510 LSM confocal microscope and ×100 objective. Bar, 12 μm.
the formation of a membrane-impermeable form upon protonation in low pH environments (Lemieux et al., 2004). The accumulation of LysoTracker into BcCVs of cftr \(^{-/-}\) and cftr \(^{+/+}\) macrophages infected with live B. cenocepacia was quantitatively assessed 8 h after internalization by counting BcCVs in 21 fields in triplicate experiments. At 8 h post-infection, 23\(\pm\)4\% of BcCVs within cftr \(^{-/-}\) macrophages colocalized with the acidic probe compared with 83\(\pm\)3\% of BcCVs within cftr \(^{+/+}\) macrophages (\(P<0.001\); data not shown). These results demonstrate that the vacuoles containing B. cenocepacia in cftr \(^{-/-}\) macrophages remain poorly acidified at a time that corresponds to the extended delay in BcCV maturation. Consistent with poor acidification and the absence of LAMP-1 localization to the BcCVs, vacuoles carrying live B. cenocepacia in cftr \(^{-/-}\) macrophages did not colocalize with dextran-labelled lysosomes (data not shown).

It has recently been suggested that CFTR plays a role in the pH regulation of alveolar macrophage lysosomes (Di et al., 2006). However, others have observed no pH variation in organelles of cells that lack CFTR function (Gibson et al., 2000; Haggie & Verkman, 2007). Since acidification of endosomes is a critical factor for phagosomal maturation (Clague et al., 1994; van Weert et al., 1995) and LAMPs are required for lysosomal fusion (Huynh et al., 2007), we investigated the relationship between CFTR and lysosome acidification in our cell lines. Ratiometric fluorescence analyses between pH-insensitive (TMR–dextran) and pH-sensitive (FITC–dextran) dyes were performed to evaluate acidification of lysosome-like compartments in both resting and activated macrophages. Macrophages from cftr \(^{-/-}\) and cftr \(^{+/+}\) genetic backgrounds were incubated with pH-sensitive and pH-insensitive fluorescently labelled dextrans (see Methods) and fluorescence ratios (FITC/TMR) were compared over time. As controls, fluorescence ratios were determined after treatment with CFTRinh-172 and concanamycin A, a V-H\(^+\) ATPase inhibitor (Whyteside et al., 2005). FITC fluorescence in the endosomal compartments from both cftr \(^{-/-}\) and cftr \(^{+/+}\) macrophages was quenched, indicating lumenal acidification, but the endosomes remained strongly fluorescent in cells pre-treated with concanamycin A (Fig. 6a). The fluorescence ratios (FITC/TMR) of concanamycin A-treated cells were around 1, while the fluorescence ratios of cftr \(^{-/-}\), cftr \(^{+/+}\) and RAW 264.7 macrophages treated with the CFTR inhibitor averaged between 0.6 and 0.7 (Fig. 6b), demonstrating no statistically significant differences between the three experiments. It could be argued that one major confounding factor in our experiments is the presence of extracellular bacteria during the entire course of the experiment. However, similar results were also obtained from radiometric analyses using macrophages that were activated by treatment with 500 ng B. cenocepacia J2315 LPS (data not shown), a condition that leads to macrophage cell activation by stimulating the synthesis and secretion of tumour necrosis factor-1 and the oxidative burst (Saini et al., 1999; K. Keith and M. A. Valvano, unpublished results). Together, these data demonstrate that in agreement with an earlier report (Haggie & Verkman, 2007), CFTR \(^{-/-}\) macrophages or macrophages treated with CFTRinh-172 do not have an intrinsic defect in their ability to acidify lysosomal compartments. It has been shown elsewhere that the CFTR protein in macrophages and neutrophils can be visualized in the membrane of vacuoles (Di et al., 2006; Painter et al., 2006). Experiments in our laboratory using five different CFTR-specific antibodies, including the one used in an earlier report (Di et al., 2006), did not yield any results. However, CFTR expression was detected by RT-PCR (data not shown). These results support the notion that CFTR is present in these cells at very low levels of expression.

**Concluding remarks**

Macrophages are increasingly recognized as having a central role in the maintenance of immunological homeostasis and host defence in the respiratory tract (Holt et al.,
Therefore, although the virulence factors responsible for the persistence of *B. cenocepacia* and the bacterial properties associated with tropism for the airways of CF and other immunocompromised patients are still not fully understood, the ability of these bacteria to survive intracellularly may play a role in pathogenesis (Chiu et al., 2001; Valvano et al., 2005, 2006). The purpose of the present study was to determine whether macrophages carrying a mutated *cftr* gene or a functionally defective CFTR protein have any alterations in their ability to process *B. cenocepacia*. Our results demonstrate that in CFTR-defective macrophages, the BcCVs exhibit a pronounced delay in phagolysosomal fusion, which is associated with a reduced acidification of the lumen. We also show that this process is specific for live *B. cenocepacia*, since heat-inactivated bacteria are trafficked normally to the lysosomal compartment upon phagocytosis. Furthermore, the non-CF pathogen *S. enterica* does not affect the acquisition of the phagolysosomal marker LAMP-1 in CFTR-defective cells. Di et al. (2006) have recently demonstrated that lysosomal compartments within *cftr*<sup>-/-</sup> alveolar macrophages fail to acidify, leading...
to a bactERICidal defect, suggesting that CFTR is involved in the pH regulation of the phagocytic vacuole. Our results from ratiometric fluorescent intravacuolar pH measurements do not support the notion that CFTR plays a direct role in the acidification of the phagocytic vacuole, as proposed by Di et al. (2006), as we did not find a constitutive acidification defect in our cftr−/− macrophages. This is in agreement with a recent detailed analysis using fluorescence ratio imaging to measure the endosomal pH against an internal standard, which convincingly demonstrated that phagolysosomal acidification in macrophages is CFTR-independent (Haggie & Verkman, 2007). It has been reported that in human neutrophils CFTR channel dysfunction affects neutrophil chlorination of phagocytosed bacteria (Painter et al., 2006, 2008), raising the possibility that CFTR contributes to bacterial clearance rather than phagolysosomal acidification. Our experiments with B. cenocepacia-infected CFTR-defective macrophages showing an extended delay in the trafficking of BcCVs to lysosomes do indeed support a role for CFTR in the mechanism of clearance of the intracellular infection, as we have shown before that B. cenocepacia localized to the lysosome rapidly loses cell envelope integrity (Lamotte et al., 2007). Furthermore, the prolonged maturation arrest of the vacuoles containing B. cenocepacia within cftr−/− macrophages could explain the persistence of the bacteria within CF patients compared to healthy individuals. Further experiments are currently under way in our laboratory to elucidate the interplay of CFTR and B. cenocepacia intracellular survival in macrophages.

ACKNOWLEDGEMENTS

The authors thank C. Marino, University of Texas Southwestern Medical Center, and D. Radzioch, McGill University, for providing CFTR-specific antibodies and the cftr−/− and cftr+/+ macrophage cell lines, and D. Laird, S. Koval and S. Kim for useful comments. This work was supported by operating grants from the Canadian Cystic Fibrosis Foundation. J. L. was supported by studentships from the Canadian Cystic Fibrosis Foundation and a Canada Graduate Scholarship from the Canadian Institutes of Health Research. M. A. V. holds a Canada Research Chair in Infectious Diseases and Microbial Pathogenesis.

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Edited by: P. Cornelis