Negative Regulation of Phagocytosis in Murine Macrophages by the Src Kinase Family Member, Fgr

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Abstract

Ingestion of opsonized pathogens by professional phagocytes results in the generation and release of microbicidal products that are essential for normal host defense. Because these products can result in significant tissue injury, phagocytosis must be regulated to limit damage to the host while allowing for optimal clearance and destruction of opsonized pathogens. To pursue negative regulation of phagocytosis, we assessed the effect of the Src kinase family member, Fgr, on opsonin-dependent phagocytosis by mouse macrophages. We chose Fgr because it is present in high concentrations in circulating phagocytes but is not essential for Fcγ receptor–mediated ingestion by mouse macrophages. Although expression of Fgr both in a macrophage cell line and in primary macrophages significantly attenuates ingestion mediated by Fcγ receptors and CR3, it does not affect macropinocytosis or receptor-mediated endocytosis. This selective effect of Fgr is independent of its tyrosine kinase function. After Fcγ receptor cross-linking, Fgr becomes associated with the immunoreceptor tyrosine-based inhibition motif (ITIM)–containing receptor, SIRPα (a member of the signal-regulatory protein family, also known as Src homology 2 domain–containing protein tyrosine phosphatase [SHP] substrate 1 [SHPS-1], brain immunoglobulin-like molecule with tyrosine-based activation motifs [BIT], and P84) and potentiates the association of the phosphatase SHP-1 with SIRPα. This association is responsible, at least in part, for decreasing positive signaling essential for optimal phagocytosis. These data demonstrate an important negative regulatory role for this Src kinase family member and suggest that this homeostatic function must be overcome for optimal uptake and clearance of opsonized pathogens.

Key words: phagocytes • Src family kinases • protein tyrosine phosphatase

Introduction

Phagocytosis is a complex biological process of specialized cells that is essential for normal host defense against infectious pathogens and for resolution of tissue injury at sites of inflammation (1–3). We and others have hypothesized that phagocytosis is primarily a recruited function at sites of infection and inflammation because ingestion mediated by both Fcγ and complement receptors is augmented by inflammatory mediators and matrix proteins that abound at sites of tissue injury (2, 4–9). These data imply that phagocytosis is regulated. Regulatory control of phagocytosis would be of substantial benefit to the host because the production of tissue-damaging products generated during ingestion of opsonized pathogens, such as reactive oxygen and nitrogen intermediates and lysosomal enzymes, would be restricted to the site of inflammation. In this paradigm, phagocytes circulating in the vasculature would have limited phagocytic potential. However, after migration into sites of infection or inflammation, their phagocytic responses would be amplified, allowing for optimal ingestion and destruction of opsonized targets. This amplification could occur by acquiring the ability to recruit additional positive signaling molecules and/or by losing or overcoming negative signaling components that act to limit or attenuate phagocytic responses. Although considerable advances have been made in elucidating the intracellular signals essential for positive
signaling in phagocytosis (1–3), knowledge of the specific signal transduction components that negatively regulate phagocytosis is somewhat lacking.

Negative regulation of signal transduction cascades is necessary for homeostasis in a variety of systems. In hematopoietic and immune system cells, failure to regulate signal transduction during cell activation can result in hyperresponsive states that lead to significant pathological sequelae, such as autoimmunity and excessive inflammation (10–17). Examples include the autoimmune-mediated tissue injury and uncontrolled leukocyte activation that occur in mice with disrupted genes encoding the antiinflammatory cytokines, IL-10 (13) and TGF-β1 (14–16), and the enhanced antibody and anaphylactic responses observed in mice that lack signal regulatory protein of the immune receptor tyrosine-based inhibition motif (ITIM)-containing receptor, signal regulatory protein of the α subtype (SIRPα), after phagocytic receptor activation.

**Materials and Methods**

Reagents. The following reagents were obtained as indicated: S-S.1 hybridoma, which secretes murine mAb IgG2a anti-CD14 (PharMingen), purified rabbit polyclonal anti-CD14 (Diamedix), purified biotin-labeled goat anti-rat Fab′2 (Jackson ImmunoResearch Labs), purified mAb 2.4G2 rat anti-murine FcγRII/FcγR III (PharMingen), purified mAb M1/70 rat anti-murine CD11b (PharMingen), purified mAb rat IgG1 anti-murine CD14 (PharMingen), FITC-avidin (PharMingen), purified mAb 26E hamster anti-murine β1 (Endogen), FITC-labeled 70-kD dextran (Molecular Probes), rhodamine-phalloidin (Molecular Probes), and purified murine fibronectin (Calbiochem). Rat IgG1 anti-murine SIRPα (PB4 antigen [23]) was purified from tissue culture supernatant by ammonium sulfate precipitation and protein G chromatography using the mAb Trap II kit (Amersham Pharmacia Biotech).

Retroviral Infections. BAC1.2F5, a subclone of a murine macrophage cell line (24) that does not express Fgr, was used for the introduction of wild-type and site-directed mutants of murine c-fgr by retroviral-mediated gene transfer. Murine c-fgr cDNA was isolated as described (25) and subsequently modified by deletion of the 5′ untranslated region to position 143 to increase translational efficiency. To generate a kinase-inactive mutant, a 21-mer oligonucleotide primer, 5′-CTT CAG CGT CCT CAC TGC CAA-3′, was used to convert Lys 279 (AAG) to Arg 279 (AGG). All mutations in c-fgr cDNA were confirmed by DNA sequencing (Sequenase; U.S. Biochemical). c-fgr constructs were cloned into the retroviral vector pLNCX (Clontech), and pCLNX-f cDNAs or a vector control were transfected into the murine packaging cell line, P8-1 (American Type Culture Collection), as described (26). After selection in 250 μg/ml G418, subclones were screened for viral titer, clonal integration, and Fgr expression by immunoblotting. 1 ml of helper-free virus at 105–106 PFU/ml was used to infect 105 BAC1.2F5 cells in the presence of 4 μg/ml polybrene (Sigma Chemical Co.). After infection, clones were selected in 250 μg/ml of G418. The following clones were used in this investigation: vector control, BAC1.2F5 cells expressing a vector control; Fgr(WT), BAC1.2F5 cells expressing various levels of wild-type c-fgr of the p59 isoform; and Fgr(K279R), BAC1.2F5 cells expressing c-fgr of the p59 isoform with a K279 to R 279 mutation that inactivates kinase function. Expression of Fgr protein was confirmed by Western blot analysis (see below for details). Lack of Fgr kinase activity in Fgr(K279R)-expressing cells was confirmed by an in vitro kinase assay based on auto-phosphorylation of Fgr immunoprecipitates. In brief, Fgr protein was immunoprecipitated with a polyclonal anti-Fgr peptide antibody (described below). The washed immunoprecipitates were incubated with [γ-32P]ATP for 30 min at 30°C, the kinase reaction was terminated, and the eluted proteins were run on SDS-PAGE. Fgr(WT) cells expressed 32P-labeled Fgr, whereas the Fgr(K279R) cells did not (data not shown).

To confirm that cell surface expression of phagocytic receptors was equivalent for all BAC1.2F5 clones, the cells were checked by fluorescent flow cytometry with a FACScan® cytometer (Becton Dickinson). FcγRI was assessed by direct binding of FITC-
labeled purified monomeric murine IgG2a mAb S-S.1. Binding of FITC-IgG2a was not blocked by mAb 2.4G2. FcγRII/FcγRIII and CD11b were isolated by indirect immunofluorescence using biotinylated rat mAb 2.4G2 and M1/70, respectively, and FITC-avidin. BAC1.2F5 clones (10\(^6\)) were incubated with 1 \(\mu\)g of antibody for 90 min at 0°C, washed, incubated with 0.1 \(\mu\)g of FITC-avidin where appropriate, washed, and fixed in 1% paraformaldehyde in PBS. Levels of expression for these phagocytic receptors were equivalent for all BAC1 clones. These cells were maintained in DMEM containing 15% FCS, 15–25% conditioned media from L929 cells as a source of C5-deficient mouse serum, 100 \(\mu\)g/ml penicillin, 100 \(\mu\)g/ml streptomycin, 4,500 mg/l glucose, and 200 \(\mu\)g/ml G418. Tissue culture reagents were purchased from Gibco BRL. All cell lines were routinely checked for Mycoplasma contamination by PCR (Boehringer Mannheim).

Isolation of Primary Phagocytes. Bone marrow macrophages, resident peritoneal macrophages, and bone marrow neutrophils (PMNs) were isolated from wild-type C57BL/6 mice, from Fgr\(^{-/-}\) mice in the C57BL/6 background (22), from motheaten viable (me/me) mice obtained from The Jackson Laboratory, and FcR knockout mice (γ chain knockout crossed with an FcR11β knockout) in C57BL/6 \(\times\) 129 mice obtained from Taconic Farms. Mice heterozygous for the targeted deletion of Fgr were bred, and the homozygous null offspring were detected by PCR (Boehringer Mannheim). Mice heterozygous for the targeted deletion of Fgr were bred, and the homozygous null offspring were detected by PCR (Boehringer Mannheim).

Adherent BAC1.2F5 cells and bone marrow macrophages were released from tissue culture flasks by incubation with 5 mM EDTA in PBS for 15 min at 37°C. All phagocytes were washed twice in buffer and resuspended in HBSS containing 10 mM Hepes, 0.5 mM Ca\(^{2+}\), 2.5 mM Mg\(^{2+}\), 50 \(\mu\)g/ml gentamicin, and 1% sterile, endotoxin-free human serum albumin (HBSS with 1% HSA) at 2 \(\times\) 10\(^6\)/ml. For some experiments, analyses were performed in complete tissue culture medium containing 15% LCM with qualitatively similar results. Reactions with cells in suspension were performed in 12 \(\times\) 75 mm polypropylene tubes (Falcon Labware), and reactions with adherent cells were performed on either polystyrene 24-well plates (Falcon Labware) or on 8-chamber Lab-Tek glass slides (Nunc). To facilitate equivalent cell adhesion for all of the transfected cells, adherent assays were performed on plates or slides that had been coated overnight at 4°C with 2 \(\mu\)g/ml murine fibronectin in PBS.

Opsonization. Erythrocytes (BioW hittaker) were opsonized with polyclonal rabbit anti–sheep erythrocyte (ElgGR) as described previously (5, 6). For ElgG2a and ElgG2b (27), erythrocytes were washed in veronal-buffered saline (VBS), and 200 \(\mu\)l (10\(^9\)/ml) was incubated with the indicated volume of the respective hybridoma tissue culture supernatant or purified antibody for 30 min at 37°C. For EC3bi, erythrocytes were washed and suspended in dextrose-buffered VBS containing 170 mM dextrose, 0.15 mM CaCl\(_2\), and 1.0 mM MgCl\(_2\) (at 10\(^9\)/ml) and incubated with an equal volume of a subagglutinating dilution of rabbit IgM anti-erythrocyte (6) for 30 min at 37°C. 200 \(\mu\)l of ElgM was incubated with an equivalent volume of either a 1:5 or 1:10 dilution of C5-deficient mouse serum (Jackson ImmunoResearch Labs) for 60 min at 37°C. Opsonized erythrocytes were then washed and resuspended in the respective buffer at 5 \(\times\) 10\(^9\)/ml.

Attachment and Phagocytosis Assays. To assess binding of the opsonized erythrocytes, washed macrophages (2 \(\times\) 10\(^9\)/ml) were centrifuged at 100 g with either 7.5 \(\mu\)l of ElgG2a or ElgG2b, or 25 \(\mu\)l of EC3bi in 100 \(\mu\)l of buffer for 1 min to initiate contact. The reaction mixtures were incubated at 4°C for 30 min for ElgG2a and ElgG2b, and at 37°C for 1 h for EC3bi. Attachment was assessed by light microscopy, and the attachment index (AI) was quantitated as the number of erythrocytes bound per 100 macrophages. Additionally, attachment of ElgG2a and ElgG2b at 37°C to macrophages that had been preincubated with 10 \(\mu\)M cytochalasin B (Sigma Chemical Co.) to inhibit internalization gave equivalent results. For optimal EC3bi attachment, most experiments were performed in the presence of 1.0 mM Mn\(^{2+}\). To prove that EC3bi attachment was mediated by CR3, macrophages were incubated with 1 \(\mu\)g of mAb 2E6 hamster anti–murine β\(_2\) for 30 min at 0°C before the addition of EC3bi. mAb 2E6 blocked all EC3bi attachment.

Phagocytosis was assessed with macrophages either in suspension or adherent to fibronectin-coated plates. For the suspension assay, washed macrophages (2 \(\times\) 10\(^9\)/ml) were centrifuged at 100 g with either 7.5 \(\mu\)l of ElgG2a, ElgG2b, or ElgGR, or 25 \(\mu\)l of EC3bi at 100 g for 1 min. The mixtures were gently resuspended in 100 \(\mu\)l buffer. For EC3bi, 30 ng/ml of phorbol dibutyrate (PDBu) and 1 mM MnCl\(_2\) were added to stimulate ingestion. PMN phagocytosis of EC3bi was assessed in the presence of buffer or various concentrations of FMLP as described (27). After incubation for 30 min at 37°C, unengested erythrocytes were removed by hypotonic lysis. Phagocytosis was assessed by phase microscopy and quantitated as a phagocytic index (PI), the number of opsonized erythrocytes ingested per 100 macrophages. Additionally, the percentage of phagocytosis was tabulated by counting the number of cells ingesting at least 1 opsonized erythrocyte per 100 macrophages. Phagocytosis was also assessed using adherent cells. Washed macrophages (10\(^9\)/ml) were added to fibronectin-coated 24-well plates. After incubation at 37°C for 2 h, the cells were washed and 100 \(\mu\)l of ElgGR was added. After 30 min at 4°C, the non-adherent ElgGR were washed away and ingestion was initiated by incubation at 37°C for 30 min. Unengested erythrocytes were removed by hypotonic lysis. The cells were fixed with 2.5% glutaraldehyde in PBS for 10 min at room temperature. Phagocytosis was assessed by photomicroscopy.

Macropinocytosis Assay. Macropinocytosis was defined by the uptake of FITC-labeled 70-kD dextran as described by Araki et al. (29). Vector control and Fgr(WT)-expressing BAC1.2F5 cells were cultured for 16 h in the absence of LCM to remove the influence of C5-deficient mouse serum. Washed cells were resuspended at 10\(^9\)/ml in HBSS with 1% HSA. A 100–100 \(\mu\)l aliquot of this suspension was pulsed with FITC-labeled 70-kD dextran (5 mg/ml) for 1 h at 37°C in the absence or presence of 6 ng of murine recombinant C5-deficient mouse serum (R & D Systems), washed three times, and chased for 1 h at 37°C in HBSS without HSA. In some experiments, the macrophages were preincubated with 10 nM wortmannin to inhibit phosphatidylinositol 3-kinase (PI 3-kinase) before incubation with FITC-dextran (29). The cells were washed three times at 4°C and placed on ice until fluorescence was assessed by flow cytometry. Autofluorescence of cells that were not incubated with
the FITC-dextran was subtracted to obtain specific fluorescence. The data are reported as the mean channel of fluorescence (MCF). The mean channel for autofluorescence was <2 for all experiments.

Receptor-mediated Endocytosis. R receptor-mediated endocytosis of FcγRII/FcγRIII was assessed by an adaptation of the method of Lamaze et al. (30). R receptor cross-linking and internalization were initiated by incubating 1.0 × 10^6 vector control and Fgr(WT)-expressing BAC1.2F5 cells at 0°C with 1 μg of rat mAb 2.4G2 in 50 μl of serum-free culture medium containing 20 mM Hepes and 0.2% BSA. After 1 h, the cells were washed and incubated with 1 μg biotinylated F(ab)2 goat anti-rat IgG. After washing, separate aliquots of the cells were incubated either at 0°C to measure the total amount of cross-linked receptors or at 37°C to initiate receptor internalization. After the indicated times (15 min, 30 min, or 1 h), all of the samples were washed and incubated with 0.1 μg FITC-avidin at 0°C to measure surface fluorescence. The samples were held at 0°C until examined by flow cytometry. The loss of surface staining on the 37°C-incubated samples was attributed to the internalization of the cross-linked receptors. The percentage of endocytosis was calculated by the following equation: % endocytosis = [(FL1 at 0°C autofluorescence) – (FL1 at 37°C autofluorescence)]/FL1 at 0°C autofluorescence)] × 100. The data are represented as the mean ± SEM, n = 3.

Detection of F-actin Distribution by Fluorescence Microscopy. The extent of F-actin content in phagocytic cups surrounding the opsonized E was measured by the method of Greenberg et al. (31, 32). Vector control and Fgr(WT) BAC1.2F5 cells adherent for glass coverslips were incubated with ElgGR in HBS with 1% HSA at 4°C. After 1 h, the cells were washed three times and warmed buffer was added to initiate ingestion. After 8 min at 37°C, the cells were fixed with 1% paraformaldehyde in PBS for 30 min at 37°C. After washing, the cells were permeabilized with 1% Triton X-100 in PBS for 30 min at room temperature, washed, and incubated with a 1:50 dilution of rhodamine-phalloidin for 2 h at 4°C. Slides were mounted with Fluoromount G (Southern Biotechnology) and examined by fluorescence microscopy. The extent of actin cup formation by macrophages that had bound at least 4 ElgGR was assessed by quantitating the number of actin cups formed by 100 resorted macrophages and the percentage of cells that had at least 1 actin cup. No actin cups were detected at 5 min after the initiation of phagocytosis.

Immunoprecipitations and Western Blotting. To assess expression of Fgr by Western blotting, a rabbit polyclonal antibody to a peptide corresponding to an N H2-terminal sequence Phe23-Ser43 of murine Fgr was generated. The Ser 43 residue in the synthetic peptide was replaced by Cys to facilitate coupling of the protein to KLH. R Rabbits were immunized with 200 μg peptide-coupled KLH in complete Freund's adjuvant. Affinity-purified antibodies were prepared from high titer sera on immobilized peptide columns (Sulfolink Coupling Gel; Pierce Chemical Co.). The affinity-purified antibody does not react with either murine Hck or murine Lyn. Washed and resuspended macrophages were lysed in 1% NP-40, 50 mM Tris-HCl, pH 7.5, containing 150 mM NaCl, 5 mM EDTA, 50 mM Naf, 10 mM Na pyrophosphate, 2 mM Na2VO4, 1 mM PM SF, and 2 μg/ml aprotinin (lysin buffer). Cell lysates standardized for protein content (Bio-Rad Labs) were precleared with rabbit IgG bound to protein-Sepharose 4B (Amersham Pharmacia Biotech) before SDS-PAGE on 10% acrylamide gels. After electrophoresis, proteins were transferred to Immobilon-P membranes (Millipore Corp.) and probed with the affinity-purified anti-NH2-terminal peptide at 2 μg/ml in 10% nonfat milk in PBS. R rabbit antibody was detected by 0.05 μg/ml horseradish peroxidase (HRP)-conjugated goat polyclonal anti-rabbit IgG (Vector Labs), and the membrane was developed with enhanced chemiluminescence (ECL; Pierce Chemical Co.) and exposed to BioMax MR film (Eastman Kodak Co.).

For immunoprecipitation of SIRPα after FcγRII cross-linking, vector control and Fgr(K279R) kinase-inactive BAC1.2F5 cells were cultured in the absence of LCM for 24 h. The cells were detached, washed, and resuspended in HBS with 1.0 mM CaCl2 and 1.0 mM MgCl2. To aggregate FcγRII, 6 × 10^5 cells were incubated with 25 μg/ml of rat mAb 2.4G2 at 4°C for 30 min. Equivalent data were obtained when IgG-opsonized ghosts were used to aggregate FcγRII. After washing, the cells were resuspended to 50 μl and incubated with 25 μg/ml of goat IgG anti-rat IgG (Sigma Chemical Co.) at 37°C for 0 s, 30 s, 2 min, and 5 min. The cells were pelleted and lysed with 1% Triton X-100 in lysis buffer for 15 min at 0°C. The lysates were clarified by centrifugation, normalized for protein levels, and probed by incubation with protein G-agarose for 1 h. SIRPα was immunoprecipitated by addition of purified rat IgG1 mAb P84 (23) bound to protein G overnight at 4°C. Beads were washed with lysis buffer containing 1 mM Na2VO4 and 1 mM PM SF and eluted by boiling 5 min in 2× Laemmli SDS buffer. The samples were run on SDS-PAGE, and the resolved proteins were transferred to polyvinylidene difluoride membrane. The membranes were blocked with 2% BSA, 25 mM Tris-HCl, pH 7.5, 137 mM NaCl, 0.1% Tween 20, and then probed individually with the following antibodies: 0.125 μg/ml anti-PY RRLIEDAEpYAARG (Transduction Labs), 2 μg/ml rabbit anti-Fgr NH2 peptide, 1 μg/ml rabbit IgG anti-SHP-1 (Upstate Biotechnology), or 1 μg/ml of rabbit IgG anti-SIRPα (Alexis). The R C20-HRP-probed membranes were developed directly by ECL, whereas the rabbit antibody-probed membranes were incubated with HRP-conjugated goat anti-rabbit IgG before ECL and exposure to film. Controls included cell lysates incubated with protein G-Sepharose alone and immunoprecipitation with a rat mAb against murine CD29.

Tyrosine Phosphatase Activity Associated with SIRPα. To assess functional tyrosine phosphatase activity associated with SIRPα after FcγRII activation, we incubated SIRPα immunoprecipitated from vector control and Fgr(K279R)-expressing BAC1.2F5 cells as described above with a phosphotyrosine peptide (R LLIEDAEpYAARG) overnight at 37°C. Liberated phosphate was detected by malachite green using a Tyrosine Phosphatase Assay Kit 1 (Upstate Biotechnology), following the manufacturer's instructions. Liberated phosphate was measured in picomoles from a standard curve. To assess specific phosphatase activity, the phosphate levels detected with protein G-agarose beads incubated with cell lysates in the absence of anti-P84 were subtracted from the levels detected with anti-P84. This amount was <10% of the total phosphate detected. The data are representative of two experiments, each performed in duplicate.

Results. Fgr negatively regulates phagocytosis mediated by multiple Ligand-Receptor Pairings Both in a Macrophage Cell Line and in Primary Macrophages. To test our hypothesis that Fgr mediates negative regulation of phagocytosis, we used BAC1.2F5 cells, which exhibit many of the functions of mature macrophages (24), and which we had determined in preliminary experiments did not express Fgr protein. The cells were retrovirally infected with either a vector...
control or wild-type murine c-fgr and assessed for the ability to bind and ingest ElgG2a, ElgG2b, and EC3bi. Both the vector control and Fgr(WT)-expressing cells had comparable levels of Src, Hck, Lyn, and Syk and expressed equivalent amounts of cell surface receptors, including FcγRI, FcγRII/FcγRIII, and CR3 (data not shown). Ingestion of both ElgG2a and ElgG2b was significantly suppressed in Fgr(WT)-expressing cells at all levels of opsonization (Fig. 1, A, C, and D). Moreover, ingestion was suppressed for both the total number of opsonized targets ingested (Fig. 1, A, A, and C) and the percentage of cells ingesting (Fig. 1, B and D). However, FcrR-mediated phagocytosis in the Fgr(WT)-expressing cells did increase with increasing concentrations of opsonin (Fig. 1, A and C), indicating that the cells were capable of mounting a phagocytic response, but that it was attenuated relative to the vector control cells. At levels of opsonization where 80–90% of the vector control cells ingested, only 40–45% of the Fgr(WT) cells were capable of internalizing the bound targets (Fig. 1, B and D). The same results were obtained with BAC1.2F5 cells adherent on fibronectin-coated plates and ingesting rabbit IgG-opsonized erythrocytes (data not shown). These data indicate that Fgr suppresses phagocytosis by FcγR's on murine macrophages and that the suppression cannot be overcome by increasing the concentration of ligand.

Because signal transduction pathways could vary for phagocytosis mediated by different classes of receptors (2, 3, 6, 33–35), the ability of Fgr to suppress ingestion could be limited to a single class of receptor, i.e., FcγR. Therefore, we assessed the effect of Fgr expression on CR3 (CD11b/CD18)-mediated ingestion of EC3bi. Unlike FcγRs, CR3 is not constitutively able to ingest C3bi-opsonized targets, and additional stimuli, like phorbol esters, are required for efficient internalization (2, 4, 6, 33). Even in the presence of 30 ng/ml of PDBu, Fgr(WT)-expressing macrophages ingested significantly fewer EC3bi than did vector control cells (Fig. 1 E). In addition, the percentage of cells ingesting EC3bi was suppressed in Fgr(WT) cells (Fig. 1 F). Increasing the concentration of PDBu could not overcome the suppression of ingestion (data not shown). No phagocytosis of EC3bi was observed with either cell type in the absence of PDBu (data not shown). These data indicate that Fgr is able to suppress phagocytosis mediated by multiple phagocytic receptors.

One trivial explanation for our results could be that expression of Fgr alters the optimal attachment of the opsonized target to the phagocyte. Therefore, we quantitated attachment for ElgG2a, ElgG2b, and EC3bi to vector control and Fgr(WT)-expressing cells to rule out this possibility. Equivalent AIs were obtained for ElgG2a (AI = 386 ± 25 and 389 ± 16, n = 3, for vector control and Fgr(WT), respectively) and ElgG2b (AI = 426 ± 20.4 and 422 ± 18, n = 3, for vector control and Fgr(WT), respectively) whether attachment was assessed at 4°C or 37°C with cytochalasin B–treated macrophages. In addition, AIs for EC3bi binding at 37°C in the absence of PDBu but in the presence of Mn²⁺ were equivalent for the two cell types (AI = 140 ± 9 and 154 ± 9, n = 3, for vector control and Fgr(WT), respectively). These data indicate that Fgr suppresses FcγR- and CR3-mediated ingestion, but not ligand binding. Taken together, all of the above data suggest that Fgr suppresses ingestion mediated by multiple receptors at a point subsequent to receptor ligation.

Because overexpression of Fgr could be artificially sup-

![Figure 1](image-url)
pressing phagocytosis, we assessed ingestion in Fgr(WT) BAC1.2F5 cells expressing varying levels of Fgr protein and in primary phagocytes from wild-type and mice deficient in Fgr expression by targeted gene deletion (22). Three Fgr(WT)-infected clones that expressed from 1 to 4 Fgr protein compared with the Fgr-negative vector control (Fig. 2 A) were assessed for their ability to ingest ElgG2a, ElgG2b, and EC3bi. As shown in Fig. 2 B, increasing concentrations of Fgr protein resulted in significantly decreased phagocytosis for all three types of opsonized erythrocytes. Moreover, resident peritoneal macrophages and bone marrow macrophages from Fgr(−/−) mice ingested significantly greater numbers of ElgG2a and EC3bi compared with macrophages from wild-type mice (Fig. 3, A and B). However, ingestion of ElgG2b was not as dramatically affected by Fgr expression as it was in the BAC1.2F5 cells (Fig. 3 B). The greatest difference was observed in resident peritoneal macrophages that expressed much higher levels of Fgr protein than did cultured bone marrow macrophages (data not shown). We also observed that the time, method, and concentration of LCM used for culture of the bone marrow macrophages affected Fgr expression, consistent with previous work (25). In contrast to macrophages, PMNs from Fgr(−/−) mice ingested significantly fewer ElgG2b even with FMLP stimulation to obtain optimal levels of ingestion (27). The fact that Fgr is essential for optimal FcγR-mediated phagocytosis in PMNs has not been observed previously (22) and suggests that the role of Fgr in regulating PMN phagocytic responses is more complex. The data depicted in Figs. 2 and 3 indicate that expression of Fgr either in a macrophage cell line or in primary macrophages negatively regulates the magnitude of the phagocytic response.

Figure 2. (A) Variable Fgr expression in three different BAC1.2F5 clones as assessed by Western blotting. (B) Effect of variable levels of Fgr(WT) expression (graded 1 to 4) by Western blotting, hatched and dotted bars compared with a vector control (black bars) on ingestion of ElgG2a, ElgG2b, and EC3bi by BAC1.2F5 macrophages. The data are depicted as the mean ± SEM, n = 3. These data indicate that Fgr suppresses phagocytosis in a dose-dependent manner.

Fgr Does Not Affect Macropinocytosis, Another Mechanism for Uptake of Particles as Large as Bacteria can Occur (36, 37). Formation of macropinosomes requires PI 3-kinase and actin polymerization (29, 36), two signaling elements required by both FcγR- and CR 3-mediated ingestion (29, 31, 33). We assessed macropinocytosis by the uptake of FITC-labeled 70-kD dextran (29) into macropinosomes measured both by fluorescence microscopy and flow cytometry to quantitate the amount of cell-associated fluorescence. Vector control and Fgr(WT)-expressing BAC1.2F5 cells took up the labeled dextran equivalently, and large macropinosomes were visualized in both cell types (data not shown). In addition, the level of cell-associated fluorescence was quantitatively identical as assessed by flow cytometry (MCF = 120 ± 19 vs 104 ± 16, n = 3, for the vector control and Fgr(WT) cells, respectively). Moreover, inclusion of CSF-1 to stimulate macrophages (29) increased the cell-associated fluorescence equivalently in both cell lines (MCF = 233 ± 53 vs 216 ± 41; data not shown). To prove that the mechanism of uptake was similar in both the absence and presence of Fgr, we assessed the effect of 10 nM wortmannin, a PI 3-kinase inhibitor, on macropinocytic uptake. The histograms of both the vector control and the Fgr(WT)-expressing cells were shifted significantly to the left by the inclusion of wortmannin, indicating an essential role for PI 3-kinase in macropinocytic uptake by both cell lines (data not shown). These data indicate that all mechanisms for uptake by macrophages are negatively affected by Fgr expression.

Fgr Negatively Regulates FcγR-Mediated Ingestion at a Point Distal to Receptor Clustering and Aggregation, but Proximal to the Reorganization of F-actin into Phagocytic Cups. To delineate the point in phagocytosis where Fgr exerts its effect, we examined both an early (receptor cross-linking) and a later (phagocytic cup formation) process that are essential for FcγR-mediated ingestion. Opsonin-dependent phagocytosis is initiated by engagement of specific receptors with ligands on the target surface, which induces receptor aggregation and migration into membrane domains where cytoplasmic signaling molecules are activated (1–3, 37). In fact, cross-linking of FcγRII/FcγRIII with a rat mAb against these receptors and a polyclonal anti-rat antibody is sufficient to activate downstream signaling molecules that are essential for phagocytosis mediated by these receptors and to mediate receptor internalization via endocytic mecha-
nisms (1, 34–38). As an example, the small GTPase, Rho, is required for phagocytosis, and the earliest step it regulates is Fcγ receptor clustering (38). Because the primary intracellular location for Src family kinases is the plasma membrane (39), Fgr could interfere with phagocytosis by blocking receptor aggregation and subsequent endocytosis. To examine this, we assessed the effect of Fgr expression on the endocytosis of FcγRII/FcγRIII induced by antibody-mediated aggregation as a measure of receptor clustering and aggregation. The percentage of internalized FcγRII/FcγRIII was equivalent for both vector control and Fgr(WT)-expressing cells at all three time points examined up to 1 h (from 36.2 ± 4.6% at 15 min to 69 ± 1.6% at 1 h for the vector control cells, compared with 34.4 ± 9.2% at 15 min and 62 ± 5.2% at 1 h for the Fgr(WT) cells; n = 3).

Because FcγR clustering and aggregation is required for endocytosis and endocytosis is not affected by Fgr expression, these data indicate that this early event is not affected by Fgr expression. Because this early event was not involved, we wanted to determine if the kinetics of ingestion was affected by Fgr expression or whether it simply blocked ingestion after a certain number of targets were internalized. As shown in Fig. 4, Fgr expression suppressed internalization as early as 10 min, suggesting that it was attenuating the phagocytic response throughout the time course of the assay. These data indicate that Fgr suppresses ingestion at some point distal to FcγR clustering and activation and that this suppression occurs early in the internalization process.

After FcγR cross-linking and initiation of intracellular signaling, within 5-10 min, pseudopodia containing F-actin extend over the surface of the target and create actin-rich phagocytic cups (1, 31–33). To assess the effect of Fgr on actin cup formation, adherent vector control and Fgr(WT)-expressing cells were allowed to attach ElgGR at 4°C for 2 h. After washing away the unbound ElgGR, phagocytosis was initiated by incubation at 37°C for 8 min. The distribution of F-actin relative to the bound ElgGR was visualized by rhodamine-phalloidin staining and fluorescence microscopy. At this time point, before a significant level of ingestion had occurred (Fig. 4), a majority of the vector control cells had F-actin distributed along the surface of the attached target-forming fluorescent phagocytic cups that extended from the surface of the cells (Fig. 5, A and B, arrows). In contrast, very few of the Fgr(WT)-expressing cells had F-actin distributed along the surface of the attached target-forming fluorescent phagocytic cups even though they had bound equivalent numbers of the opsonized targets (Fig. 5, C and D, arrows). In addition, the F-actin subjacent to the attached targets did not extend as far from the surface of the cell, suggesting a blunting of pseudopod extension. To quantitate these data, we assessed the number of actin cups formed per 100 cells that had attached 4 or more ElgGR as well as the percentage of rotsetting cells that had formed at least 1 actin cup. As shown in Fig. 5 E, Fgr(WT) macrophages had significantly fewer actin cups formed per 100 cells, and only 30% of these cells did not contain actin cups.
formed at least 1 actin cup, compared with 60–70% of the vector control cells. The number of actin cups correlated well with the PIs observed in the phagocytosis assays (Fig. 1). We also assessed actin cup formation with E1gG2b and obtained equivalent affects with Fgr expression (data not shown). These data indicate that Fgr negatively regulates FcγR-mediated phagocytosis at a point before the redistribution of F-actin into phagocytic cups surrounding the attached targets.

Fgr That Lacks Tyrosine Kinase Function Is Sufficient to Suppress Ingestion Mediated by Multiple Phagocytic Receptors. The members of the Src tyrosine kinase family have a homologous domain structure, and the sequence of the COOH-terminal kinase catalytic domain is highly conserved (39). Within this domain, a critical lysine residue (K279 in murine Fgr) is essential for ATP binding and phosphorylation of targeted tyrosine residues (40). Therefore, we assessed the effect of this lysine residue on the suppression of phagocytosis mediated by Fgr(WT). BAC1.2F5 cells expressing a mutant Fgr in which the lysine at position 279 was changed to an arginine (K279R) were compared with vector control and Fgr(WT)-expressing cells for phagocytic responses. Surprisingly, Fgr(K279R) was equally able to suppress ingestion mediated by both Fcγ and CR3 receptors, as was the wild-type form of murine Fgr (Fig. 6). Therefore, these data indicate that Fgr negatively regulates phagocytosis by a mechanism that does not involve its ability to phosphorylate any other protein.

Fgr Associates with a Complex of the ITIM-containing Receptor SIRPa and the Phosphatase SHP-1 after FcγR Aggregation. Positive signaling for FcγR-mediated phagocytosis involves multiple kinases, including tyrosine, lipid, and serine-threonine kinases (1–3). To address the signaling mechanism by which Fgr could be suppressing phagocytosis, we began by first assessing tyrosine phosphorylation patterns in vector control and Fgr(K279R)-expressing cells after FcγR aggregation. The use of the kinase-inactive cells (K279R), which have suppressed phagocytic responses (Fig. 6), allowed us to examine tyrosine phosphorylation independently of the additional phosphorylation mediated by Fgr. In the vector control cells, tyrosine phosphorylation of multiple proteins was increased within 0.5–5 min after FcγR activation (Fig. 7 A). In contrast, tyrosine phosphorylation in the Fgr(K279R) cells was significantly reduced, especially for proteins in the 95–120- and 50–65-kD range as soon as 30 s after FcγR activation. Tyrosine-phosphorylated Fgr is prominent in the 56-kD range. This altered pattern of tyrosine phosphorylation after FcγR aggregation in the Fgr(K279R) cells suggested the suppression of phagocytosis could be mediated, at least in part, by the recruitment of a tyrosine phosphatase to FcγR signaling domains.

Several inhibitory pathways involving phosphatase recruitment can be triggered by the activation of ITIM-containing receptors (41). In addition to the ITIM-containing FcγRIIB
receptor, mouse macrophages express other members of the inhibitory receptor family, including paired Ig-like receptor B (PIR-B), SIRPα, and platelet-endothelial cell adhesion molecule 1 (PECAM-1) (42–44). Because Fgr suppresses ingestion of both IgG- and C3bi-opsonized targets and because FcγRIIB is thought to inhibit only IgG-mediated responses (12), we speculated that other ITIM-containing receptors would be important for this negative regulation. Recently, both PIR-B and SIRPα were identified as major substrates and binding partners for the phosphatase SHP-1 in both BAC1.2F5 cells and bone marrow macrophages (42, 43). Because macrophages from mice with inactivating mutations in SHP-1 (motehean and motehean viable) are profoundly hyperresponsive, we hypothesized that SHP-1 would be a major regulator of phagocytic responses and that this regulation might be mediated through PIR-B and/or SIRPα. Because preliminary investigations revealed minimal expression of PIR-B relative to SIRPα on the vector control BAC1.2F5 cells, as suggested by others (43), we assessed whether Fgr would coassociate with SIRPα after FcγR aggregation and whether targeting SIRPα could affect phagocytic responses. To immunoprecipitate SIRPα, we used a rat mAb to an antigen first identified on neurons, P84, and subsequently shown to be identical to SHP substrate 1 (SHPS-1), BIT, and SIRPα (45). Vector control and Fgr(K279R) cells were stimulated via FcγR aggregation for various times, and the cell lysates were immunoprecipitated for SIRPα. The immunoprecipitates were subjected to immunoblotting to assess SIRPα protein expression, phosphorylated tyrosine content, Fgr association, and SHP-1 association. As shown in Fig. 7B, the vector control and Fgr(K279R) cells expressed equivalent levels of SIRPα that was constitutively tyrosine phosphorylated. After FcγR aggregation, the level of tyrosine phosphorylation declined, and this reduction was much more pronounced in the Fgr-negative cells. After 30 s, Fgr coassociated with SIRPα in the Fgr(K279R) cells, and this association was pronounced at 5 min. Minimal Fgr levels were detected in cell lysates in-
cubated with protein G alone or immunoprecipitated with rat anti-CD29 (data not shown). In addition, anti-PECAM-1 mAb did not immunoprecipitate with Fgr (data not shown). As reported by others (42, 43), SHP-1 was constitutively associated with SIRPα in both cells. However, in the absence of Fgr, the level of coassociated SHP-1 declined significantly after stimulation and was almost absent at 5 min. In the presence of Fgr, there was an initial decrease in coassociated SHP-1 after FcyR stimulation, with a subsequent increase at 2 and 5 min. These data suggest that Fgr coassociates with SIRPα after FcyR aggregation, and at the time of its maximal association (5 min), the tyrosine phosphorylation level of SIRPα is higher and there is a greater amount of coassociated SHP-1 compared with this time point in the absence of Fgr. To address whether this correlated with functional tyrosine phosphatase activity, we assessed the ability of the SIRPα immunoprecipitates to release phosphate from a pY-containing peptide. As shown in Fig. 7 C, after FcyR aggregation and activation, more tyrosine phosphatase activity was associated with SIRPα in the Fgr(K279R)-expressing compared with the vector control BAC1.2F5 macrophages.

To obtain evidence that SIRPα and SHP-1 could be implicated in regulating phagocytic responses, we assessed the effect of the P84 mAb against SIRPα on phagocytosis of EC3bi and ElgG2a by wild-type and Fgr⁻/⁻ bone marrow macrophages (Fig. 8 A) and by vector control and Fgr(K279R)-expressing BAC1.2F5 cells (Fig. 8 B). Incubation of Fgr-expressing primary macrophages or BAC1.2F5 cells with mAb P84 anti-SIRPα significantly enhanced phagocytosis of both EC3bi and ElgG2a. An isotype control antibody that recognizes an antigen expressed by both cell types, CD14, had no effect. In contrast, incubation of Fgr⁻/⁻ macrophages with anti-SIRPα had no effect on their phagocytic responses. Antibody against PECAM-1, another ITIM-containing receptor on these cells, also had no effect on ingestion by either cell type (data not shown). To assess whether the absence of the ITIM-containing receptor, FcyRIIB, could modulate this, we examined the effect of mAb anti-P84 on ingestion of EC3bi by macrophages that lack all three FcyRs, including FcyRIIB. Ingestion in the presence of the isotype control mAb was equivalent for both the wild-type and FcyR knockout macrophages (PI = 80 ± 4 vs. 76 ± 2, n = 3, respectively; data not shown). In addition, treatment with mAb P84 increased ingestion equivalently for both cell types (PI = 187 ± 14 vs. 176 ± 8, n = 3, respectively). These data indicate that the absence of the inhibitory FcyRIIB does not enhance phagocytosis of EC3bi and that anti-SIRPα is able to enhance EC3bi ingestion even in the absence of FcyRIIB. These data indicate that mAb P84 anti-SIRPα can reverse the negative regulatory effect of Fgr on FcyR- and CR3-mediated phagocytosis in a manner independent of the inhibitory FcyRIIB receptor.

To examine the role of SHP-1, we assessed ingestion of ElgG2a, ElgG2b, and EC3bi by cultured bone marrow macrophages from motheaten viable mice compared with control mice. As shown in Fig. 8 C, ingestion of both ElgG2a and EC3bi was markedly enhanced by me/me macrophages. As with the Fgr⁻/⁻ macrophages (Fig. 4), ingestion of ElgG2b was not significantly augmented. To examine whether SIRPα modulates ingestion by me/me macrophages, we assessed the effect of mAb P84 on the ingestion of ElgG by these cells. While mAb P84 significantly enhanced ingestion by wild-type macrophages (PI = 80 ± 7 control mAb vs. 247 ± 40 mAb P84, n = 3), it had no effect on ingestion by the me/me macrophages (PI = 267 ± 35 control mAb vs. 270 ± 34 mAb P84; data not shown). Because PIs in these cells can be >400 (Fig. 8 C), these data cannot be explained by a failure to enhance an already optimal level of ingestion. These data indicate that macrophages that contain a functionally inactive SHP-1 have enhanced phagocytic ability. These data suggest that mice with me(v) mice (hatched bars).

**Figure 8.** Effect of mAb P84 anti-SIRPα on phagocytosis by Fgr⁻/⁻ and Fgr⁻/⁻ bone marrow cultured macrophages (A) and vector control and Fgr(K279R)-expressing BAC1.2F5 cells (B). Cells were incubated with rat IgG1 anti-CD14 or rat IgG1 P84 for 15 min at room temperature. Ingestion of either ElgG2a or EC3bi was assessed as described. These data indicate that anti-P84 (dotted bars) enhances phagocytosis by Fgr-expressing macrophages compared with the isotype control (hatched bars). (C) Comparison of phagocytosis of ElgG2a, ElgG2b, and EC3bi by bone marrow macrophages from wild-type (black bars) and motheaten viable (me(v)) mice (hatched bars). These data indicate that mice with a nonfunctional form of SHP-1 have enhanced phagocytic ability.
Discussion

Phagocytes play an essential role in homeostasis by mediating the internalization and destruction of microbial pathogens. Two distinct molecular mechanisms for pathogen uptake have been proposed: one is the "zipper" mechanism that depends on repeated receptor-ligand interactions to drive pseudopod advance over the particle surface, and the other is a "trigger" mechanism that depends on the formation of membrane ruffles that fold back onto the surface of the cell, creating macropinosomes containing extracellular fluid and associated particles (37). Swanson and Baer (37) have observed that the two mechanisms differ in the magnitude of the pseudopodia produced, relative to the size of the particle: zipper produces a small pseudopod and triggering a large one. These authors hypothesize that this difference arises by spatially controlled production of both excitatory and inhibitory signals in the ligand-receptor-driven model of zipper (37). We contend that limitation or spatial control of the magnitude of the signal is beneficial to the host. Because ligation of FcγRs can initiate signals that generate reactive oxidants and release of lysosomal enzymes, spatial control would limit their production to the forming phagolysosome. A prediction of this hypothesis would be that negative signaling molecules could be found in zipper but not triggering mechanisms. Our present work indicates that the Src kinase family member, Fgr, is a negative signaling molecule for the zipper mechanism, phagocytosis, but not for the triggering mechanism, macropinocytosis. Moreover, the data presented in our paper are consistent with a model of negative regulation whereby Fgr raises the activation threshold necessary to generate a given magnitude of response. In this model, increasing the number of ligated and cross-linked receptors on a particle of sufficient size to trigger a phagocytic response (46) enhances the final readout of the pathway (i.e., ingestion) but cannot entirely overcome the negative signaling component contributed by the inhibitor molecule.

These data demonstrate that Fgr, like the Src family kinase member Lyn (17, 47, 48), plays an important role in negative regulation of hematopoietic cell activation. Because Lyn negatively regulates activation of the B cell antigen receptor and the high affinity receptor for IgE via phosphorylation of ITIM-containing receptors that recruit the lipid phosphatase SHIP (47, 48), we hypothesized that Fgr might work by a similar mechanism. Although we have obtained data implicating an ITIM-containing receptor expressed by macrophages, SIRPα, and a phosphatase, SHP-1 (Figs. 7 and 8), Fgr is clearly not responsible for phosphorylation of the ITIM motif in SIRPα (Fig. 7 B), and a kinase-inactive mutant is fully sufficient for the negative regulation of phagocytosis (Fig. 6). These data suggest that another kinase must be responsible for phosphorylating SIRPα and for regulating docking of SHP-1 in the plasma membrane. Because Lyn actively phosphorylates ITIMs in other receptors, it is intriguing to speculate that Lyn is responsible for phosphorylating SIRPα. If true, then the ability of Fgr to negatively regulate phagocytosis would be dependent on the function of Lyn. In this paradigm, Fgr may be functioning as an adaptor, through its SH2 and SH3 domains, to facilitate the association of SHP-1 with tyrosine-phosphorylated SIRPα (Fig. 9 B). Our data suggest a model whereby Fgr, through its SH2 domain, could protect SIRPα from dephosphorylation while allowing the recruitment and docking of SHP-1. Although we have focused the current studies on the regulation of tyrosine phosphorylation by Fgr (Fig. 7), other signaling elements, such as lipid and serine-threonine phosphatases, could also be involved. Further study will be required to determine if all of Fgr's ability to attenuate phagocytosis can be explained by recruitment of SHP-1.

These data, along with those of Hunter et al. (49) and Clynes et al. (12), which assessed the ability of inhibitory FcγRIIB to negatively regulate ingestion by activating FcγRs, are the first to define signal transduction elements that negatively regulate phagocytosis. However, there are some significant differences between our study and theirs. In our studies, the IgG-opsonized particle binds both the inhibitory receptor and the activating receptor, and the observed inhibition is dependent on specific interactions in the cytoplasmic regions of these two receptors (49). Thus, human FcγRIIB suppresses ingestion mediated by human FcγRIIa but not ingestion mediated by human FcγRIII (49), whereas in our work Fgr suppressed ingestion by multiple receptors including C.3, a non-IgG binding receptor. In fact, ingestion of ElgG2b, which is recognized by both inhibitory FcγRIIB and activating FcγRIII in murine macrophages, was not enhanced significantly in the absence of Fgr (Fig. 4) or by the lack of functional SHP-1 (Fig. 8). Moreover, the absence of FcγRIIB did not enhance ingestion of EC3bi, whereas anti-SIRPα did enhance ingestion. These data suggest that the SIRPα–Fgr–SHP-1 pathway we describe here is primarily involved in regulating other ligand-receptor pairings and is independent of FcγRIIB-mediated suppression. This fact does bring up an interesting question that relates to whether engagement of SIRPα is ligand dependent. Unlike FcγRIIB, ligands for most ITIM-containing receptors (41), including members of the SIRP family (45), have not been identified. In this regard, integrin-associated protein (IAP, CD47) was identified recently as a ligand for SIRPα expressed by neurons (P84 neutral adhesion molecule [50]). Because IAP is expressed by phagocytes and regulates matrix protein-augmented phagocytosis (51), it is intriguing to speculate that phagocytic receptors associate with molecules like SIRPα and IAP in membrane domains, and thus IAP may function as a coreceptor for SIRPα after phagocytic receptor activation.

Two other studies have examined the effect of Fgr on FcγR-mediated phagocytosis by monocytes or macrophages, and both indicated that it had no effect (22, 52). There are several reasons that may explain the discrepancy between our results and theirs. First, both studies presumed that Fgr would have a positive signaling role in phagocytosis in these cells and assumed that ingestion would be in-
creased in its presence. This is reasonable because Fgr is present in high concentrations in mature phagocytes (18–20, 25, 53) and immunoprecipitates with phagocytic receptors after cell stimulation (21). Therefore, the phagocytosis assays were done with one level of opsonin, which could have represented such a high concentration that suppression of ingestion would have been difficult to observe (22, 52). In contrast, we examined phagocytosis over a wide dose–response range to demonstrate differences in the level of ingestion. In addition, the ligand on the opsonized particle affects the magnitude of the Fgr effect. For example, we found the greatest effect in wild-type cells with IgG2a- and C3bi-opsonized particles. Therefore, studies using polyclonal rabbit IgG–opsonized erythrocytes, which could target multiple receptors, might not observe negative regulation. Moreover, the work of Faulkner et al. (52) was performed with the human monocytic cell line, U 937, which was transfected with human Fgr. U 937 cells express FGR endogenously (52), and therefore, ingestion may have been suppressed constitutively such that increasing the level of FGR by transfection would not have an effect. In addition, we have found that the concentration of SIRPα expressed by phagocytic cell lines can vary (Gresham, H. D., unpublished observations), and SIR Pα is required to observe the negative effects of Fgr.

Our data indicate that the role of Fgr in regulating phagocytic responses is completely different in neutrophils than it is in macrophages (Fig. 3). Although this result was unexpected, it is consistent with other data that implicate a positive signaling role for Fgr in neutrophils (53–55). Fgr exists in intracellular granules in resting neutrophils and translocates to the plasma membrane with cell activation (53). Moreover, it immunoprecipitates with the phagocytic receptors FcγRIIA (54) and CR3 (55) after neutrophil activation. If this positive signaling role for Fgr were mediated through its kinase function, a negative signaling component could still be contributed independently of its kinase activity through recruitment of SHP-1. Therefore, we believe that Fgr could have both positive and negative roles in regulating neutrophil phagocytic responses. Even in macrophages where a negative role for Fgr was clearly demonstrated, the magnitude of the response varied. We observed that the source of the macrophage and cell culture conditions had a significant effect on Fgr expression. These data confirm in vivo observations that indicate that the level of Fgr present in monocytic cells varies as they mature into tissue-fixed macrophages (56). In fact, some macrophage populations, such as Kupffer cells, alveolar macrophages, and Peyer's patch macrophages, do not express detectable levels of Fgr (56). Intriguingly, these cells are some of the most phagocytic cells in the reticuloendothelial system. Alternatively, exposure of macrophage populations to agents like LPS or CSF-1, which could occur at sites of inflammation or infection, can induce the expression of Fgr (25, 56). Therefore, the contribution of Fgr to the balance of positive and negative signaling in phagocytosis will depend on the phagocyte, its maturation level, its activation state, and the ligand-opsonized target.

These data significantly extend the knowledge of Fgr biology in macrophages. Although the targeted deletion of Fgr in combination with other Src kinases such as Hck and Lyn has revealed significant information about the roles of Src kinases in neutrophil and macrophage function (22, 57–59), very little has been ascertained about Fgr’s unique role in hematopoietic cells. Therefore, our data demonstrating an important kinase-independent role for Fgr in negative regulation of phagocytosis are significant. While our data do not address the possible kinase-dependent functions of this Src kinase family member, it is intriguing to speculate that Fgr, like Lyn (17, 47, 48), could have both positive and negative roles in regulating hematopoietic cell function.

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